



CRANES:

Their Biology, Husbandry, and Conservation

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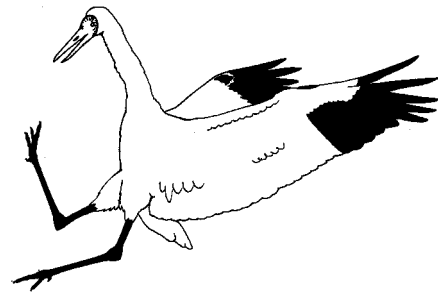
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Table of Contents



AUTHORS AND ADDRESSES	vii
FOREWORD.....	ix
PREFACE	xi
1. CRANE BIOLOGY.....	I
George W. Archibald, James C. Lewis	
2. GENERAL HUSBANDRY	31
Scott R. Swengel, James W. Carpenter	
3. EGG AND SEMEN PRODUCTION	45
Claire M. Mirande, George F. Gee, Ann Burke, Peter Whitlock	
4. INCUBATION AND HATCHING.....	59
Robert R. Gabel, Thomas A. Mahan	
5. CHICK REARING	77
Marianne Wellington, Ann Burke, Jane M. Nicolich, Kathleen O'Malley	
A. VETERINARY TECHNIQUES FOR REARING CRANE CHICKS.....	95
Glenn H. Olsen, Julia A. Langenberg	
6. BEHAVIOR MANAGEMENT	105
Scott R. Swengel, George W. Archibald, David H. Ellis, Dwight G. Smith	
A. IMPRINTING, ATTACHMENT, AND BEHAVIORAL DEVELOPMENT IN CRANES	117
Robert H. Horwich	
7. REPRODUCTIVE PHYSIOLOGY	123
George F. Gee, Shirley E. Russman	
8. MEDICINE AND SURGERY.....	137
Glenn H. Olsen, Julia A. Langenberg, James W. Carpenter	
9. GENETIC MANAGEMENT.....	175
Claire M. Mirande, George F. Gee, Scott R. Swengel, Christine Sheppard	
10. RECORDS	185
David H. Ellis, Joanna A. Taylor, Claire M. Mirande, Julia A. Langenberg, Marianne Wellington, B. H. Powell, Janet L. McMillen	

II. SPECIAL TECHNIQUES

A. ARTIFICIAL INSEMINATION 205
George F. Gee, Claire M. Mirande

B. CRYOPRESERVATION 219
Timothy L. Hargrove, George F. Gee

C. SEX DETERMINATION 223
Scott R. Swengel

D. REINTRODUCTION TECHNIQUES 231
Meenakshi Nagendran, Richard P. Urbanek, David H. Ellis

E. FLIGHT RESTRAINT 241
David H. Ellis, F. Joshua Dein

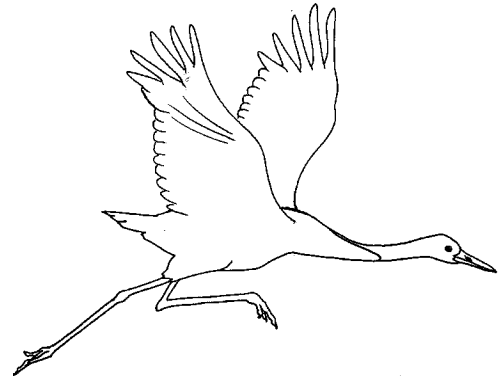
F. PREDATOR AND PEST MANAGEMENT 245
Thomas E. Lewis

12. FACILITIES 253
Scott R. Swengel, Richard W. Besser

13. ECOLOGY, STATUS, AND CONSERVATION 263
Curt Meine, George W. Archibald

APPENDIX. EQUIPMENT AND SUPPLIERS 293

INDEX 301



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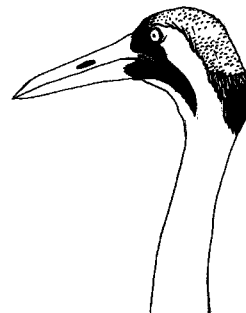
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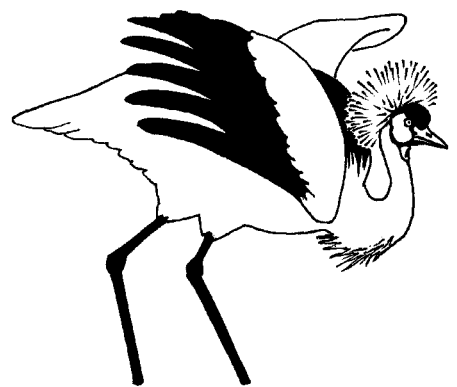
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Foreword

Captive Management and Conservation

Worldwide, extinction of our flora and fauna is proceeding at catastrophic rates. We humans are fouling our planetary nest and making it uninhabitable both for many thousands of wild species and ultimately for ourselves. For utilitarian, aesthetic, and ethical reasons, we must do all that we can to prevent extinction. The most important conservation strategy for most species is habitat preservation. This almost always means trying to prevent or to reverse man-caused changes in the environment.

While habitat conservation is the key, captive breeding sometimes plays a crucial role. It may then be possible to release captive-bred animals once their habitat has been rescued, as has so successfully been achieved with the Arabian Oryx in Oman and Jordan, and with the Hawaiian Geese bred at the Wildfowl Trust. Even if no immediate prospect of release can be foreseen, endangered species must be maintained in captivity to prevent extinction and in hopes that habitat will one day be available.

Captive management is not only a vital conservation tool in helping to prevent extinction, but there are also many spin-off benefits. The worldwide effort to propagate and conserve cranes is a shining example. Not only have there been many successes in rearing endangered cranes, but there has also developed an international spirit of cooperation between individuals and institutions in a dozen nations around the world. In addition to fostering international good-will in this way, the captive cranes provide many opportunities for research, answering questions that would be impossible to resolve in the field.

*Sir Peter Scott, CH CBE DSC FRS
Slimbridge
October 10, 1988*

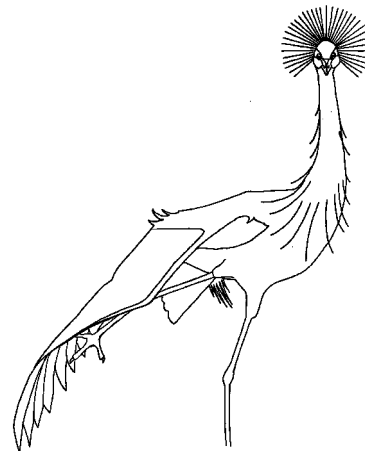
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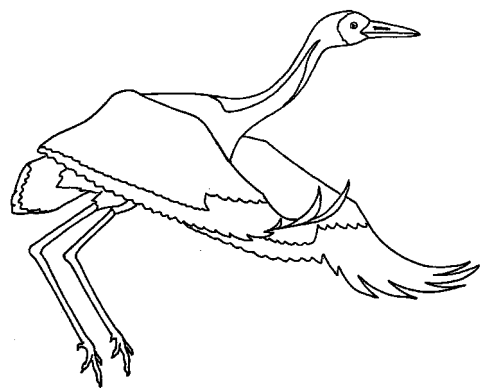
The natural world lost a premier advocate when Sir Peter Scott died on 29 April 1989 at age 79. His thoughts on endangered species, conservation in general, and crane propagation in particular, continue to ring true.

The first published reference to crane husbandry is probably Marco Polo's account of several species of cranes (some of which were more likely phasianids) in the gardens of Kublai Khan in the late 13th Century. With more than two decades of propagation research behind us at the International Crane Foundation (ICF) and nearly three at the Patuxent Wildlife Research Center (Patuxent), crane husbandry for most species is now operational. It is finally time to collect the best of crane avicultural science and husbandry between two covers.

Experimentation with crane reintroduction techniques over the last two decades allows us to also include a section on this culminating aspect of crane conservation.

David H. Ellis





Preface

Unbridled, mankind is unique in having the ability to erase the cranes, one or all, from our world. Ironically, it is also within the power of man to recognize the beauty and worth of something not our kin (or stated more correctly, to recognize our kinship to a dissimilar species), to recognize and revere. This book is dedicated to these lovely creatures, and to those persons living, remembered, and yet unborn who will continue the struggle to ensure a world rich with cranes.

In the paragraphs that follow, we will cite some of those whose contributions were greatest in completing this volume and in supporting our husbandry efforts. Others will be mentioned in photo captions and text. However, this act (i.e., citing our fellow workers) should not, in any way, diminish our awareness of, and appreciation for, the true inspiration for this volume, namely, the 15 species of cranes that grace our planet. This book is a celebration of the magnificence of these creatures and of man's efforts to ensure survival of each species and each race of this resplendent group.

As we, the editors, complete the eight year ordeal that culminates in publication of this volume, it is our pleasure to look back and thank those who gave of their time in reviewing the chapters, those who devoted months (often without salary) to rearing chicks or caring for adults, and most of all we thank those who created and maintained our programs at ICF and Patuxent. In particular, Dr. Ray C. Erickson and Dr. Ronald T. Sauey deserve our deep appreciation. Ray's foresight led to the creation of Patuxent's endangered species program in the late 1960's. Ron's dedication was essential in

the creation of ICF in 1973. Without the efforts of these two and of Dr. George W. Archibald, much of the information encompassed in this book would be as yet unassembled. We also wish to acknowledge Dr. H. Randolph Perry, Jr. and Mr. James Harris who have shouldered much of the administrative load through the years and thereby provided the opportunity for us to create this volume.

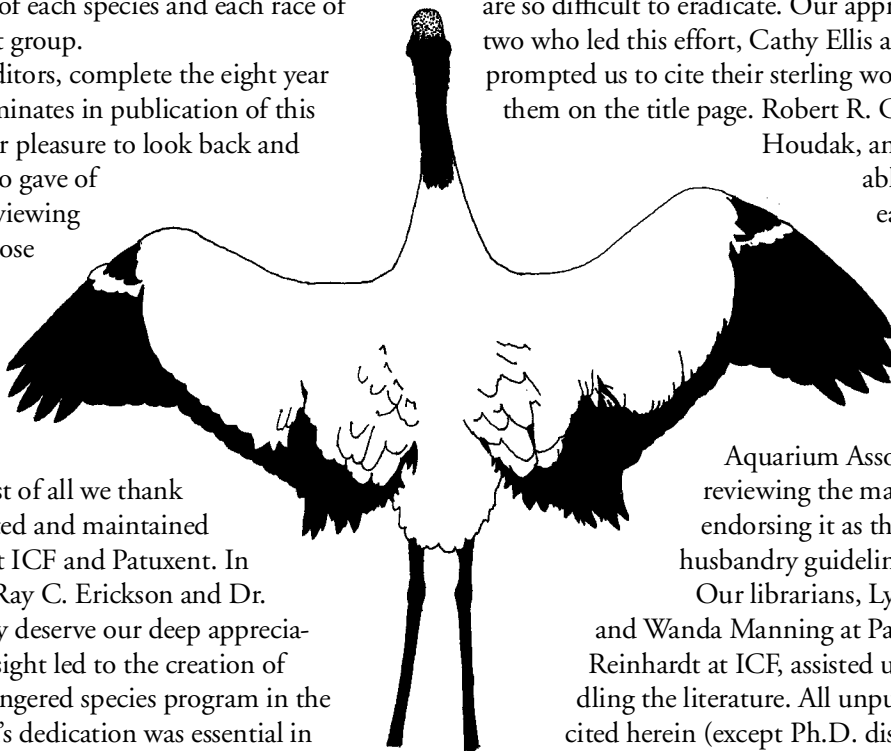
As editors, we appreciate the work of the authors of each chapter. Their patience was often exemplary as they worked with us in shaping and honing the text. Several gave selflessly in preparing tables, graphics, or even writing text for chapters not their own. Dozens participated in review of the book and preparation of the materials, and all have been thanked personally. Here we wish to acknowledge those whose contributions were extensive. Marie Childress, Cathy Ellis, Jennifer Gieg, Kathleen O'Malley, and Lorie Shaull spent many hours reviewing chapters and proofing tables, always in quest of the erroneous minutia that are so difficult to eradicate. Our appreciation for the two who led this effort, Cathy Ellis and Lorie Shaull, prompted us to cite their sterling work by including them on the title page. Robert R. Gabel, Sandy

Houdak, and Linda J. Miller ably assisted in the early reviews.

We thank the Crane Taxon Advisory Group of the American Zoo and

Aquarium Association for reviewing the manual toward endorsing it as the official husbandry guidelines for cranes.

Our librarians, Lynda Garrett and Wanda Manning at Patuxent and Annie Reinhardt at ICF, assisted us greatly in handling the literature. All unpublished reports cited herein (except Ph.D. dissertations



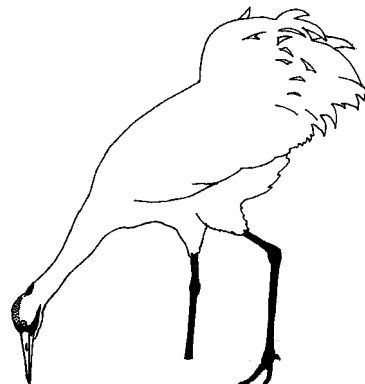
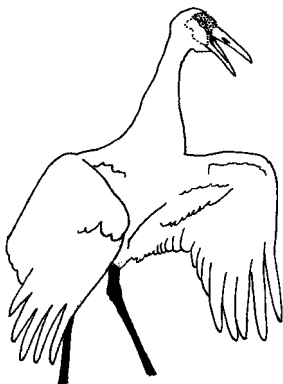
produced in the U.S. which are available through University Microfilms, Box 1764, Ann Arbor, MI 48106) are on file at the Ron Saucy Memorial Library at ICF.

Although we will not cite here all who contributed to each chapter, we wish to make special mention of Milford Muskett for preparing the maps gratis, Kinnard Boone for his computer graphics assistance in several chapters, Lyena Romanova for her computer programming efforts towards automating the Patuxent records system, Dr. David H. Thompson for help with the photographs, and Kathleen O'Malley for composing forms and entering data for the records chapter.

Other significant contributions were made by those who provided the photographs and illustrations for the text. Most gave without reimbursement. In particular we thank David Rankin (who supplied the end sheet/dust jacket painting at cost), Billi Wagner (who provided the crane line drawings with black zones) and Paul Triatak (who provided the crane line drawings without black zones, gratis).

Inasmuch as crane conservation has become a banner cause in conservation around the globe, it is a fitting, if somewhat belated, act to produce this "how to" volume on raising cranes, something that has been done haphazardly for some species for centuries and has become operational for most species during the last two decades.

The Editors



Crane Biology



George W. Archibald and James C. Lewis

Cranes, an ancient family of birds, have graced our planet's skies and stalked the grasslands and wetlands for at least 40 million years. The fossil record includes at least 17 extinct species, many of which were closely related to African Crowned Cranes (Brodkorb 1967). No crane species has become extinct within recorded history. Fifteen species (Figs. 1.1–1.16) and 14 recognized subspecies survive (Table 1.1).

There have been four comprehensive volumes written about the biology of the world's 15 species of cranes (Blaauw 1897; Makatsch 1970; Walkinshaw 1973; Johnsgard 1983). In addition, there have been comprehensive single species accounts on Sandhill Cranes (Walkinshaw 1949), Whooping Cranes (Allen 1952; McNulty 1966; Doughty 1989), Red-crowned Cranes (Masatomi 1970–1974), and Siberian Cranes (Sauey 1985). Major contributions on Black-necked Cranes (Bishop 1994), Grey Crowned Cranes (Gichuki 1995), and Blue Cranes (Allan 1995) will soon be available.

Special centers for crane research provide an abundance of published and unpublished information about the husbandry of cranes. These include the Department of the Interior's Patuxent Wildlife Research Center (11510 American Holly Drive, Laurel, MD 20708-4019), International Crane Foundation (E-11376 Shady Lane Road, Baraboo, WI 53913-0447), Oka State Nature Reserve (391072 Lakash, Spasskogo Raiona, Ryazanskoi Oblasti, Okskii Zapovednik, Russia), Vogelpark Walsrode (D-3030, Walsrode, AM Rieselbach, Germany), Kushiro Crane Park (c/o Kushiro Zoo, 11 Ninishibetu Akan Cho, Akan Gun, Hokkaido, Japan), Serendip Research Center (P.O. Box 2, Lara, Victoria, 3212, Australia), Beijing Zoo (137 Xi Zhi Men Wai St., Beijing, China 100044), Shenyang Center for the Study, Preservation, and Breeding of Cranes (No. 1, Wanquan St., Dadong District, Shenyang, China 110015), and the Conservation and Research Center of the Smithsonian Institution (The Wildlife Survival Center, Front Royal, VA 22630).

The Ron Sauey Memorial Library for Bird Conservation at the International Crane Foundation (ICF) is a repository for the world's literature on cranes and their habitats. Ron Sauey was a co-founder of the International Crane Foundation. In 1987, at the age of 37, he tragically passed away as a consequence of a cerebral hemorrhage. The construction of the library was supported by the Sauey family in memory of Ronald. The library contains English translations of the most important non-English publications. The library is connected by modem to the library system of the University of Wisconsin and can be accessed at the following telephone number: 1-608-262-8670.

Natural History

Taxonomy

Cranes are found on five continents. There is no evidence that cranes ever inhabited South America. The current concentration of crane species in Asia and Africa suggests an Old World origin of Gruinae, with a more recent colonization of Australia and North America (Archibald 1976a). Most fossil species, however, have been found in North America (Brodkorb 1967), reflecting both the proportionately greater amount of paleornithological work in North America (Archibald 1976a) and the possible origins of cranes in the West. Krajewski (1988), however, believes that cranes originated in Europe near the end of the Paleocene Epoch.

All cranes are in one of two subfamilies, *Balearicinae* or *Gruinae*, in the family *Gruidae*. The two species of African Crowned Cranes are placed in the subfamily *Balearicinae* (Peters 1934). They are distinguished from other species by their ability to roost in trees, and their loose plumage, straight trachea, elaborate crests, and colorful facial markings. The inability to tolerate extended periods of freezing temperatures perhaps led to the extinction

TABLE 1.1

World species and subspecies of cranes and their geographic distribution (Walkinshaw 1973).

Common Name	Scientific Name	Distribution
Family Gruidae		
Subfamily Balearicinae		
Black Crowned Crane	<i>Balearica pavonina</i>	
West African Crowned Crane	<i>B. p. pavonina</i>	West Africa
Sudan Crowned Crane	<i>B. p. ceciliae</i>	Central Africa
Gray Crowned Crane	<i>Balearica regulorum</i>	
East African Crowned Crane	<i>B. r. gibbericeps</i>	East Africa
S. African Crowned Crane	<i>B. r. regulorum</i>	Southern Africa
Subfamily Gruinae		
Wattled Crane	<i>Bugeranus carunculatus</i>	Africa
Blue Crane	<i>Anthropoides paradisea</i>	Southern Africa
Demoiselle Crane	<i>Anthropoides virgo</i>	Asia, Africa
Siberian Crane	<i>Grus leucogeranus</i>	Asia
Sandhill Crane	<i>Grus canadensis</i>	
Lesser Sandhill Crane	<i>G. c. canadensis</i>	East Siberia Arctic N. America
Canadian Sandhill Crane	<i>G. c. rowani</i>	Boreal Canada
Greater Sandhill Crane	<i>G. c. tabida</i>	Northern USA
Florida Sandhill Crane	<i>G. c. pratensis</i>	Southeast USA
Mississippi Sandhill Crane	<i>G. c. pulla</i>	Mississippi
Cuban Sandhill Crane	<i>G. c. nesiotis</i>	Cuba
White-naped Crane	<i>Grus vipio</i>	East Asia
Sarus Crane	<i>Grus antigone</i>	
Indian Sarus Crane	<i>G. a. antigone</i>	India
Eastern Sarus Crane	<i>G. a. sharpii</i>	Southeast Asia
Australian Sarus Crane	<i>G. a. gilli</i>	Australia
Brolga	<i>Grus rubicunda</i>	Australia
Eurasian Crane	<i>Grus grus</i>	
European Crane	<i>G. g. grus</i>	Europe, west Asia
Lilford's Crane	<i>G. g. lilfordi</i>	East Asia
Hooded Crane	<i>Grus monacha</i>	East Asia
Black-necked Crane	<i>Grus nigricollis</i>	Tibetan Plateau
Red-crowned Crane	<i>Grus japonensis</i>	East Asia
Whooping Crane	<i>Grus americana</i>	North America

of Crowned Cranes on the northern continents during the Pliocene Epoch. Today, the two surviving species inhabit the wetlands and savannas of Africa. Paleontological, anatomical, behavioral, and DNA studies all indicate that Crowned Cranes are closest to the ancestral stock that gave rise to the more recent subfamily *Gruinae* (Archibald 1976a; Wood 1979; Krajewski 1988) which includes the other 13 species.

The 13 species of *Gruinae* were traditionally divided into three genera: *Bugeranus*, *Anthropoides*, and *Grus*. Recent DNA hybridization studies, however, suggest that *Anthropoides* and *Bugeranus* should be merged with *Grus* (Ingold 1984; Krajewski 1988). *Grus* includes four distinct groups of closely related species: the Sarus Species Group (White-naped, Sarus, Brolga), the Whooping Crane Species Group (Eurasian, Hooded, Black-necked, Red-crowned, Whooping Crane), and the Sandhill Crane and the Siberian Crane which each stand alone. The Sandhill Crane is probably most closely allied to the Sarus Group (Archibald 1976a). Ethology and anatomy weakly link the Siberian Crane to the Wattled Crane (Archibald 1976b; Wood 1979), but DNA and recent behavior work suggest that the Wattled Crane and *Anthropoides* (Demoiselle and Blue) are closely related (Krajewski 1988; Ellis et al. *in prep*), and that the Siberian Crane is distinct from many of the other *Gruinae* species groups and should perhaps be placed in a separate genus (Krajewski 1988).

Food Habits

Cranes are omnivorous and some species rely heavily on aquatic foods (Walkinshaw 1973). Most cranes probe the subsurface with their bills and take foods

from the soil surface or vegetation. Young chicks are fed by their parents and gradually become more independent in their feeding until they separate from the parents prior to the next breeding season. During these first 10 months of development, captive cranes are extremely inquisitive. Perhaps this drive to investigate novel objects helps them discover food items in the wild.

Sandhill Cranes feed primarily on small grains (corn, wheat, barley, and sorghum) in fall, winter, and spring, but during the nesting season (when they associate more with wetlands), the greater part of the diet consists of crayfish, plant tubers, chufa, rodents, frogs, berries, bird's eggs, and nestlings (Walkinshaw 1949; Lewis 1974; Bennett 1978; Mullins and Bizeau 1978; Iverson et al. 1982; Herter 1982). Summer foods of the Whooping Crane include frogs, minnows, berries, and large nymphal and larval forms of insects (Allen 1956; Novakowski 1966). Principal winter foods of Whooping Cranes include blue crabs, clams, marine worms, amphibians, crayfish, fish, snails, insects, and sedge tubers found in coastal marshes and estuaries, but these cranes also feed in uplands on berries, acorns, insects, and small vertebrates (Allen 1952; Uhler and Locke 1969; Hunt and Slack 1987).

The 15 crane species can be divided into several groups based on the habitats in which they feed during the breeding and nonbreeding season (Table 1.2). The less common species worldwide, like the Whooping Crane, Siberian Crane, Wattled Crane, Red-crowned Crane, Black-necked Crane, and White-naped Crane, are more dependent on aquatic habitats throughout the year and not just during the breeding season.

TABLE 1.2

Breeding and nonbreeding season food habits of cranes.

Habitat	Breeding Season	Nonbreeding Season
Primarily feeding in uplands	Demoiselle, Blue Cranes	Crowned, Demoiselle, Blue, Sandhill, Eurasian Cranes
Feeding in both uplands and wetlands	Crowned, Sandhill, Sarus, Eurasian, White-naped, Black-necked Cranes, Brolga	Sarus, White-naped, Red-crowned, Black-necked, Hooded Cranes, Brolga
Primarily feeding in wetlands	Wattled, Siberian, Hooded, Red-crowned, Whooping Cranes	Wattled, Siberian, Whooping Cranes

A Color Signature of Cranes Around the World

Figure 1. *Black Crowned Crane.*
Photo by Damian Debski

Figure 2. *Gray Crowned Crane, Zambia.*
Photo by L. H. Walkinshaw

Figure 3. *Wattled Cranes, Wakkerstroom, South Africa.*
Photo by Warwick Tarboton

Figure 4. *Blue Cranes, Wakkerstroom, South Africa.*
Photo by Warwick Tarboton

Figure 5. *Demoiselle Cranes.*
Photo by J. H. Dick

Figure 6. *Siberian Crane at Keoladeo National Park, India.*
Photo by ICF

Figure 7. *Sandhill Crane, Wisconsin.*
Photo by George W. Archibald

Figure 8. *White-naped Crane at Zhalong, China.*
Photo by Sture Karlsson

Figure 9. *Sarus Cranes Unison-call at Bharatpur, India.*
Photo by M. Phillip Kahl

Figure 10. *Brolgas, Queensland, Australia.*
Photo by George W. Archibald

Figure 11. *Eurasian Cranes at Zao Hai, China.*
Photo by George W. Archibald

Figure 12. *Hooded Cranes at Izumi, Japan.*
Photo by Sture Karlsson

Figure 13. *Black-necked Cranes at Cao (or Zao) Hai, China.*
Photo by George W. Archibald

Figure 14. *Red-crowned Cranes.*
Photo by Terao Sato

Figure 15. *Whooping Cranes at Aransas National Wildlife Refuge, Texas.*
Photo by Mary Bishop

Figure 16. *Demoiselle Cranes captured on migration in Pakistan are eaten or rendered flightless and tamed.*
Photo by Steven E. Landfried



Fig. 1.1. *Black Crowned Crane* (*Balearica pavonina*).

Photo Damian Debski



Fig. 1.2. *Gray Crowned Crane (Balearica regulorum), Zambia.*

Photo L. H. Walkinshaw



FIG. 1.3. Wattled Cranes (*Bugeranus carunculatus*), Wikkertstroom, South Africa.

PHOTO WARWICK TARBOTON



FIG. 1.4. *Blue Cranes (Anthropoides paradisea), Wakkerstroom, South Africa.*

PHOTO WARWICK TARBOTON



FIG. 1.5. *Demoiselle Cranes (Anthropoides virgo)*.

PHOTO J. H. DICK



Fig. 1.6. *Siberian Crane (Grus leucogeranus)* at *Keoladeo National Park, India, 1982.*

Photo ICF



Fig. 1.7. *Sandhill Crane* (*Grus canadensis*), *Wisconsin*.

Photo George W. Archibald



PHOTO STURE KARLSSON

FIG. 1.8. *White-naped Crane (Grus vipio) at Zhulong, China.*



FIG. 1-9. *Sarus Cranes (Grus antigone) Unison-call at Bharatpur, India, 1967.*

PHOTO M. PHILLIP KAHL



FIG. 1.10. *Grus rubicunda*, Queensland, Australia.

PHOTO GEORGE W. ARCHIBALD



FIG. 1.11. *Eurasian Cranes (Grus grus) at Zao Hai, China.*

PHOTO GEORGE W. ARCHIBALD



PHOTO STURE KARLSSON

FIG. 1.12. Hooded Cranes (*Grus monacha*) at Izumi, Japan.



FIG. 1.13. *Black-necked Cranes (Grus nigricollis) at Cao (or Zao) Hai, China.*

PHOTO GEORGE W. ARCHIBALD



FIG. 1.14. *Red-crowned Cranes (Grus japonensis)*.

PHOTO TERAO SATO



FIG. 1.15. Whooping Cranes (*Grus americana*) at Aransas National Wildlife Refuge, Texas.

PHOTO MARY BISHOP



Fig. 1.16. *This Demoiselle crane, captured on migration in Pakistan, became a pet.*

Photo Steven E. Landfried

Plumage Coloration

Crane chicks (Fig. 1.17) are first covered with natal down which is largely concealed or replaced by juvenal plumage by fledging time (Stephenson 1971). During the first few weeks, the legs and neck of a crane chick grow proportionately faster than the wings. Juvenile cranes are either predominantly reddish brown (Crowned, Siberian, White-naped, Sandhill, Red-crowned, and Whooping Cranes) or gray (Demoiselle, Blue, Wattled, Sarus, Eurasian, Hooded, and Black-necked Cranes, and Brolgas). The juvenile colors probably provide anti-predator camouflage.

Adult cranes are either white, gray, black, or combinations thereof. White cranes inhabit vast open wetlands where excellent visibility makes white birds extremely apparent. Being white may help territorial pairs become more obvious to potential intruders and thus minimize the amount of time and energy spent in aggressive encounters. The gray cranes occupy smaller wetlands that are often partially or completely covered by trees. Terrestrial predators are undoubtedly more of a threat in forested wetlands than on open wetlands. The gray color helps the crane conceal itself in the marsh and on the nest.



Fig. 1.17. An 11-day-old Sandhill Crane.

Photo Glen Smart

Sandhill and Eurasian Cranes also perform feather painting that may provide further protective coloration especially for incubating adults. In early spring, they paint their feathers with mud, the iron oxides of which penetrate and permanently stain the feathers a rust color (Taverner 1929).

Social Behavior

Cranes have a wide variety of vocal and visual displays (Ellis et al. *In prep.*). The African Crowned Cranes have rather simple loud “honking” calls. The Siberian Crane has a high-pitched, flute-like call, whereas Wattled Crane calls are high-pitched but raspy. Demoiselle, Blue, and Sandhill Cranes have low, broken calls, and the remaining species have shriller calls. The trachea of Gruinae cranes coils within, and fuses with, the sternum to varying degrees in each species (Blaauw 1897). Tracheal development is greatest in the Whooping Crane Species Group, and the pitch of the calls in these species is higher than in most other cranes. The trachea and sternum amplify the calls produced in the larynx (Prange et al. 1985).

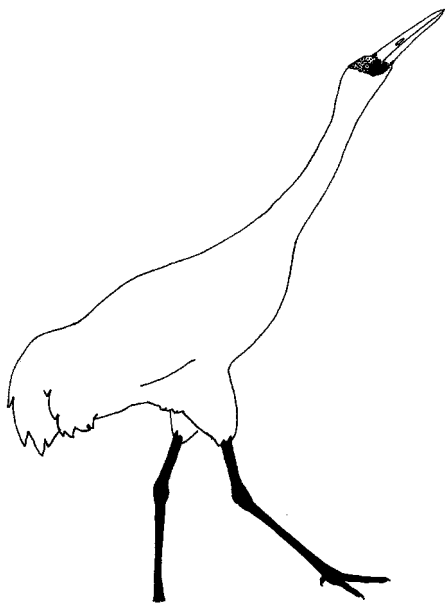
Crane calls include low, purr-like Contact Calls, slightly louder Pre-flight Calls, purr-like or shrill Pre-copulatory Calls, groan-like or scream-like Distress Calls, scream-like plaintive Location Calls, abrupt Alarm Calls, and loud Flight Calls and Guard Calls. Crane calls also include loud, complex duets called Unison Calls (Allen 1952; Masatomi and Kitagawa 1975; Archibald 1976a, 1976b; Voss 1976) which have both sexual and threat functions (Archibald 1976a).

Plumage-wise, cranes are sexually monomorphic, but the vocal and visual components of the Unison Call (an antiphonal duet) are sexually distinct (Fig. 11C.1), the exceptions being the Black Crowned, Gray Crowned, and Siberian Cranes. Wattled Cranes seldom Unison Call, but when they do the male slightly elevates his wings above his back for a second at the end of the duet. In other cranes, the male typically emits a long series of low calls, and the female accompanies him with two or three high-pitched calls for each low call of the male. In Blue Cranes and the Sarus Species Group, the males invariably elevate their wings and droop their primaries during the Unison Call, while the females keep their wings closed. Demoiselle Cranes call with wings closed, but the female usually holds her neck back, slightly beyond

the vertical. Sandhill Cranes also call with closed wings, but in contrast to Demoiselle Cranes, the male holds his head close to the vertical position while the female calls with her beak horizontal. In Siberian Cranes and the Whooping Crane Species Group, the wings may be elevated in either sex depending on the intensity of the situation, with wing elevation being proportional to the level of threat or intensity of display.

Other social displays include rigid threat posturing, rigid Strutting, Ritualized Preening of the back or thigh, feather ruffling, Stamping, Flapping, tail fluttering, Crouching, Growling, and Hissing. Cranes also perform an elaborate dance involving Bowing, Leaping, Running and Flapping, tossing an object (often a feather) into the air, and more (see Chapter 6).

The form of the complex visual and vocal displays of cranes is apparently independent of learning or the species of the foster parent; these displays appear to be genetically determined. Even blind cranes in captivity are able to perform a full complement of crane behavior. The object at which the display is oriented, however, is learned. If a crane chick is reared by people, it will prefer to associate with people and not cranes. Learned species recognition is important in maintaining reproductive barriers between sympatric species. For example, White-naped Cranes and Red-crowned Cranes are sympatric on many of their nesting and wintering areas, but hybrids have not been reported in the wild.



Breeding Biology

Annual Cycle

The annual cycle of cranes can be divided into a 3–5 month nesting period and a longer non-breeding period. Many species (Demoiselle, Siberian, White-naped, Eurasian, Hooded, Black-necked, Red-crowned, Whooping, and three migratory subspecies of Sandhill Cranes) migrate hundreds, or even thousands, of kilometers between breeding and wintering grounds. Except for Wattled Cranes, which sometimes remain on nesting territories throughout the year (Tarboton 1984), all cranes become more gregarious during the non-breeding period and move to regions where food is abundant. Eurasian Cranes, and possibly Red-crowned Cranes, do not breed consistently every year, an aspect of crane biology that requires further research.

Pair Formation and Duration

Successful breeding depends on securing a compatible mate and a breeding territory. In the Sandhill Crane, unpaired males sometimes establish a breeding territory and wait for the arrival of a female. Unmated females, by contrast, search for a male that has an established territory (Nesbitt 1989).

In most cranes, breeding usually begins between ages 3 and 6. Whooping Cranes sometimes breed as early as age 3 (Kuyt and Goossen 1987), but most produce fertile eggs at age 4 or 5. Breeding, on average, occurs later in Whooping Cranes in captivity (Ellis et al. 1992). Sandhill Cranes begin breeding at ages 2 to 5 depending on subspecies and location (Radke and Radke 1986; Nesbitt and Wenner 1987; Tacha 1988; Nesbitt 1992). A young crane is perhaps more likely to breed when paired with an experienced breeder that has lost its mate.

In Sandhill Cranes, sub-adult pairings are usually ephemeral (Nesbitt 1989). Nesbitt and Wenner (1987) found that the average, sub-adult Sandhill Crane paired five times before successfully breeding, with pair bond duration related to the production of young. Pairing can be rapid, or it may require many months of interaction (Nesbitt and Wenner 1987). Unison Calling and dancing are particularly important in the development of pair bonds.

Although young pairs often sever ties at the end of a breeding season (Drewien 1973; Nesbitt and Wenner 1987), established pairs return to the same breeding territory each year and defend it vigorously. Unison Calls and chases are particularly frequent during the several weeks before eggs are laid. Territory size is extremely variable, ranging from a few to several hundred hectares, with territory size roughly proportional to the openness of the landscape (Johnsgard 1983).

Breeding Season

The crane breeding season is either associated with distinct seasonality in the higher latitudes or with the wet/rainy season in lower latitudes. For species that breed in arctic to north temperate regions (Siberian, Lesser Sandhill, Hooded, and Whooping Cranes), spring is so short that renesting is seldom possible. Mid-latitude breeders, however, frequently renest if the first attempt fails. Cranes breeding in tropical and subtropical regions (Crowned, Blue, and Sarus Cranes, and Brolgas) usually breed on seasonal wetlands created during the rainy season (Archibald and Swengel 1987; Konrad 1987). Crowned Cranes can nest in any month depending on the rains (Walkinshaw 1964; Brown and Britton 1980; Pomeroy 1980), Blue Cranes usually nest at the beginning of South Africa's rainy season in November or December, and Sarus Cranes nest during southern Asia's July to October monsoons. Brolgas in northern Australia breed during the January to March rainy season, while those in the south begin nesting in spring (September to October) (Walkinshaw 1973). Most of the Wattled Cranes in southern Africa breed during winter, from May to October (Konrad 1981; Johnsgard 1983; Tarboton 1984) and at the end of the dry season, although they may breed at any time of year in Natal (Cyrus and Robson 1980; Tarboton 1984).

Nests, Eggs, and Chicks

Grassland nesters (Demoiselle and Blue Cranes, and sometimes Brolgas) usually lay their eggs on the bare ground with a nest composed of only a few twigs or pebbles (Van Ee 1966; Winter 1991); most other cranes build a **low platform nest** (Fig. 1.18) in shallow water. Water depth and the nest size are closely related: the deeper the water, the larger the nest. During flooding, cranes rapidly add material to the nest to keep the eggs above water. Wattled Crane pairs will not breed if their wetland territory lacks a small shallow pond for



Fig. 1.18 *Sandhill Crane nest in Florida.*

Photo Gene Knoder

nesting, but if a small area of open water is created, they sometimes nest immediately (Johnson and Barnes 1991).

Crane **clutch size** varies from two to three eggs for Crowned Cranes, two eggs for most other species, and usually one egg for Wattled Cranes. Eggs of Crowned Cranes are a light bluish white. Sarus, Brolgas, and some Red-crowned Cranes have plain white eggs with a few speckles of green or gray. Eggs for other cranes are heavily spotted with a light to dark brown background. Although there is remarkable variation between species, crane eggs from hot climates usually have less pigmentation than those in cold climates.

Both sexes assist in incubation, and the female usually incubates at night (Walkinshaw 1965). Incubation exchanges take place several times during the day and are sometimes accompanied by Unison Calling (Voss 1976). These vocalizations can facilitate humans finding birds or nests. The incubation period varies from 29 days in Demoiselle and Siberian Cranes to as many as 34 days in Wattled Cranes (see Table 4.1). Except for Wattled Cranes, which abandon the second egg (rarely laid) after the first chick has hatched, most cranes incubate until all live eggs have hatched. If the eggs are infertile or added, cranes will sometimes incubate 30–50 days beyond projected hatch dates (Walkinshaw 1965).

Some Crowned Crane clutches hatch synchronously (Walkinshaw 1964), but there is a one-to-two-day interval between the hatching of chicks in most other species. Sibling rivalry is important in determining chick survival. One chick is usually dominant over its sibling, and the dominant chick gets most of the food from the parents. Fighting between chicks is somehow linked to hunger. If food is scarce, the subordinate chick usually perishes. Sibling aggression has been observed in Greater Sandhill Cranes (Littlefield and

Ryder 1968; Drewien 1973), but is less pronounced in Black-necked (Lietal. 1991), Florida Sandhill (Layne 1982), and Demoiselle Cranes, although chicks of the latter species frequently compete for parental feedings (Winter 1991). Sibling aggression is so severe in Siberian Cranes that there are few reports of a pair rearing two chicks. Because of aggression even when food is available *ad libitum*, it is very difficult to captive-rear Whooping Crane chicks in groups. Two wild chicks are sometimes reared with each adult leading a chick on separate, but nearby, paths.

Species nesting in ephemeral wetlands (Crowned and Demoiselle Cranes) or in the Arctic where the nesting season is brief (Siberian, Lesser Sandhill Cranes) have shorter pre-fledging periods than species that inhabit permanent wetlands (Wattled Cranes) or regions with longer growing seasons. Immature cranes remain with their parents for 8–10 months until the onset of the next breeding season (Alonso et al. 1984). An abrupt change in the chick's voice from high frequency "peep-like" calls to the loud deeper voice of the adult coincides with the period during which the chick either leaves its parents of its own volition or is driven off (Nesbitt 1975; Nesbitt and Archibald 1981; Alonso et al. 1984).

After leaving their parents, young cranes gather in flocks with other non-breeders and move to foraging and roosting sites where they remain while the adults breed elsewhere (Kuyt 1979). Later they are joined by unsuccessful mated pairs and eventually by family groups. At approximately 18 months of age, a young crane exhibits **adult-like social behavior** including well-developed epigamic sign stimuli (e.g., red crown or fully grown wattles), threat displays, Guard and Unison Calls, and dancing (Bishop 1984; Nesbitt and Wenner 1987). Pairing can occur from this time onward.

Cranes in Captivity

Since ancient times, people have been fascinated by cranes and have kept them in captivity (Derrickson and Carpenter 1987). Cranes are depicted on the temple walls of the Egyptians (Whymper 1909), and cranes were raised by Chinese royalty more than 2,200 years ago (Cheng 1981). Continuing in the tradition of their ancestors, Africans today take wild chicks, raise them, and keep them as pets. In Australia, hand-raised Brolgas, popular pets, are sometimes called "Native

Companions." Thousands of Demoiselle and Eurasian Cranes are trapped during migration through the passes of the Hindu Kush mountains of Pakistan; many are eaten, others are sold as pets (Fig. 1.16). Cranes have always been popular in zoos in Europe and in the Orient (Johnsgard 1983:51).

Japan's "Mr. Zoo," Dr. Tadamichi Koga, was the first to treat the captive management of cranes in a scientific manner. Prior to the Second World War, Japanese zoos imported wild cranes from the mainland. During the war most of the zoo animals perished, and after the war importation was no longer possible. Unless cranes could be induced to breed in captivity, there would soon be no cranes for Japanese zoos. Dr. Koga noticed that if cranes lose their eggs, they rapidly re-nest, and by collecting and then artificially incubating the eggs, several clutches could be produced from a single pair (Koga 1961, 1976). Resulting chicks were hand-raised and then distributed to zoos throughout Japan.

About the same time, Dayton Hyde (1957) noted that cranes usually lay two eggs but rarely raise two young. He suggested that **a captive Whooping Crane flock could be established** without detriment to the wild population **by removing one egg** from each clutch. Using this reasoning, about 400 Whooping Crane eggs have been removed from the Wood Buffalo population in Canada from 1967 to the present. Productivity data, before and during this era, suggest that this egg harvest may have actually increased the number of chicks fledged each fall in Canada (Kuyt 1987; F. G. Cooch, Migratory Birds Branch, Ottawa, Ontario, Canada, personal communication).

Following Dr. Koga's example, several **crane propagation centers** have been established in recent decades. In 1966, the U.S. Fish and Wildlife Service, in cooperation with the Canadian Wildlife Service, established a captive breeding center for Whooping Cranes at Patuxent in Maryland. Patuxent subsequently, and most effectively, applied captive propagation to the conservation of the endangered Mississippi Sandhill Crane. Following the example of Patuxent, a private organization, the International Crane Foundation, was established in Baraboo, Wisconsin in 1973 with the intention of helping all 15 species of cranes. In 1979, the Soviets established a breeding center for Siberian Cranes near Moscow at the Oka State Nature Reserve, and in 1984, the Royal Forest Department of Thailand set up a center near Bangphra for the captive management of Eastern

Sarus Cranes. Other centers, notably the Baltimore Zoo, Beijing Zoo, Kushiro Crane Park in Japan, the London Zoological Society, the National Zoo in Washington, D.C., the Bronx Zoo in New York City, Tama Zoo in Japan, and Vogelpark Walsrode in Germany, have all made significant contributions to the captive management of cranes. The addresses of many other institutions with crane colonies can be obtained through ISIS (International Species Information System; see Chapter 10).

Some **longevity** records of captive cranes are remarkable. A male Siberian Crane (Fig. 1.19) that died from an injury in 1988 was captured, presumably as an adult, early in the 20th century. He had survived the two World Wars by residing at a zoo in Switzerland, and finally, in his late 70's, he fathered chicks at ICF through artificial insemination. In the studbook of the White-naped Crane (Sheppard 1990), reference is made to longevity records of more than 67 years and more than 64 years with breeding of birds over 60. Other remarkable records include a wild trapped female Siberian Crane which survived 61 years and 9 months at the Philadelphia Zoo (Davis 1969), a Wattled Crane at the New York Zoological Society that produced eggs over a 33-year period (Conway and Hamer 1977), and a Eurasian Crane which lived in a zoo for almost 43 years (Mitchell 1911).

Longevity records for wild cranes are unknown because marking individual cranes for identification purposes did not begin until recently. Because life in the wild is more hazardous, it is unlikely that wild cranes survive as long as their captive counterparts.

Status and Conservation

Because most cranes are highly visible at great distances and vulnerable to the loss and degradation of their wetland and grassland habitats, populations of most species have been reduced to a small fraction of their former numbers (Table 1.3) (Archibald and Meine 1995; Meine and Archibald *In prep.*). Seven of the fifteen species are considered threatened at the species level, while several additional subspecies are also at risk of extinction. It is no surprise that the four white species (Siberian, Red-crowned, Black-necked, and Whooping Cranes) are the most endangered. These species are not only the most easily seen, and thus shot, but they are also the most dependent upon aquatic habitats.



Fig. 1.19 Wolf, a 70+ year old Siberian Crane at ICF

Photo Lynn M. Stone

The Whooping Crane has staged a remarkable (although incomplete) recovery. Birds in the migratory population have come back from a low point of about 15 or 16 birds in 1941 to 133 cranes during the winter of 1992–93. These cranes breed in the vicinity of Great Slave Lake in northwestern Canada, and winter 2,500 miles away on the coast of Texas. There are also a few wild Whooping Cranes in an experimental flock (Fig. 1.20) in the western United States. These are all that remain from 289 eggs cross-fostered to Sandhill Cranes beginning in 1975. High chick mortality, disease, collisions with powerlines, and sexual imprinting on Sandhill Cranes have led to the discontinuation of the effort. Eggs for this flock, which peaked at about 35 birds in 1985, were produced in captivity at Patuxent (73 eggs) and collected from the wild cranes in Canada (216 eggs from 1975–1983; Ellis et al. 1992). Since 1985, one viable fertilized egg has been moved from nests where two viable eggs were present. These “second eggs” were placed in nests where all eggs failed to show signs of life (Lewis 1986). Eggs removed from the latter category were then collected. Some of these, however, proved to be fertile and were hatched at the captive breeding centers. In addition to the wild birds, there are now over 120 Whooping Cranes in captivity. Nearly all of these birds are at Patuxent or ICF, with a third captive breeding center recently established at the Calgary Zoo in Canada. There are also about 50 wild birds in a second experimental population in the Kissimmee Prairie in Florida where since 1993 captive-reared cranes have been released into a non-migratory setting.

There are two geographically isolated populations of Red-crowned Cranes: a group of 600–650 cranes in southeastern Hokkaido, Japan, with several more on

TABLE 1.3

Approximate size of crane populations.¹

Species or Subspecies	Wild	Captivity	Status ²
Black Crowned Crane	70,000	450	Threatened
Gray Crowned Crane	90,000	1,200	Non-endangered
Wattled Crane	14,000	172	Threatened
Blue Crane	21,000	1,000	Threatened
Demoiselle Crane	250,000	1,000	Non-endangered
Siberian Crane	3,000	115	Endangered
Sandhill Crane (all races)	700,000	500	Non-endangered
Cuban Sandhill Crane	150	?	Endangered
Mississippi Sandhill Crane	120	40	Endangered
White-naped Crane	5,000	400	Endangered
Sarus Crane (all races)	20,000	350	Non-endangered
Eastern Sarus Crane	1,000	50	Endangered
Brolga	25,000	33	Non-endangered
Eurasian Crane (all races)	225,000	280	Non-endangered
Hooded Crane	10,000	100	Endangered
Black-necked Crane	5,800	90	Endangered
Red-crowned Crane	1,800	750	Endangered
Whooping Crane	170	120	Endangered

¹ Approximate size of world populations, 1995.

² Status: Endangered, likely to become extirpated in the wild during the next century if present population trends continue; Threatened, threatened with eventual extirpation in the wild; Non-endangered, populations generally stable or declining only in a portion of their range.

the neighboring Kurile Islands, now part of Russia, and a population of perhaps 1,000 birds on mainland Asia (Feng and Li 1985; Masatomi et al. 1989; Anonymous 1991). The island population migrates locally from the marshes to several artificial feeding stations near the city of Kushiro. Aided by feeding programs initiated by the local people and now supported by the government, this population has grown to its present size from about 30 birds in 1952. The mainland flock migrates to the Korean peninsula and to coastal wetlands of China just north of the mouth of the Yangtze River. The wetlands where these cranes breed in northern China, southeastern Siberia, and Japan are valuable for agricultural development (Archibald 1987). Wetland loss is the major limiting factor for the species. Japan's first wetland national park, Kushiro Marsh National Park, and one of China's first protected areas, Zhalong Nature Reserve, have been established to protect major nesting areas of



Fig 1.20 *Gray's Lake, Idaho, where Whooping Crane eggs were cross-fostered to Sandhill Cranes.*

Photo Scott R. Derrickson

these cranes. Red-crowned Cranes are popular exhibit birds in zoos, and they breed readily in captivity.

Siberian Cranes breed in the Arctic of both eastern and western Russia (Fig. 1.21) and winter in Iran (ca 10 birds), India (5-10 birds), and China (ca 3000 birds: K. Ozaki, Yamashina Institute for Ornithology, Abiko City, Japan, personal communication). They are exclusively dependent on wetlands for their breeding and their wintering grounds. Hunting continues to threaten the survival of the remnant flock that migrates through heavily hunted regions of Afghanistan and Pakistan. Loss of wetlands on the wintering grounds and migration staging areas has undoubtedly contributed to the decline of this species. A proposed dam across the Yangtze River poses a threat to the wintering grounds of the majority of Siberian Cranes. With difficulty, Siberian Cranes have been induced to reproduce in captivity at ICF in the United States, at the Oka State Nature Reserve in Russia, at Beijing Zoo in China, and at Vogelpark Walsrode in Germany.

Black-necked Cranes, believed to number about 5,800, breed in freshwater wetlands scattered across the Tibetan Plateau. In winter they migrate to slightly lower elevations in southern Tibet, Yunnan and Guizhou Provinces of China, and several valleys in Bhutan. This species has declined due to hunting on the breeding and wintering areas in China in recent decades and the loss of barley fields and wetlands in which the cranes forage in winter. Several pairs of captive Black-necked Cranes breed at Beijing and Xining zoos in China, and single pairs breed at Vogelpark Walsrode in Germany and at ICF in the United States.

The continuing increase in human numbers, particularly in southern Asia and throughout most of Africa, increasingly threatens the wetlands and grasslands needed by cranes (Archibald and Mirande 1985). But humans can also improve the chances for the survival of cranes through habitat protection, education, and reintroduction. Husbandry will play a central role in this broad conservation agenda. If proper husbandry and genetic management practices are followed, captive breeding can perhaps indefinitely maintain viable populations of each crane species and provide birds for reintroduction efforts. During the past two decades, Patuxent, ICF, Oka State Nature Reserve, Beijing Zoo, Bronx Zoo, and other zoos have developed techniques for the successful management of cranes in captivity. Much of that valuable information is presented in this volume.



Fig 1.21 *Siberian Crane marshes in western Siberia.*

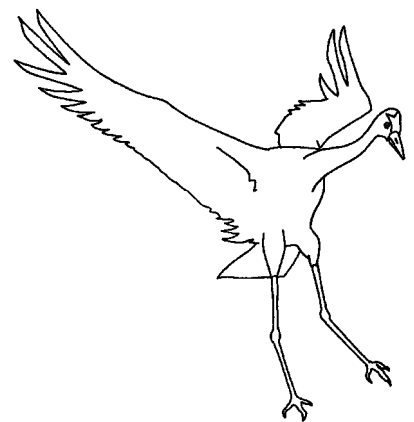
Photo David H. Ellis

Literature Cited

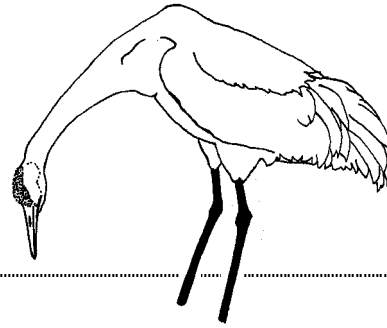
- Allan, D. G. 1995. [Blue Crane biology]. In R. D. Bielfuss, editor. Proceedings of the African Crane and Wetland Training Workshop. International Crane Foundation, Baraboo, Wis. *In press*
- Allen, R. P. 1952. The Whooping Crane. Research Report No. 3, National Audubon Society, New York. 246 pp.
- Allen, R. P. 1956. A report on the Whooping Crane's northern breeding grounds. National Audubon Society, New York. 60 pp.
- Alonso, J. A., J. P. Veiga, and J. C. Alonso. 1984. Familienaufklärung und Abzug aus dem Winterquartier beim Kranich *Grus grus* Journal für Ornithologie 125:69-74. [In German with English summary. English translation available from ICF.]
- Anonymous. 1991. Winter counts of endangered cranes. ICF Bugle 18(4):8.
- Archibald, G. W. 1976a. The unison call of cranes as a useful taxonomic tool. Ph.D. dissertation, Cornell University, Ithaca, New York. 167 pp.
- Archibald, G. W. 1976b. Crane taxonomy as revealed by the unison call. Pages 225-251 in J. C. Lewis, editor. Proceedings International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Archibald, G. W., and C. Meine. 1995. Family Gruidae. In J. del Hoyo and A. Elliot, editors. Handbook of the birds of the world. Vol. 2. Lynx Edicions, Barcelona.
- Archibald, G. W., and C. M. Mirande. 1985. Population status and management efforts for endangered cranes. Pages 586-602 in Proceedings 50th North American Wildlife Natural Resources Conference.
- Archibald, G. W., and S. R. Swengel. 1987. Comparative ecology and behavior of Eastern Sarus Cranes and Brolgas in Australia. Pages 107-116 in J. C. Lewis, editor. Proceedings 1985 Crane Workshop. Platte River Whooping Crane

- Habitat Maintenance Trust and U.S. Fish and Wildlife Service, Grand Island, Nebr.
- Archibald, K. 1987. The conservation status of the breeding ground of the Red-crowned Crane in Hokkaido, Japan. Pages 63-86 *in* G. W. Archibald and R. R. Pasquier, editors. Proceedings 1983 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Bennett, A. J. 1978. Ecology and status of Greater Sandhill Cranes in southeastern Wisconsin. M.S. thesis, University of Wisconsin, Stevens Point. 110 pp.
- Bishop, M. A. 1984. The dynamics of subadult flocks of Whooping Cranes wintering in Texas, 1978-79 through 1982-83. M.S. thesis, Texas A&M University, College Station. 127 pp.
- Bishop, M. A. 1994. [Black-necked Crane biology]. Unpublished reports at International Crane Foundation, Baraboo, Wis.
- Blaauw, F. E. 1897. A monograph of the cranes. E. J. Brill, London, England and Leiden, Netherlands. 69 pp.
- Brodkorb, P. 1967. Catalogue of fossil birds. Bulletin of the Florida State Museum 11:145-153.
- Brown, L. H., and P. L. Britton. 1980. The breeding seasons of East African birds. East Africa Natural History Society, Nairobi, Kenya.
- Cheng, Tso-hsin. 1981. Cranes of China. Pages 47-48 *in* J. C. Lewis and H. Masatomi, editors. Crane research around the world. International Crane Foundation, Baraboo, Wis.
- Conway, W., and A. Hamer. 1977. A 36-year laying record of a Wattled Crane at New York Zoological Park. *Auk* 94:786-787.
- Cyrus, D., and N. Robson. 1980. Bird atlas of Natal. Natal University Press, Pietermaritzburg, South Africa. 320 pp.
- Davis, M. 1969. Siberian Crane longevity. *Auk* 86:347.
- Derrickson, S. R., and J. W. Carpenter. 1987. Behavioral management of captive cranes—factors influencing propagation and reintroduction. Pages 493-511 *in* G. W. Archibald and R. F. Pasquier, editors. Proceedings of the 1983 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Doughty, R. W. 1989. The return of the Whooping Crane. University of Texas Press, Austin. 182 pp.
- Drewien, R. C. 1973. Ecology of Rocky Mountain Greater Sandhill Cranes. Ph.D. dissertation, University of Idaho, Moscow. 82 pp.
- Ellis, D. H., J. C. Lewis, G. F. Gee, and D. G. Smith. 1992. Population recovery of the Whooping Crane with emphasis on reintroduction efforts: past and future. Proceedings North American Crane Workshop 6:142-150.
- Ellis, D. H., S. R. Swengel, G. W. Archibald, and C. B. Kepler. *In prep.* A sociogram for the cranes of the world.
- Feng Ke-min and Li Jin-lu. 1985. Aerial surveys on the Red-crowned Cranes (*Grus japonensis*) and other rare water birds. Pages 17-34 *in* H. Masatomi, editor. International spring censuses on *Grus japonensis* (Red-crowned Crane) in 1984. Wild Bird Society of Japan, Tokyo.
- Gichuki, N. 1995. [Grey Crowned Crane biology]. *In* R. D. Bielfuss, editor. Proceedings of the African Crane and Wetland Training Workshop. International Crane Foundation, Baraboo, Wis. *In press*
- Herter, D. R. 1982. Staging of Sandhill Cranes on the eastern Copper River Delta, Alaska. Pages 273-280 *in* J. C. Lewis, editor. Proceedings 1981 International Crane Workshop. National Audubon Society, Tavernier, Florida.
- Hunt, H., and R. Slack. 1987. Winter foods of the Whooping Crane based on stomach content analyses. Pages 217-218 *in* J. C. Lewis, editor. Proceedings 1985 Crane Workshop. Platte River Whooping Crane Habitat Maintenance Trust, Grand Island, Nebr.
- Hyde, D. O. 1957. Crane notes. *Blue Jay* 15:19-21.
- Ingold, J. L. 1984. Systematics and evolution of the cranes (Aves: Gruidae). Ph.D. dissertation, Miami University, Oxford, Ohio. 62 pp.
- Iverson, G. C., T. C. Tacha, and P. A. Vohs. 1982. Food contents of Sandhill Cranes during winter and spring. Pages 95-98 *in* J. C. Lewis, editor. Proceedings 1981 International Crane Workshop. National Audubon Society, Tavernier, Fla.
- Johnsgard, P. A. 1983. Cranes of the world. Indiana University Press, Bloomington. 257 pp.
- Johnson, D. N., and P. R. Barnes. 1991. The breeding biology of Wattled Cranes in Natal. Pages 377-386 *in* J. T. Harris, editor. Proceedings 1987 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Koga, T. 1961. Studies on the reproduction of cranes, especially on their artificial incubation and breeding. Journal of the Japanese Association of Zoological Gardens and Aquariums 3(3):51-58. [In Japanese with English summary.]
- Koga, T. 1976. Increasing captive production of Japanese and White-naped Cranes. Pages 351-355 *in* J. C. Lewis, editor. Proceedings International Crane Workshop. Oklahoma State University Printing, Stillwater.
- Konrad, P. M. 1981. Status and ecology of Wattled Crane in Africa. Pages 220-237 *in* J. C. Lewis and H. Masatomi, editors. Crane research around the world. International Crane Foundation, Baraboo, Wis.
- Konrad, P. M. 1987. Rainy season ecology of South African Grey Crowned Cranes in the Luangwa Valley, Zambia. Pages 337-344 *in* G. W. Archibald and R. R. Pasquier, editors. Proceedings 1983 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Krajewski, C. 1988. Phylogenetic relationships among cranes (Aves: Gruidae) based on DNA hybridization. Ph.D. dissertation, University of Wisconsin, Madison. 342 pp.
- Kuyt, E. 1979. Banding of juvenile Whooping Cranes and discovery of the summer habitat used by nonbreeders. Pages 109-111 *in* J. C. Lewis, editor. Proceedings 1978 Crane Workshop. Colorado State University Printing Service, Fort Collins.
- Kuyt, E. 1987. Management and research of Whooping Cranes, 1965-1982. Pages 365-1982 *in* G. W. Archibald and R. F. Pasquier, editors. Proceedings of the 1983 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Kuyt, E., and J. P. Goossen. 1987. Survival, age composition, sex ratio, and age at first breeding of Whooping Cranes in Wood Buffalo National Park, Canada. Pages 230-244 *in* J. C. Lewis, editor. Proceedings 1985 Crane Workshop. Platte River Whooping Crane Habitat Maintenance Trust and U.S. Fish and Wildlife Service, Grand Island, Nebr.

- Layne, J. N. 1982. Status of sibling aggression in Florida Sandhill Cranes. *Journal of Field Ornithology* 53:272-274.
- Lewis, J. C. 1974. Ecology of the Sandhill Crane in the south-eastern Central Flyway. Ph.D. dissertation, Oklahoma State University, Stillwater. 213 pp.
- Lewis, J. C. 1986. The Whooping Crane. Pages 659-676 in R. C. DiSilvestro, editor. Audubon Wildlife Report 1986. National Audubon Society, New York.
- Li Dehao, Zhou Zhijun, We Zhikang, Li Zhumei, and Wang Youhui. 1991. On the structure and behavior of the breeding population of Black-necked Cranes in Songpan Meadow of Sichuan Province. Pages 83-87 in J. Harris, editor. Proceedings 1987 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Littlefield, C. D. and R. A. Ryder. 1968. Breeding biology of the Greater Sandhill Crane on Malheur National Wildlife Refuge, Oregon. *Transactions of the North American Wildlife and Natural Resources Conference* 33:444-454.
- Makatsch, W. 1970. Der Kranich. Die Neve Brehn Bucherei 229:28-81. Wittenberg, Germany.
- Masatomi, H. 1970-1974. Ecological studies on the Japanese Crane, *Grus japonensis*. *Journal of Bibai Agricultural Engineering College* Part I (1970) 1:37-45; Part II (1971) 2:93-111; Part III (1972a) 3:153-161; Part IV (1972b) 4:139-152; Part V (1974) 5:1-16.
- Masatomi, H., and T. Kitagawa. 1975. Bionomics and sociology of Tancho or the Japanese Crane, *Grus japonensis*. II Ethogram. *Journal of the Faculty of Science, Hokkaido University, Series VI, Zoology* 19:834-878.
- Masatomi, H., K. Momose, and M. Takeshita. 1989. Wintering population of the tancho *Grus japonensis* in Hokkaido, 1987-'88. *Journal of Environmental Science Laboratory, Senshu University-Hokkaido* 1:73-82.
- McNulty, F. 1966. The Whooping Crane. E. P. Dutton and Co., New York. 190 pp.
- Meine, C., and G. W. Archibald. *In prep.* Action plan for the conservation of cranes. IUCN, Gland, Switzerland.
- Mitchell, P. C. 1911. On longevity and relative viability in mammals and birds, with a note on the theory of longevity. *Proceedings of the Zoological Society of London*, pages 425-548.
- Mullins, W. H., and E. G. Bizeau. 1978. Summer foods of Sandhill Cranes in Idaho. *Auk* 95:175-178.
- Nesbitt, S. A. 1975. Voice maturity in Sandhill Cranes. *Florida Field Naturalist* 3:19.
- Nesbitt, S. A. 1989. The significance of mate loss in Florida Sandhill Cranes. *Wilson Bulletin* 101:648-651.
- Nesbitt, S. A. 1992. First reproductive success and individual productivity in Sandhill Cranes. *Journal of Wildlife Management* 56:573-577.
- Nesbitt, S. A., and G. W. Archibald. 1981. The agonistic repertoire of Sandhill Cranes. *Wilson Bulletin* 93:99-103.
- Nesbitt, S. A., and A. S. Wenner. 1987. Pair formation and mate fidelity in Sandhill Cranes. Pages 117-122 in J. C. Lewis, editor. Proceedings 1985 Crane Workshop. Platte River Whooping Crane Habitat Maintenance Trust and U.S. Fish and Wildlife Service, Grand Island, Nebr.
- Novakowski, N. W. 1966. Whooping Crane population dynamics on the nesting grounds, Wood Buffalo National Park, Northwestern Territories, Canada. Canadian Wildlife Service, Research Report Series 1. 20 pp.
- Peters, J. C. 1934. Check-list of the birds of the world. Vol. II. Harvard University Press, Cambridge, Mass. 401 pp.
- Pomeroy, B. S. 1980. Aspects of the ecology of Crowned Cranes *Balearica regulorum* in Uganda. *Scopus* 4:29-35.
- Prange, H. D., J. S. Wasser, A. S. Gaunt, and S. L. L. Gaunt. 1985. Respiratory responses to acute heat stress in cranes: the effects of tracheal coiling. *Respiratory Physiology* 62:95-103.
- Radke, M. F., and W. R. Radke. 1986. Breeding by a two-year old Sandhill Crane. *Western Birds* 17:192-193.
- Sauey, R. T. 1985. The range, status, and winter ecology of the Siberian Crane *Grus leucogeranus*. Ph.D. dissertation, Cornell University, Ithaca, New York. 428 pp.
- Sheppard, C. 1990. International Studbook of the White-naped Crane, *Grus vipio*, as of December 1989. New York Zoological Society, New York.
- Stephenson, J. D. 1971. Plumage development and growth of young Whooping Cranes. M.S. thesis, Oregon State University, Corvallis. 56 pp.
- Tacha, T. C. 1988. Social organization of Sandhill Cranes from mid-continental North America. *Wildlife Monograph* 99:1-37.
- Tarboton, W. R. 1984. The status and conservation of the Wattled Crane in the Transvaal. Pages 665-679 in J. Ledger, editor. Proceedings of the Fifth Pan African Ornithological Congress. South African Ornithological Society, Johannesburg.
- Taverner, P. A. 1929. The red plumage coloration of the Little Brown and Sand-Hill Cranes, *Grus canadensis* and *mexicanus*. *Auk* 46:228-230.
- Uhler, F. M., and L. N. Locke. 1969. A note on the stomach contents of two Whooping Cranes. *Condor* 52:216.
- Van Ee, C. A. 1966. Notes on the breeding behaviour of the Blue Crane, *Tetraptyx paradisea*. *Ostrich* 37:23-29.
- Voss, K. 1976. Behavior of the Greater Sandhill Crane. M.S. thesis, University of Wisconsin, Madison. 137 pp.
- Walkinshaw, L. H. 1949. The Sandhill Cranes. *Cranbrook Institute Science Bulletin* 29, Bloomfield Hills, Mich. 202 pp.
- Walkinshaw, L. H. 1964. The African Crowned Cranes. *Wilson Bulletin* 76:355-377.
- Walkinshaw, L. H. 1965. One hundred thirty-three Sandhill Crane nests. *Jack-Pine Warbler* 43:136-143.
- Walkinshaw, L. H. 1973. Cranes of the world. Winchester Press, New York. 370 pp.
- Whymper, C. 1909. Egyptian birds for the most part seen in the Nile Valley. A. and C. Black, London. 221 pp.
- Winter, S. W. 1991. The Demoiselle Crane in the agricultural landscape of the Ukrainian steppe zone. Pages 285-294 in J. Harris, editor. Proceedings 1987 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Wood, D. S. 1979. Phenetic relationships within the family Gruidae. *Wilson Bulletin* 91:384-399.



General Husbandry



Scott R. Swengel and James W. Carpenter

Captive cranes need a clean, safe, low stress environment to remain healthy and to breed. Because disturbances are stressful to cranes (Mirande et al. 1988 unpubl.), schedule and perform husbandry practices to minimize disturbance (see Chapter 6 for details). Frequent observation (Fig. 2.1) of confined cranes enables quick detection of changes in a bird's behavior (see Chapter 6). By understanding the behavior of cranes, managers can choose appropriate husbandry practices.

Animal Welfare

Humane treatment of captive animals is an important part of the conservation ethic. The **Animal Welfare Act** (and USDA regulations 9 CFR Parts 1-3, as amended in Federal Register 1989) was enacted by the U.S. Congress to govern the use of animals in government-funded research projects. The USDA regulations currently exclude birds, but have been adapted for birds by some agencies. We recommend voluntary compliance with these guidelines.



Fig. 2.1. Early each day a caretaker, here Jane Nicolich, Patuxent's flock manager, walks through the colony to assess the health of each bird.

Photo David H. Ellis

An important requirement is that each institution create an **Animal Care and Use Committee (ACUC)**. Included are a veterinarian and a person who is not employed by the organization doing the research. The ACUC reviews proposed research projects and evaluates facilities. The January 1987 issue of **Laboratory Animal Science** is devoted to the effective use of an ACUC and provides more details on how these committees can operate. Other animal welfare guidelines are provided by the Canadian Council on Animal Care (1984) and the American Ornithologists' Union (1988).

The Physical Environment

Crane Pens

Crane pens should be large enough to prevent microorganisms and parasites from building up in the soil or in shelters. Normally, cranes are moved to a fallow pen each year (see below). Minimum pen sizes are presented in Chapter 12 and in Carpenter and Derrickson (1987).

The pen walls should be designed to minimize injuries. Use fencing that is smooth and lacks projections. Visual barriers serve the dual purpose of making the fence smoother and reducing stress on the cranes. Cranes are more likely to breed when they have visual barriers isolating them from their neighbors (see Chapter 6). Soft outdoor and indoor pen substrates help keep crane feet healthy. Grass, or other natural outdoor surfaces, and wood shavings inside shelters are good choices. Cranes that are locked indoors require light and ventilation.

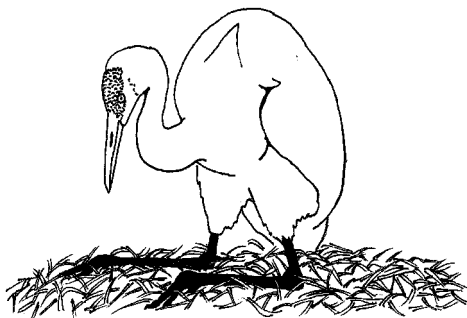
Cleaning, Sanitation, and Pen Rotation

Outdoor Pens. Clean pens are important to the continued health of cranes. Pen rotation is one of the best ways to keep large outdoor pens clean. Rotation

allows many of the soil pathogens to die by removing the crane host that is a critical link in the life cycle of the pathogen. If cranes are rotated to alternate pens annually and the pen has 50 m² of space per crane, the outdoor pens do not normally need to be cleaned. If, however, the soil has a high pathogen load or is known to harbor one very dangerous pathogen, it is advisable to disinfect the outdoor pen before reintroducing cranes. This can be accomplished by tilling the topsoil and applying lime, formalin, or a commercial disinfectant that is effective against the disease agent(s) in question. Cranes are moved to the fallow pen in mid-summer, in fall, or just prior to the onset of egg laying (so the chicks have a clean pen). All pairs in a row or colony should be moved on the same day so all pairs are separated by an empty pen.

Cross contamination of pens can be minimized by using an antibiotic (antibacterial and antiviral) **footbath**. A footbath is a shallow pan at least 40 cm in diameter containing 6-10 cm of fluid and located at the doorway to each pen complex. Caretakers dip the soles of their shoes each time they enter and leave the pen complex. The bath is changed weekly, or more often if the bath is diluted by rain. The bath solution can contain any of several agents including Broad Spec, Environ, or Nolvasan.

Indoor Pens. If the cranes have four-sided shelters with bedding, such as sand or wood shavings, clean the bedding at one or two day intervals. In pens with shavings on the floor, pick up the fecal material with a scoop or rubber glove. Sand floors can be cleaned by sifting the droppings through a 3-mm mesh screen. Sand, however, will occasionally get into chicks' eyes and cause conjunctivitis or other ocular lesions. Remove wet bedding during daily cleaning.



Wet bedding, especially wood shavings, promotes fungal and bacterial growth particularly during warm, wet weather. Chicks are more susceptible to *Aspergillus* and other pathogens than adult cranes. See Chapter 5 for details of cleaning chick pens.

At least annually, or more frequently depending on usage, indoor pens should be thoroughly cleaned. Remove the bedding and disinfect the floor and walls by spraying or wiping them with bleach or a commercial disinfectant diluted with water. Because disinfectants are potentially toxic to animals, they should be used judiciously; follow the directions on the label and never use a higher concentration than is recommended. Allow pens, especially the soil, to lie idle long enough for the chemical to be rendered harmless to the cranes (usually 1-2 days longer than the half life listed on the label). ICF has used Environ or Nolvasan mixed 1:250 with water and sprayed on soiled pen walls. These chemicals (see Appendix) were chosen because they have short half lives.

Change the bedding more often if the cranes are locked inside a shelter for extended periods. Put new bedding into the shelter after the building is thoroughly dry.

Wading Pools

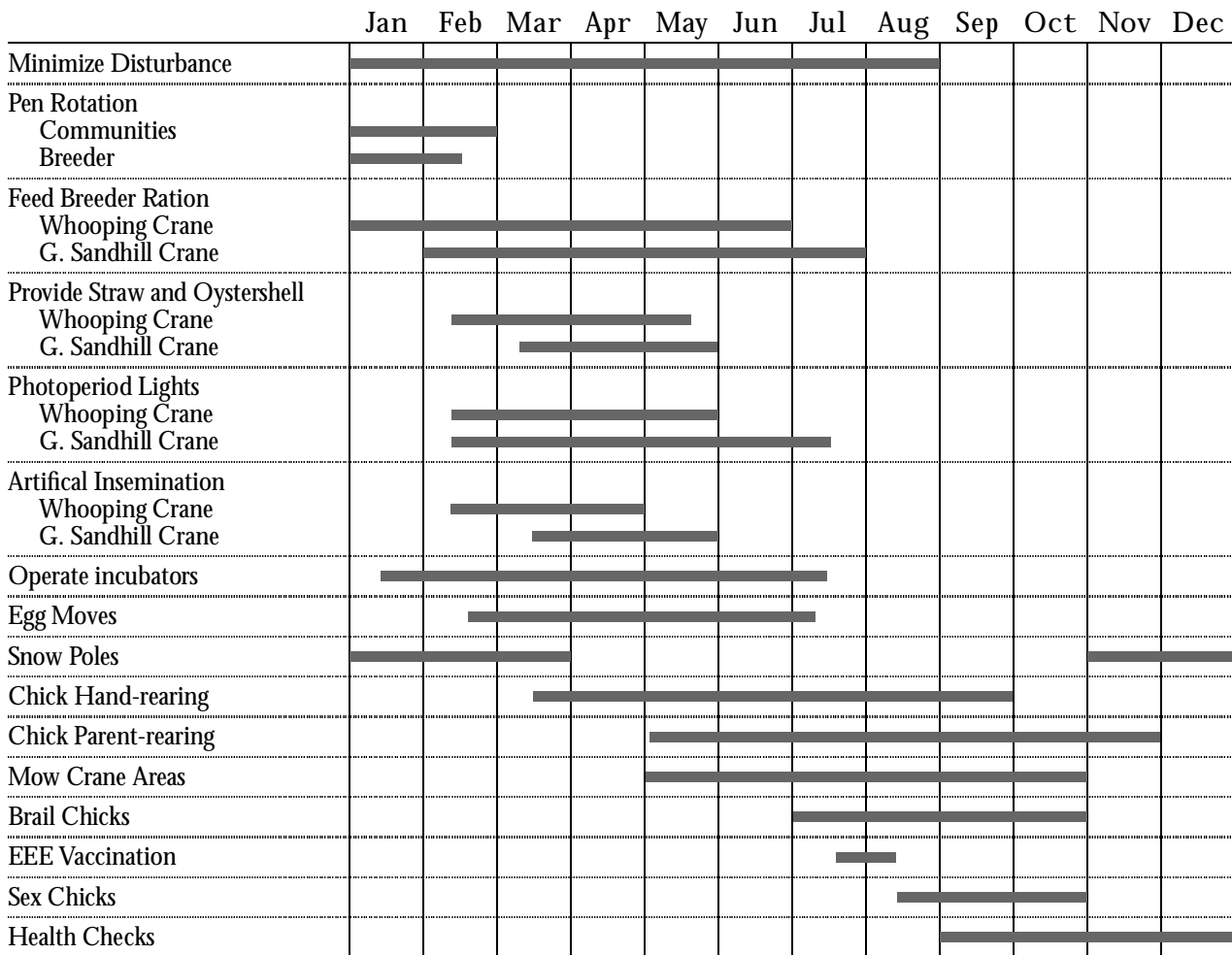
Shallow pools in which the cranes can wade and bathe can make the pen environment more natural and may promote breeding (see Chapter 3). Pools should either have a slow, continuous flow of water through them or be cleaned every 3-5 days (or more often if there is a chick in the pen). If the pool stagnates, deadly bacteria, such as *Clostridium botulinum*, may flourish. Other potential health hazards associated with wading pools are bacterial or parasitic infections through contamination by feces from the cranes, rodents, and wild birds. See Chapter 12 for details of pool construction and maintenance.

Annual Cycle of Management Activities

Most activities related to crane management are seasonal in nature or should otherwise be done on a regular schedule planned to minimize disturbance. Figure 2.2 is an example of a schedule for such activities.

FIG. 2.2.

Time line for maintenance activities.



Non-breeding Season

Most activities that are not directly related to breeding are best conducted in the non-breeding season when cranes are less susceptible to disturbance. Further, the non-breeding season is usually less busy for caretakers. This is, in some ways, the best time to move cranes between pens because cranes are less aggressive during this period and the move will not disrupt reproduction. Fall moves allow cranes two or more months to adjust before the next breeding season. **Annual health checks** (or physical examinations) should also be conducted on the cranes in the fall, generally in conjunction with wing clipping.

Winter

Temperate and subarctic cranes can tolerate temperatures of -30°C (-22°F) or colder as long as they have

shelter and food. All of these species require only a three-sided shelter (wind break). Subtropical cranes need supplemental heat or should be locked indoors when the temperature is below 0°C (32°F) (see Table 2.1). African Crowned Cranes should be kept inside during brief winter warm spells if the ground is still frozen. Other cranes may be allowed outside any time the temperature is higher than that listed in Table 2.1. On windy days these temperatures should be adjusted slightly upward to account for wind chill, but cranes usually seek shelter from the wind without assistance.

Unfortunately when cranes are locked inside, some may become stressed from the close contact with humans. All but the African Crowned Cranes may be let outside during brief cleaning periods unless they are overly stressed when herded into their house. Sliding doors that operate from outside the pen can make this operation easy, since most subtropical cranes will go back inside on their own within a few

TABLE 2.1.

Ambient temperatures at which warm-climate cranes should be moved indoors and supplied with supplemental heat.

Species	Locked Inside	Heat Needed
Black Crowned	0° C (32° F)	0° C (32° F)
Gray Crowned	0° C (32° F)	0° C (32° F)
Eastern Sarus	-5° C (23° F)	-15° C (5° F)
Brolga	-10° C (14° F)	-20° C (-4° F)
Indian Sarus	-20° C (-4° F)	-30° C (-22° F)
Blue	-20° C (-4° F)	-35° C (-31° F)
Wattled	-20° C (-4° F)	-40° C (-40° F)

minutes after their shelter is cleaned. If a crane attacks the keeper, either let the crane outside during pen servicing or wear protective gear when it is too cold to let the cranes out. Alternatively, one person can fend off the crane while another services the pen.

When wary cranes must be kept inside during pen servicing, stay low, approach indirectly, and avoid direct eye contact with the birds while inside the pen. It can help to provide a window in one side of the building to allow viewing of the crane without entering the pen. Additional translucent windows or skylights are useful for lighting the pens and providing solar heat during winter. Cranes stay healthier when their quarters are well-lit. See Chapter 12 for details of insulated buildings and heating systems.

Egg Laying Season

One to two months before cranes are expected to lay eggs, change to Breeder Diet (Table 2.2) and supply crushed oyster shell (mixed with pelleted food or in a separate container) as a calcium supplement. Visual barriers must also be in place prior to the breeding season. Place nesting material in the pen before the cranes lay eggs to stimulate nest building. It is also important to condition the males to artificial insemination (AI) before the females are expected to lay eggs so the males will produce semen at the appropriate time to fertilize the eggs.

Initiate egg searches (see Chapter 4) when the cranes are expected to lay eggs. Do this in a way that balances the disturbance incurred by the searches with the importance of finding eggs immediately. Egg searches can generally be done from outside the pen.

Some cranes may not lay eggs if caretakers regularly enter their pens to search for eggs.

Cranes that breed at high latitudes often breed better when they experience artificially lengthened days (see Chapter 3 for details). Begin to extend the photoperiod a month or so before the intended start of egg laying to ensure maximum physiological response. This helps to stimulate an early breeding season prior to the onset of hot weather which causes most cranes to stop laying.

Chick Rearing Season

Several activities are tied to the development of chicks. The chick-rearing house and its exercise pens should be repaired, cleaned, and disinfected before the expected hatch date of the first egg. Other major seasonal activities timed with chick development are sexing (Chapter 11C), flight restraint (Chapter 11E), and formation of release cohorts (Chapter 11D).

Food and Drinking Water

Crane Food

Diets. Crane diets were adapted from poultry diets (Serafin 1982). Cranes consume about 4% of their body weight per day (Halibey 1979 unpubl.). Commercial diets have made it more convenient and less expensive to feed a controlled diet to cranes (see Appendix).

There are usually three types of formulated crane diets (Tables 2.2 and 2.3). Adult cranes receive Maintenance or Breeder Diets depending on the season. Chicks are provided a Starter Diet. Most formulated crane diets are composed largely of vegetable matter and less than 10% animal matter. The Patuxent diet is 15.0% protein (Maintenance Diet) or 22.0% protein (Breeder Diet). The ICF diet is 19.4% and 20.5% protein for Maintenance and Breeder Diets. Patuxent and ICF Breeder Diets also have a higher calcium level (2.45%) than the Maintenance Diets (1.0%). Starter Diets for chicks have increased protein, calcium, and vitamin B levels (Tables 2.2 and 2.3). Chicks also need a higher calcium/phosphorus ratio in their food than non-breeding adult cranes, because of mineral demands for bone and feather growth. Begin feeding Breeder Diet two months before the anticipated egg laying season (Russman and Putnam 1980).

TABLE 2.2.

Feed formulas for chicks, non-breeding adults, and breeding adults.

	Starter	Maintenance	Breeder
Ground yellow corn	24.4%	38.8%	41.2%
Soybean meal (44% protein)	—	13.1%	15.0%
Soybean meal (49% protein)	31.5%	—	—
Wheat middlings	12.0%	12.6%	10.0%
Fish meal (60% protein)	—	4.0%	5.0%
Ground oats	11.5%	15.7%	7.5%
Meat and bone meal	—	5.2%	4.0%
Alfalfa meal (17% protein)	5.0%	5.2%	5.0%
Corn distillers solubles	3.0%	—	1.5%
Brewers dried yeast	2.5%	—	2.0%
Corn oil	3.3%	—	—
Dried whey	1.2%	3.2%	3.5%
Limestone	1.5%	0.5%	3.5%
Dicalcium phosphate	3.0%	0.5%	1.0%
Iodized salt	0.25%	0.5%	0.5%
Vitamin/mineral premix	0.5%	0.5%	0.5%

Composition of Formulated Diets

Percent protein	23.8%	19.4%	20.5%
Metabolizable energy, kcal/kg	2689	2530	2533
Percent calcium	1.4%	1.0%	2.45%
Percent phosphorus	0.90%	0.86%	0.89%
Percent methionine and cystine	0.7%	—	—
Percent Lysine	1.3%	—	—

The **type of protein** in a chick diet is very important. To minimize sulphur amino acids (cystine and methionine), Starter Diets (Table 2.2) should use **vegetable protein** only. Chicks that are provided Starter Diets containing high proportions of sulphur amino acids develop more leg and wing abnormalities than chicks that consume diets low in sulphur amino acids (Serafin 1982). Avoid feeding animal products, especially fish, on a daily basis because they contain more sulphur amino acids than most vegetable proteins.

Pellet Size. We recommend that crane feed, except for young chicks, should be pellets that are 5 mm in diameter and 6-15 mm long. Chicks less than 2-3 weeks old should be fed crumbles (2-5 mm diameter nuggets) and then gradually introduced to the larger pellets according to the schedule given in Chapter 5.

Food Storage. Feed should be stored at 1.7-4.4° C (35-40° F) with low humidity. It is very important that crane food be kept dry to eliminate mold and reduce bacterial growth. Storage areas should be clean and free of rodents and insects. Some ingredients in synthetic diets, especially vitamins, have a limited storage life (Carpenter 1979). If a refrigerator is not available, store no more than a one-month supply at ambient temperature; refrigerated food can be held up to three months. Feed can be frozen for up to one year, but it will lose some of its nutritional value, may become easier to pulverize, and may acquire odors or tastes that make it less palatable. Water condenses on feed bags removed from a freezer during warm, humid weather, so allow the bags to stand separately and dry.

TABLE 2.3.

Vitamin/mineral premix for feed formulas¹

	Starter	Breeder and Maintenance
Choline chloride 60%	40%	40%
DL-Methionine	13%	13%
Vitamin E 227	7%	
Niacin 99.5%	7%	7%
Calcium pantothenate 160	1.1%	1.1%
Vitamin B ₁₂ 300	0.5%	0.5%
Riboflavin 100	0.5%	0.5%
Vitamin A 650	0.25%	0.25%
Vitamin D ₃ 400	0.1%	0.1%
Selenium 0.8%	27%	27%
Zinc oxide 72%	3.0%	3%
Manganese oxide 60	4.5%	4.5%
E.D.D.I. 79.5%	0.001%	0.001%
Biotin 1%	0.4%	0
Folic acid 10%	0.1%	0

¹ *ICF custom premix. Commercial pre-mixes for turkeys or chickens are also used with manufacturer's inclusion rates followed.*

Crane Feeding

Place the food in a hopper feeder or an elevated bucket to reduce its accessibility to vermin and to facilitate removal of spilled food. Place the feeder in a shelter to shield it from rain, snow, and sunlight. To further limit water contact, keep the feeder at least 1 m from the water supply.

Frequency of Feeding. In warm, humid climates, it is necessary to change the food daily. In temperate climates, check the food daily or at least three times a week, depending on the weather (more often when wet or snowy), and note the amount of feed consumed. Low use may indicate illness or a **taste aversion**. However, during warm winter days or the first days of spring, some cranes stop eating for a day or more and rely on stored fat. An easy test of food use is to **mound the food into a cone** in the feeder and look for depressions in the cone that day or the next. Placing a favorite food, such as smelt or corn, on top of the food also confirms whether a crane is eating. Another method is to mark the food level and determine quantitatively how much the food

level is reduced. Be aware, however, of spillage and consumption by wild birds.

Discard wet or pulverized feed before it loses its nutritional value or becomes moldy. Completely change the feed monthly and disinfect the feeder or bucket if it becomes wet.

Drinking Water

Cranes need fresh drinking water at all times. Constantly flowing, elevated, watering cups (Fig. 12.13) are preferred because they are the most sanitary watering system available and require a minimum of maintenance. Float-operated water troughs that automatically fill provide fresh water for extended periods and have also been used successfully. If cranes are housed in enclosures with fresh, running water, an artificial supply is not needed. Water should not flow from one crane to another. Clean the cups or automatic trough waterers at least once a week using a stiff brush. Check the water delivery system daily to make sure it is functioning properly. Nine-liter, heavy-duty, rubber buckets placed in a secure spot can be an alternate water source. However, these buckets require more effort to keep clean and they are a greater health hazard. If used, clean daily and disinfect once or twice each month. If an individual crane stands or defecates in its water container, elevate the container so the crane can no longer do so. Buckets should always be kept on hand for use when the automatic waterers fail or freeze.

In cold climates, drinking water may require heating to prevent freezing. Some automatic watering systems have built-in heaters. Flowing water may not require heating, but for buckets, a pole-type water heater (see Appendix) works well.

Handling and Transporting Cranes

Handling and Physical Restraint of Cranes

Safety Precautions. The capturing and restraining of cranes sometimes causes injuries to long legs, necks, and wings. Fledged colts seem particularly vulnerable to wing injuries. Fleeing cranes also hurt themselves by crashing into fences or flight netting. To minimize injury, we recommend slowly herding cranes into **capture corners** (padded corners lined

with tennis netting or another soft material). Once the cranes are cornered, the caretakers rush in the last few meters to grab the birds.

Capturing and restraining cranes can also be risky to handlers because of potential injury from the bill or feet. When in full attack, a crane stabs with the bill, rakes with the talons, and strikes with the wings. A single stab can blind a person. Anecdotes are available of one human mortality and other near death injuries from crane bills (e.g., Bent 1926:227). Most experienced handlers have many minor scars. Some caretakers have sustained blows to their protective goggles. Always wear **eye protection** when capturing large cranes or dangerous individuals, and when performing AI, use leather leg coverings (chaps) to protect legs against claw and bill injuries. Aggressive cranes can be fended off by one person holding a **broom** (Fig. 2.3) or **T-stick** (a lightweight, 1-m long handle with a sturdy wooden crossbar at one end) against the crane's chest to keep it at bay.

Cranes that thrash around or peck at the handler while being restrained can usually be calmed down by covering the crane's head and eyes with a **hood**. A hood is a tube-shaped piece of fabric that can slip over the head, bill first, and then be fastened around the back of the head to keep it in place. At Patuxent, hoods are seldom used except with chicks during veterinarian exams. When using a hood, keep the nares uncovered. It is advisable to place a rigid paper or plastic disk over the crown to lift the fabric away from the eyes and thereby eliminate corneal abrasion.

Capturing Adult Cranes. Two to four people should herd the crane into a capture corner. Approach the crane slowly with arms outstretched to herd the crane into the desired location. When the crane is about to escape past the caretakers, rush in and grab the bustle (the rearward, protruding, elongate tertiaries), both wings, or one wing (humerus) and the neck. If the crane goes into a shelter, catch it as quickly as possible so that it does not jump into a wall and injure itself. For cranes that tend to jump, angle your arms upward as well as outward when cornering the crane, and be prepared to grab a wing as the crane tries to jump over or past you.

Parent-reared and wild-caught cranes are usually more difficult to capture. For such birds, a special capture corner is important and more people may be required. The wildest cranes may require a temporary capture lane constructed of wire panels (Fig. 2.4).

Hand-raised cranes are usually easier to capture. Often a single person can approach close enough for

the capture, although some hand-reared cranes are aggressive and dangerous enough that two people are required; one diverts the crane while the other quickly grabs the crane by the wing and the base of the neck. Most cranes that are captured regularly (as for AI) can be trained to go to the same part of the pen each time the AI crew herds them in a certain manner.

Restraining Adult Cranes. Restrain wings and legs as soon as possible after catching the crane (Fig. 2.5). The handler immediately pulls the crane's body against his/her own and turns away from the bird's



Fig. 2.3. Brooms are used to defend against aggressive cranes; Linda Miller retrieves the chick while Jane Nicolich defends and Scott Hereford exchanges the chick's food and water.

Photo David H. Ellis



Fig. 2.4. A portable capture lane is constructed of rigid, self-standing panels; Carlyn Williamson (left) and Jane Nicolich herd cranes. Also note tennis netting, capture corner.

Photo David H. Ellis



Fig. 2.5. *Restrain wings and legs after capturing an adult crane. For very dangerous birds, the head can be restrained by a second person. Scott Hereford (left) and Thom Lewis with a Whooping Crane.*

Photo David H. Ellis



Fig. 2.6. *Hold the adult crane's wings and body with one arm and its legs with the other; Sandy L. Meyerhoff.*

Photo Glenn H. Olsen

bill to avoid facial injury. A second person should hold the head of cranes that peck people. Restrain the head by encircling the bird's upper neck with one hand without restricting the airway; do not cover the nares. Hood if necessary. Another method is to hold the wings and body of the crane with one arm and the legs with the other (Fig. 2.6). Allow the lower legs to flail if they do not need to be folded for the procedure. Grasp the legs just above the hocks, but always place one finger between the hocks (Fig. 2.11) to prevent the legs from abrading one another. Make sure the tarsi cannot contact the crane's neck or the person holding the crane.

If the crane's legs must be folded, gently force the tarsi around, but if the crane locks its hocks rigidly, **do not force the legs to fold**. Instead, maintain steady pressure on the tarsi until the crane allows you to fold the legs. When holding a crane with folded legs, support the crane's weight with the arm holding its body. **Do not support the crane's weight on its folded legs**. Do not keep a crane's legs folded for more than 30 minutes. These precautions will help prevent slipped tendons, capture myopathy (Carpenter et al. 1993), and in some cases (usually in chicks), leg fractures. The leg-folded carry is the primary method used at ICF for all but Whooping Cranes and individuals with a history of leg problems. It is the most convenient carry and helps avoid injury to cranes and humans caused by flailing legs, but has been implicated in some crane injuries.

At ICF, cranes whose legs are folded are sometimes placed in a sitting position on the ground for examinations or treatments (Fig. 2.7). Kneel with your legs surrounding the crane's wings but without placing weight on the crane. Use your hands to hold the crane down if it struggles to rise. This position is useful for examining the head and dorsum or force feeding.

For brief periods of restraint, it is often helpful to hold and stroke the crane as for AI while a second person quickly examines the crane or administers medical treatments. AI stroking is also effective in neutralizing an aggressive crane.

Releasing Cranes After Restraint. When releasing a crane, allow its legs to touch the ground before letting go of the body or wings. We hold the bustle or one wing briefly while releasing the legs to insure that the crane is stable on its feet. As the crane gains stability, move forward a step or two with the bird. These precautions help prevent leg injuries to cranes that are unable to stand on their own without a few seconds of support. This practice is not advisable



Fig. 2.7. *Scott R. Swengel restrains a Brolga by sitting across it. Only the hands actually press down on the bird.*

Photo David H. Thompson



Fig. 2.8. *Yuri Markin weighing a Siberian Crane chick in cloth sack; Mini Nagendran looks on. Northwest Siberia.*

Photo David H. Ellis

for cranes that thrash violently on release because they are more likely to become injured by thrashing about than from release while temporarily unstable.

Handling Chicks. Chicks are more fragile than adult cranes. Growing legs, wings, and flight feathers are especially vulnerable to injuries. Use less force when capturing and restraining chicks. It is also important to minimize the length of time that chicks are restrained because chicks seem to become severely stressed after only a few minutes of handling. Chapter 5 describes the proper methods of handling young crane chicks. Chicks that are close to adult size should be captured and held like adults except with extra caution.

Weighing Cranes

Young chicks can be placed in a cardboard box that is then placed on a scale (see Chapter 5). The box should be tall enough to prevent the chick from climbing out, and a carpet or mat should be placed in the box for good footing. The caretaker should keep one hand close to the top of the box to make sure the chick does not climb out or tip the box.

Larger cranes (>2 kg) can be weighed on a platform scale. Some cranes are calm enough to stand on the scale on their own. ICF currently weighs cranes while being held by a person standing on a scale.

A 10-15 kg capacity suspension spring scale with 0.1 kg accuracy can be used to more accurately weigh cranes in their pens or in the field. The method used for many years with minimal injury is to place the

crane in a cloth sack tail first, with the neck and head projecting from the bag (Fig. 2.8). Gather the slack material into a roll over the crane's back, and pierce the hook of the spring scale through the bag just under the roll. Suspend the crane and bag from the scale, while holding one hand just under the bag to control the crane's movements while in the bag. Weighing cranes in this way requires that the crane's legs be folded. Occasional injuries have resulted from this method. An alternative method employs a weighing sling (that allows for the legs to be left unfolded). Perhaps the simplest sling is a meter square net that is wrapped around the crane's body, then hooked in four or more places onto the scale. Patuxent has developed an innovative cloth sling with Velcro straps in the front and rear to restrain the bird's wings (Fig. 2.9).

Transporting Cranes

To minimize stress, move birds as little as possible (Mirande et al. 1988 unpubl.). In some facilities, pen rotation can be accomplished by merely herding the bird into the adjacent pen. For moves of less than 200 m, carry the crane while walking to the new location. For longer moves, hand carry the crane into a vehicle and hold it during transport. Use a hood for nervous or aggressive birds. For long-distance moves, crate the crane. If the crate is transported in an open truck moving at highway speeds, tie the crate down, otherwise wind drag may blow it over or even out of the truck. When driving, avoid abrupt turns, sudden changes in speed, and bumps.



Fig. 2.9. Dan Sprague weighs a Sandhill Crane using a sling with Velcro fasteners.

Photo David H. Ellis

If the crane must wait for more than a few minutes after capture, place it in a crate (Fig. 2.10) until it will be examined. Never leave a crane crated for more than 10 minutes when the temperature is above 30°C (86°F) to avoid heat stress. When shipping cranes by air, crate design and shipping arrangements should comply with International Animal Transfer Association (IATA) guidelines available from the airlines or ICF.

Crates. Crates for adult cranes should be large enough for the crane to stand comfortably with its neck recurved, but small enough to prevent the crane from opening its wings or jumping enough to hurt itself. A good generalized crane crate has **inside dimensions** of 95 cm high \times 40 cm wide \times 90 cm long. We sometimes use double crates with a solid divider for carrying two cranes. The tallest cranes, Wattled and Sarus, need only a 100 cm tall crate, and shorter cranes, Demoiselle, Hooded, and African Crowned Cranes, should have crates proportionally smaller. For Demoiselle Cranes, crates should be 75 cm high \times 30 cm wide \times 65 cm long.

Most airlines will not accept crates taller than 105 cm. Construct crates to minimize the outside dimensions while maintaining adequate inside size and overall strength. If crates are taller than 80 cm, check with airlines before booking a shipment to make sure the oxygenated cargo hold of the plane has a large enough door. Label the crate with appropriate instructions, and instruct the airline personnel to keep the crate upright while moving it into the cargo hold.

To prevent feather injuries when transporting a chick, adjust the crate size to prevent the chick from turning around. For example, most adult Florida Sandhill Cranes can only with difficulty turn around in a 40 cm wide crate. For a 3-month-old Florida Sandhill chick, 35 cm would be a better width. In general, the crate should be 12.5 cm wider than the bird with folded wings. Length and height should be proportionately adjusted.

Temperature. Different species of cranes tolerate heat and cold differently. Subtropical cranes can withstand heat better than temperate ones. Red-crowned and Siberian Cranes are the least heat tolerant and the most cold tolerant of all cranes. Use the conditions in the natural environment of the species to judge its likely tolerance to heat and cold. During a long road trip in hot weather, check on the crane hourly or more often if stress is likely. At some of these checks, place a 6-cm deep water dish just inside the door of the crate for a few minutes. Avoid airline shipments when the temperature is above 21°C (70°F) and below -1°C (30°F). The airlines themselves may also have restrictions. Normally these rules allow shipment only



Fig. 2.10. Cranes are routinely transported in special crates. Bryant Tarr and Julie Langenberg pose next to a double crate and a juvenile Siberian Crane on its way to India, January 1994.

Photo David H. Thompson

between 7° C (45° F) and 27° C (80° F), but a veterinarian's letter of recommendation can sometimes persuade the airline staff to waive the rule. If the flight is nonstop and the crate will not sit outside before or after transport, it may still be possible to transport birds outside this temperature range. Allow for unforeseen events that may change the flight schedule and jeopardize the bird.

Food and Water. Adult cranes do not need food during trips of less than two days. The higher the temperature, the more often cranes need to drink during transport. At cold to moderate temperatures, cranes need to drink after one day of travel. Placing a familiar water dish just inside the door for 1/2 hour will give the crane adequate time to drink. Never install a dish as a permanent part of the crate, because birds can injure their feet or legs, break blood feathers, or hurt their heads and necks on such structures.

Special Needs of Chicks. Young cranes, especially those less than 4 months old, require special care. Because they are less tolerant of environmental extremes than adults, young cranes should not be transported except for special purposes, and even then they should be accompanied by a caretaker. Young cranes need water every few hours instead of once a day, and at least one good feeding per day. In addition, young cranes are prone to leg and wing injuries during transport; provide extra floor padding in the crate.

Materials and Construction of Crates. The sides of the crate, including the door, can be made with 0.25-in (0.6-cm) plywood and reinforced along all edges with 2 × 4 cm strips of wood. The floor should be 0.5- to 0.75-in (1.3- to 2-cm) plywood. The top can be of plywood or some other strong material. Mesh hardware cloth (1 cm) sandwiched between two layers of tennis wind-netting will serve to protect the crane's head and allow ventilation while restricting view and thereby reducing disturbance. For greater ventilation during warm weather shipments, provide a similarly constructed window over one-third to one-half of the back of the crate. It is also advisable to provide rows of 2-3 cm diameter ventilation holes near the top of the crate. However, unless the holes are covered with mesh, they increase the chance of injury if the crane protrudes its bill through a hole.

Place a 5 cm layer of wood shavings on the carpet to absorb the crane's feces. The door of the crate should be along the shortest side and slide up and down in a narrow track. The top of the door should

have a fastening system that permits the door to be locked. Attach handles made of 2 × 2 cm strips of wood running the length of the sides near the top.

Preventing Injuries. Toenail and wing trauma are the most common injuries observed when transporting cranes. Minimize these injuries by brailing the wings (see Chapter 11E), taping pads on the carpus of pinioned wings, eliminating rough edges inside the compartment, fastening grippable floor material very securely along its perimeter, and choosing an appropriate crate size. The groove for the sliding door should be as narrow as possible so that cranes cannot get their toenails hooked in the groove during transport.

Labeling. Label the crate "LIVE BIRD" on at least two sides and "THIS SIDE UP" on all four sides. "DO NOT TIP" and "DO NOT FEED OR WATER THIS BIRD" are also useful labels. Write the names, addresses, and phone numbers of the sending and receiving parties on the crate so that the airline may contact the parties if there are any difficulties during the shipment. IATA requires that feeding and watering instructions be attached to the crate.

Care of Recent Arrivals. Before a crane is shipped to you, learn about the crane's behavior, food requirements, and habits to provide better care when it arrives. Try to use feeders and waterers similar to the ones to which the crane was accustomed, then change over gradually to your system. If the crane does not eat immediately, sprinkle its previous foods on top of the new diet to encourage use of the new food.

Marking Cranes

Bands

Metal bands engraved with an identification (ID) number and placed above one hock make good permanent ID markers (see Appendix). If the sex of the crane is known before permanent banding of the bird, males can be banded on one leg and females on the other. This makes paired cranes individually recognizable at a distance.

Colored leg bands allow more recognizable marking combinations than aluminum bands and are especially useful in making behavioral observations of cranes in groups. Different colors and positions of one to three leg bands allow for thousands of combinations.



Fig. 2.11. Crane band showing coded information for captive cranes: color (taxon), 84 (hatch year), 032 (specimen number), and incised white ring (sex). Numbers on bands for release birds should be taller (at least 2 cm) and fewer to allow reading at 300 m with a telescope. (Note fingers between crane's legs preventing abrasion.)

Photo David H. Ellis

Color bands and aluminum bands can be combined to generate additional marking combinations. Bands made of laminated plastic (e.g., Gravoply, see Appendix) with two layers of contrasting colors can be engraved with unique combinations of letters and numbers. At Patuxent, we engrave a narrow ring high on the band for males (Fig. 2.11) and low for females.

To make color bands, cut strips of plastic of the appropriate size from a sheet of 3-mm thick plastic. Next, engrave the alpha-numeric code on the plastic. Heat the plastic strips in a teflon-coated and lubricated (use non-stick cooking spray or mold release) electric frying pan set at 130° C (266° F). Using a higher temperature will distort characters that are engraved in the plastic. Some plastics become pliable enough to form into bands after submersion in boiling water. When the strips become pliable, quickly form them into the right shape around the proper size dowels (wear gloves during this step of the procedure). The bands will cool and become rigid in a few seconds, so roll them quickly.

Wrap the plastic 1.25-1.75 times around the dowel, depending on the height of the band. Tall bands require less overlap than short bands. The more a band overlaps, the more difficult it is to attach and remove. When making plastic bands that wrap around 1.25 times, choose plastic strips

that are 4-4.5 times as long as the band's inside diameter. For bands that will be wrapped around 1.75 times, cut strips that are 6-6.5 times the length of the band's inside diameter. The width of the plastic strip is usually 25-80 mm.

Attaching Color Bands. Color bands require softening in 50° C (122° F) water to make them pliable enough to put on. The band will harden back to its old shape very quickly as it cools. If the band is wrapped around 1.25 and 1.75 times for 80 and 25 mm tall bands, respectively, the crane will be unable to remove the band with its bill. If you wish to glue (or weld) the band closed, place a few drops of acetone between the overlapping ends and hold the band closed for 10-15 seconds.

Band Size. The best band diameter is 14 mm inside diameter for small cranes (Black Crowned, Gray Crowned, Demoiselle, and Hooded), 18-21 mm inside diameter for large cranes (Sarus and Red-crowned), and 16-18 mm inside diameter for the remaining cranes. Aluminum bands are usually 12-16 mm high, while color bands are taller (25-80 mm) to make them easy to see. Large (>40 mm high) color bands allow more room to engrave numbers and letters. Small (25-40 mm tall) color bands are preferable if cranes will have two or more color bands.

Bands that are stacked one on top of the other should be the same diameter and have plenty of overlap to prevent the upper band from sliding over or under the lower. An interlocking aluminum band placed between two color bands also prevents one color band from slipping down over the other (S. A. Nesbitt, Florida Game and Fish, Gainesville, Florida, personal communication).

Other Marking Methods

Tattoos on the underside of the patagium are useful for permanent marking of birds but, like aluminum bands, are not good for long-range identification. **Wing tags** are poor markers because cranes often destroy them after a few months or years.

Neck collars can be dangerous to cranes. The birds can get the tips of their long bills caught in the upper end of the collar and die from starvation or neck injuries. Unless further experimentation reveals that some collars are safe, we recommend against using them.

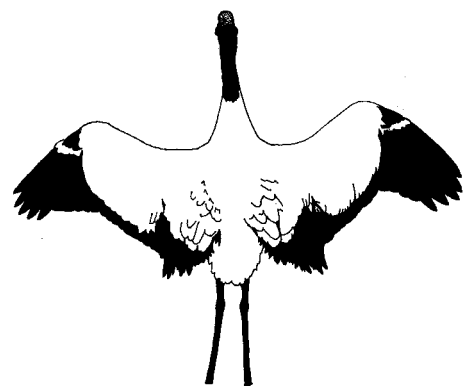
Transponders (coded electronic microchip implants) are the newest identification method to be used for cranes. The small (2 × 11 mm), sterile,

uniquely coded microchips are injected by syringe under the skin, where they can be detected and the code number read by a hand-held electronic scanner up to 0.5 m. Transponders are becoming the standard method for permanent identification of zoo animals. However, they are not useful for long-range identification, and the systems are expensive (ca US\$4-10/microchip and US\$250-1000 for the reader). The World Conservation Union (IUCN) has recommended the Trovan/A.E.G. system (see Appendix) as the global standard, and the dorsal base of the neck as the preferred implantation site for cranes. Zoos routinely using transponders in cranes have reported no health problems associated with the microchips. Transponders may also be useful for released cranes, where long-term permanent identification of even a partially consumed carcass is important.



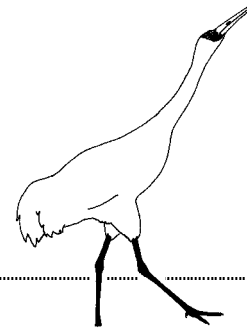
Literature Cited

- American Ornithologists' Union. 1988. Report of committee on use of wild birds in research. *Auk* 105 (1, Supplement):1A-41A.
- Bent, A. C. 1926. Life histories of North American marsh birds. Smithsonian Institution, U.S. National Museum Bulletin 135.
- Canadian Council on Animal Care. 1984. Guide to the care and use of experimental animals, Vol. 1. Public Health Service, Canadian Council on Animal Care, Ottawa.
- Carpenter, J. W. 1979. An outline of the treatment and control of crane parasites. Pages 101-108 *in* J. C. Lewis, editor. Proceedings 1978 Crane Workshop. Colorado State University Printing, Fort Collins.
- Carpenter, J. W., and S. R. Derrickson. 1987. Infectious and parasitic diseases of cranes: principles of treatment and prevention. Pages 539-553 *in* G. W. Archibald and R. F. Pasquier, editors. Proceedings of the 1987 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Carpenter, J. W., N. Thomas, and S. Reeves. 1993. Capture myopathy in an endangered Sandhill Crane. Pages 111-112 *in* J. H. Olsen and M. Eisenacher, editors. Proceedings 1989 Annual Meeting American Association of Zoo Veterinarians.
- Halibey, T. 1979 unpubl. Food and feeding habits of captive cranes. International Crane Foundation report.
- Mirande, C. M., J. W. Carpenter, and A. Burke. 1988 unpubl. The effect of disturbance on the reproduction and management of captive cranes. Paper presented at 1988 North American Crane Workshop. 18 pp.
- Russman, S. E., and M. S. Putnam. 1980. Nutrition and successful propagation of cranes. Pages 261-282 *in* E. R. Maschgan, M. E. Allen, and L. E. Fisher, editors. Proceedings of the First Annual Dr. Scholl Nutrition Conference.
- Serafin, J. A. 1982. The influence of diet composition upon growth and development of Sandhill Cranes. *Condor* 84:427-434.
- United States Department of Agriculture. 1989. Part IV, Department of Agriculture Animal and Plant Health Inspection Service: 9 CFR Parts 1, 2, and 3, Animal Welfare; final rules. *Federal Register* 54(168):36112-36163.



Egg and Semen Production

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This chapter reviews ways to induce egg and semen production in captive cranes and provides managers with guidelines for establishing and evaluating their programs.

Behavioral Factors

Pair Bond

Captive cranes need a **strong pair bond** to breed. In the wild, cranes freely choose mates, but in captivity, we choose mates based on availability and genetic concerns. During courtship, there are strong feedback mechanisms between **behavior and hormonal status** (Murton and Westwood 1977; see Chapter 7 for greater detail). Courtship synchronizes mates and promotes development of the reproductive system. This synchrony is critical because crane pairs must cooperate closely to copulate, incubate, and successfully raise chicks. Behavioral management is a critical component of successful captive propagation, and readers are encouraged to study Chapter 6.

Disturbance

In general, transferring pairs to different pens or breeding facilities disrupts reproductive activity (Mirande et al. 1989 unpubl.). However, cranes may benefit from **transfers**, such as removal from display or return to a pen where the pair previously bred. Annual rotation of pairs between adjacent pens (for disease management) does not seem to have an adverse affect. If moves are needed, they should be scheduled after the breeding season. Valuable breeding pairs should be kept “off-exhibit” because cranes maintained on **public display** produce significantly fewer eggs (Mirande et al. 1989 unpubl.).

Persistent **aggressive interactions** with cranes in neighboring pens can inhibit breeding and should be

managed with visual barriers, buffer zones, or pen switches. Birds should be carefully monitored for behavioral **signs of stress** associated with disturbances, such as frequent pacing of pen boundaries, excessive preening, decreased or increased calling, egg breaking, etc. If you observe such behavior and cannot correct the problem, consider moving the birds (Derrickson and Carpenter 1987).

Egg and Chick Adoptions

Some pairs that have not laid eggs can be induced to incubate eggs and adopt chicks (see Chapter 5 for details of chick adoptions). The behavior associated with incubation and chick rearing strengthens the pair bond, may stimulate the pair to breed earlier, and may induce non-breeders to reproduce in subsequent years. We speculate that successful adoptions elevate the prolactin level in the pair.

Patuxent has introduced dummy eggs to eight pairs of Whooping Cranes that have never before laid eggs: two pairs accepted and incubated eggs. One of these pairs hatched a chick and the other adopted a chick. Both pairs laid eggs for the first time in the following year. ICF has attempted three egg adoptions with non-laying Whooping Cranes: none were successful.

To adopt an egg, watch pairs for **signs of laying**, such as increased Bill-down posturing (Fig. 3.1), nest building or attraction to one area of the pen, broodiness, aggressiveness, increased calling, or Copulation. Parents should be chosen based on their previous experience with chicks and eggs (see Chapter 5 section on Choosing Parents). To avoid disrupting natural reproduction, egg adoptions should not be attempted if a pair seems likely to lay. However, if the laying season is ending and the pair either shows no clear signs of laying or a decrease in nest attendance, it may work to give the pair a dummy egg to try to induce them to adopt. See Chapter 5, Choosing Parents section, for parameters to use in evaluating surrogate parent's adoption potential. Unrated or first time pairs



Fig. 3.1. *Nest searching or Bill-down posturing of a female Siberian Crane.*

Photo Patty McCourt

should only be given non-endangered or non-valuable eggs for the first few years.

Quickly create a nest in an area of the pen where the pair seems most likely to lay. Then, without allowing the pair to see the egg in hand, **place a dummy egg** in the nest. If the pair initially ignores or attempts to break the egg, continue the adoption. It may take 2 to 7 days for the pair to accept the egg and begin incubating. After incubating for at least 14-21 days (25-30 is preferred by Patuxent), exchange the dummy egg for a pipped egg. If everything goes well, allow the pair to hatch or adopt and rear the chick.

To stimulate parental behavior in non-reproductive pairs, ICF also places 2-week- to 4-month-old chicks in the pen adjacent to pairs. Responses vary, but some adults stand in close proximity to the chicks and defend them. ICF was successful in adopting three 2-month-old Sandhill chicks to a lone female White-naped Crane and a 2-week-old Sandhill to each of two inexperienced pairs of Hooded Cranes.

Environmental Influences on Reproduction

Environmental factors, together with physiological conditions and endogenous rhythms, ensure proper reproductive timing (Marshall 1961; Sadlier 1969; Immelmann 1971, 1972; Welty 1975:147-153; Murton and Westwood 1977; Wingfield 1983; Wada 1984; Farner 1986). Physiologists define these environmental influences as either **ultimate** or **proximate** factors (see Chapter 7 for greater detail). By understanding how these factors affect reproduction, we can increase production by providing stimulatory cues and avoiding inhibitory factors.

Daylength (Photoperiod)

Farner (1986) says for birds "...in mid-to-high latitudes, the primary proximate factor is the annual cycle in daylength." Increasing the photoperiod stimulates breeding in many species of birds (Welty 1975:153-160; Murton and Westwood 1977; Farner 1986). Cranes that breed at **high latitudes** (Siberian, Lesser Sandhill, Hooded, and Whooping) often experience 20 or more hours of light each day. Although some individuals of these species have bred without an extended photoperiod, it is believed that extending the photoperiod artificially induced the first captive breeding of Hooded and Siberian Cranes. Most Hooded, Whooping, and all Siberian Cranes that have bred in captivity were provided with floodlights on automatic timers to artificially lengthen the photoperiod to 22-24 hours. Table 3.1 lists three **photoperiod control options** for breeding these species.

Mid-latitude cranes can also respond to an extended photoperiod. When artificially photostimulated, Greater Sandhill Cranes lay eggs earlier than controls (Gee and Pendleton 1992). For tropical species, captive egg production is: (1) positively correlated with day length in those species which, in the wild, breed when days are longest (e.g., Sarus Cranes), and (2) negatively correlated with day length in species which breed in the winter (e.g., Wattled Cranes, $p=0.01$) when days are shortest (Balzano 1989 unpubl.).

The high latitude species lay eggs in the cool and moist days of late May and June. Because these species breed in more southerly latitudes in captivity, it is important to stimulate breeding earlier in the season than would occur in the wild. Warm temperatures at

TABLE 3.1.

Three artificially lengthened photoperiod schedules used for breeding high latitude cranes in northern temperate zones.

A. General Schedule—ICF		B. Natural Siberian Crane Schedule—ICF		C. Whooping Crane Schedule—Patuxent	
Date	Daylength (h)	Date	Daylength (h)	Date	Daylength (h)
22-28 Feb	12:00	28 Feb-6 Mar	13:50	16-22 Feb	15:30
1-7 Mar	13:00	6-17 Mar	14:15	23 Feb-1 Mar	15:55
8-14 Mar	14:00	17-24 Mar	14:35	2-8 Mar	16:20
15-21 Mar	15:00	24 Mar-3 Apr	14:55	9-15 Mar	16:50
22-28 Mar	16:00	3-10 Apr	21:30	16-22 Mar	17:15
29 Mar-4 Apr	17:00	10 Apr-5 Jul	24:00 ¹	23-29 Mar	17:50
5-11 Apr	18:00			30 Mar-5 Apr	18:25
12-18 Apr	19:00			6-12 Apr	18:55
19-25 Apr	20:00			13-19 Apr	19:30
26 Apr-2 May	21:00			20-26 Apr	20:05
3 May-5 Jul	22:00 ¹			27 Apr-3 May	20:40
				4-10 May	21:20
				11-17 May	22:00
				18-24 May	22:30
				25-31 May	23:05
				1 Jun-	23:45 ²

¹ To avoid possible harmful effects associated with a sudden decrease in daylength, ICF decreases photoperiod by one hour each week after production has ended (approximately 5 July). Artificial lighting is discontinued when natural daylength is reached. Patuxent completely discontinues the use of lights when the last egg hatches.

² ICF uses the same schedule for Whooping Cranes, but starts one week later.

Patuxent in early May and at ICF in late May seem to inhibit reproduction. Extending photoperiod early in the spring stimulates these species to breed when **temperature and humidity** at the captive site resemble the native habitat.

Cranes will adjust their breeding schedule based on **latitude and climate** (primarily photoperiod and temperature). For example, Greater Sandhill Cranes not exposed to an altered photoperiod start nesting two weeks later at ICF than at Patuxent. To lengthen the laying season, site-by-site photoperiod adjustments are beneficial. For example, ICF advances the cycle by 5-7 weeks for Siberian Cranes to increase the photoperiod to 21.5 hours in early April when temperatures are similar to those in the native habitat in May. Also, after ICF received Whooping Cranes from Patuxent, the photoperiod schedule used at Patuxent was started one week later to compensate for the later spring at ICF.

Light intensity, and to a lesser extent **spectral characteristics**, are important when choosing an artificial light source. From the literature for other bird groups (Morris 1967), we recommend 16 or more foot-candles (ca 170 lux) throughout the pen and shelter. To meet this goal, check the most dimly lit corners in the pen with a standard photographic light meter (incident light measurement) after dark and adjust the light source to supply this minimum. Normal incandescent bulbs provide a good color spectrum, but burn out quickly and are energy inefficient. Some quartz or metal halide lamps provide a good light spectrum, are long lasting and efficient, and require fewer lamps to provide proper intensity throughout the pen. Lamps can be mounted on poles along pen perimeters or suspended overhead. If birds are rotated to fallow pens in alternate years, lamps on poles can be swiveled to accommodate the rotation (Fig. 3.2).



Fig. 3.2. Photoperiod lights, if mounted on swivel heads, can be turned to illuminate the adjacent pen in alternate years. Thom Lewis adjusts the position of a light. Photo David H. Ellis

Most facilities control photoperiod lights by a **timer clock** mounted near the pens. Timers should be adjusted weekly and checked regularly. Although a single night with altered photoperiod is normally insufficient to disrupt breeding, great care should be taken to prevent interruptions or other unprogrammed changes in the photoperiod regime.

Twilight is a time of great activity for cranes, and its simulation may increase the rates of courtship, copulation, etc. Twilight is much longer at high latitudes (sometimes exceeding three hours) and is likely to be most important for boreal and austral cranes. Future **research** on the importance of twilight and the effects of an artificial photoperiod on reproduction in cranes is needed, although controlled experimentation with adequate sample sizes is difficult.

Rainfall

Tropical species, including the Sarus, Brolga, and Crowned Cranes, breed during the rainy season (Archibald and Swengel 1987; Konrad 1987). Rainfall may be the proximate or ultimate factor stimulating breeding in these species. The laying season of captive Sarus Cranes was positively correlated with rainfall in

two of three captive centers ($p < 0.05$) and in the wild ($p < 0.001$, Balzano 1989 unpubl.). Wild Wattled Cranes usually nest at the end of the rainy season. Egg laying in Wattled Cranes was negatively correlated with rainfall in the wild population ($p < 0.05$) and in one of three captive centers studied ($p < 0.01$, Balzano 1989 unpubl.).

Although tropical cranes have bred without **artificial rain**, sprinklers at ICF simulating the rainy season (Fig. 3.3) are believed to have stimulated the first captive breeding of Brolga Cranes. Other zoos have also used sprinklers to increase laying in Crowned Cranes.

We recommend that a **sprinkler system** provide a fine mist covering the entire pen. The system should operate two to five times each day, and the length of each shower should be adjusted to prevent the development of puddles of stagnant water in the pens. Shield nests to prevent nesting materials from becoming moldy.

Latitude

The onset of egg laying varies with latitude. For north temperate zone breeding birds (disregarding the effects of altitude), the laying season begins an average of three to four days later for each degree of increasing latitude (Welty 1975:148). Data on wild Sandhill Cranes clearly demonstrate later initiation dates and peak production periods as latitude increases (Walkinshaw 1973); the Mississippi Sandhill Crane is an exception (see Fig. 3.4). A similar pattern is observed in captive Greater Sandhill Cranes when laying dates for different breeding centers are examined (Table 3.2). Because of these temporal trends, we recommend that boreal and temperate species be maintained at higher latitudes. Tropical species do best in areas with less climatic variation.

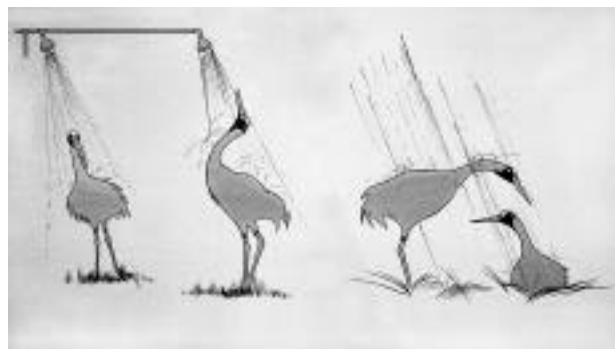


Fig. 3.3. Sprinklers can be used to simulate rainfall to promote breeding in tropical cranes, here Brolgas.

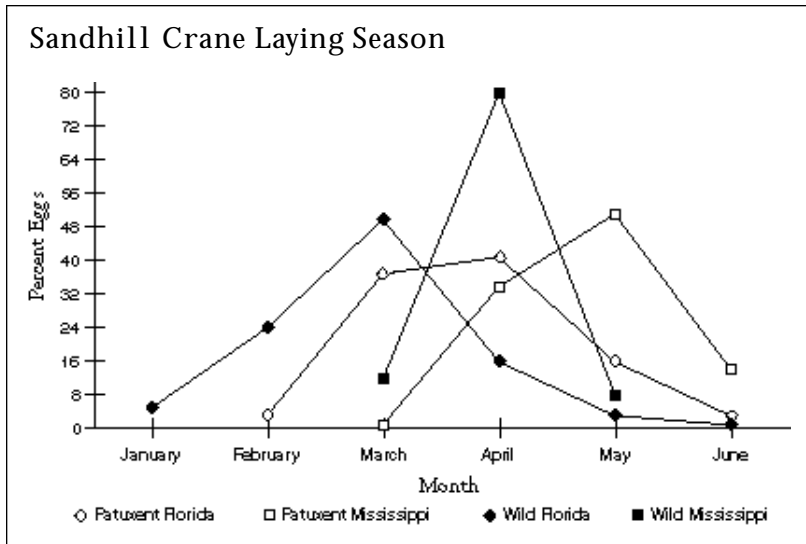


Fig. 3.4. Laying season for two nonmigratory, warm-climate races of the Sandhill Crane. The Patuxent colonies are ca 8-11° further north than the wild populations.

Temperature

Temperature affects both the onset and termination of the breeding season. Balzano (1989 unpubl.) found that temperature and egg laying rates were positively correlated in Sarus Cranes ($p < 0.05$ in 2 of 3 captive populations), negatively correlated in Wattled Cranes ($p = 0.002$ in the wild population; $p < 0.001$ in 1 of 3 captive populations), and uncorrelated in White-necked Cranes. Male Sandhill Cranes have produced semen in winter when kept indoors at moderate temperatures (21°C ; 70°F) and on a lengthened photoperiod (ca 24 hours) (Gee and Pendleton 1992).

The onset of hot weather seems to terminate semen production. For Siberian Cranes, semen production ceases above 21°C (70°F) daytime peak (Einsweiler 1988 unpubl.). This can result in insufficient semen to fertilize late eggs. For other species, the males may stop producing based on female behavior, not temperature.

Open Water

Opinions differ on the importance of standing water to promote breeding in cranes. Water conditions around nest sites variously stimulate or inhibit egg laying in birds (Lack 1933; Welty 1975:152). At the Wildlife Survival Center in Georgia operated by the Wildlife Conservation Society, cranes were successfully bred in large marshy enclosures. During a study

of Wattled Cranes in which some pens were artificially flooded (half of each pen) and controls were not, higher productivity was observed in the flooded pens (C. Sheppard, Wildlife Conservation Society, Bronx, New York, personal communication).

Unless the flooded area is large or there is good water flow, **disease risks** heighten. Factors such as surface area, flow rate, temperature, water depth, soil type, and amount of crane use need to be considered. Access to open water also increases the danger of frozen feet in cold climates. Artificial pools are expensive, labor intensive, and costly to maintain.

Patuxent formerly provided flowing water in **concrete pools** (about four feet in diameter sloping to one foot in depth). The cranes spent much time standing, bathing, and drinking in the pools. However, preliminary data indicated that pairs with flowing water (provided in elevated cups, Fig. 12.13) produced more eggs than pairs in pens with pools and did so without the maintenance and disease problems associated with pools. Nevertheless, the pairs with pools may have ultimately done better with improved husbandry.

ICF is currently examining the effects of **seasonal pen flooding** (Fig. 3.5) to stimulate breeding in nonproductive, but sexually mature, Siberian and Whooping Cranes. Preliminary observations show an increase in foraging and pair interactions and a decrease in territorial defense. Rate of flow is adjusted so water continually drains into the soil reducing disease risks. Seasonal flooding reduces disease risks by allowing soil to dry for 9-10 months each year. Sandy soils at ICF also insure good drainage.

Because many cranes breed without open water, we do not recommend pools for most breeding centers. However, when open water can be managed efficiently, it should promote breeding. One compromise is to provide open water to the most genetically valuable or most difficult to breed pairs, especially if AI is not feasible.

Nest Sites

Cranes need undisturbed nesting sites. Wild cranes generally nest in isolated places where the risk of predation is minimal. Some captive cranes (especially wild caught or nervous birds) also seek seclusion for

TABLE 3.2.

Monthly distribution of eggs (%) laid by captive and wild cranes.

Percent of Eggs Laid by Month	Jan	Feb	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
International Crane Foundation (Latitude 43° 35')—Captive												
Northern cranes												
Florida Sandhill	—	—	17	30	32	15	4	2	—	—	—	—
Greater Sandhill	—	—	—	12	68	20	—	—	—	—	—	—
Red-crowned	—	—	7	26	54	13	—	—	—	—	—	—
Demoiselle	—	—	—	46	54	—	—	—	—	—	—	—
Common	—	—	—	19	65	16	—	—	—	—	—	—
White-naped	—	—	—	23	52	23	2	—	—	—	—	—
Hooded ¹	—	—	—	—	100	—	—	—	—	—	—	—
Siberian ¹	—	—	8	43	48	1	—	—	—	—	—	—
Whooping ¹	—	—	—	50	50	—	—	—	—	—	—	—
Tropical and southern cranes												
Eastern Sarus	—	—	—	—	3	32	49	16	—	—	—	—
Indian Sarus	—	—	—	—	16	44	24	16	—	—	—	—
Blue	—	—	—	—	14	64	21	—	—	—	—	—
Wattled	16	28	16	16	16	—	—	—	—	—	3	6
Brolga	—	—	—	—	17	42	25	17	—	—	—	—
Patuxent Wildlife Research Center (Latitude 39° 02')—Captive												
Whooping ¹	—	—	11	56	33	—	—	—	—	—	—	—
Florida Sandhill	—	3	37	41	16	3	—	—	—	—	—	—
Greater Sandhill ¹	—	—	3	48	48	1	—	—	—	—	—	—
Mississippi Sandhill	—	—	1	34	51	14	—	—	—	—	—	—
Audubon Park and Zoological Gardens (New Orleans, Louisiana) and San Antonio (Texas) Zoo (Latitudes 30° 00' and 29° 25')—Captive												
Whooping	—	12	29	22	29	8	—	—	—	—	—	—
Wild Cranes²												
Northern cranes												
Florida Sandhill	5	24	50	16	3	1	—	—	—	—	—	2
Greater Sandhill ³	—	—	—	62	36	1	1	—	—	—	—	—
Red-crowned	—	—	5	63	32	—	—	—	—	—	—	—
Demoiselle	—	—	—	9	78	9	3	—	—	—	—	—
Common	—	—	—	29	54	17	—	—	—	—	—	—
Tropical and southern cranes												
Blue	11	7	2	—	—	—	—	—	—	15	26	39
Wattled	4	3	2	4	18	19	13	14	3	9	3	7
Brolga	14	25	31	5	3	—	—	8	5	3	5	—

¹ Captive birds managed under photoperiod lights² From Walkinshaw (1973).³ Cranes breeding in Michigan.



Fig. 3.5. A small pool in a Whooping Crane pen.

Photo Ann Burke

nesting. Other cranes sometimes build their nests in the most disturbed area of their pen or inside shelters. The rate of egg breakage at ICF is higher when eggs are laid indoors than when laid in the open pens. To minimize disturbance, place food and water near the pen entrance. At Patuxent and ICF, we enter food sheds from a separate outside door to minimize disturbance. For birds on display, caretaker activity and public viewing should leave sections of the pen undisturbed. Reproduction in ICF's crane exhibit building greatly increased when caretakers changed to providing food and water from the exterior, public-viewing area. The pairs selected nest sites in the undisturbed areas of the pens (Mirande et al. 1989 unpubl.).

Nest size varies greatly by species. The largest nests may be 3 m in diameter and 1 m tall in wet areas, or a mere scrape or a few arranged twigs in dry enclosures. Even within a single species, great variation occurs. Greater Sandhill Crane nests at Gray's Lake, Idaho vary from thick (0.6 m) mats 2-3 m wide on water to scrapes on the dry hillside. Some Demoiselle Cranes lay on the open steppe without any evidence of a nest. For all species, provide a suitable supply of twigs and coarse grasses to stimulate nesting behavior. Avoid materials that readily mold to reduce the risk of fungal infection. Patuxent primarily provides wheat straw (*Triticum aestivum*). In an experiment, ICF provided Siberian Cranes with four species of prairie grass including big bluestem (*Andropogon gerardi*), little bluestem (*A. scoparius*), Indian grass (*Sorghastrum nutans*), and Canada wild rye (*Elymus canadensis*). All except little bluestem, the finest grass, were used with stems 0.6 m and shorter preferred.

Food

During egg laying, birds draw on fat and calcium reserves and increase food consumption to provide essential energy, protein, and other nutrients for egg formation (Murton and Westwood 1977:147-214). Cranes may breed in response to factors associated with future food supply (e.g., rainfall, amount of open water, etc.) as well as the direct availability of food. Whooping Cranes feed more heavily on aquatic life during the breeding season, thereby increasing the amount of animal matter in the diet (Serafin and Archibald 1977 unpubl.). In captive females, Halibey (1976 unpubl.) documented increased consumption of feed and oyster shell (a calcium supplement) during egg production.

In a management regime, provide a **constant supply of fresh, nutritionally balanced food** to cue the cranes that conditions are optimal for reproduction (see Chapter 2 for diets). Patuxent provides a breeder diet with 22.0% (by weight) protein, 3.0% calcium, and 0.8% phosphorus. Similar values for ICF are 20.5%, 2.45%, and 0.89% respectively. Oyster shell is provided *ad libitum* one to two months prior to egg laying to all species and both sexes. Additional research is needed on the nutritional requirements for reproduction in cranes.

Sexual Maturity and Reproductive Lifespan

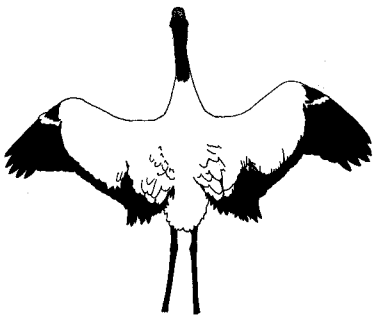
The age of **sexual maturity** and the initial appearance of reproductive behavior varies between species and individuals, and is strongly influenced by rearing history and management within species. In general, cranes form mating pairs when two to three years old and begin to reproduce when three to five years old. With rare exceptions, females lay eggs only when paired. Pair bonds persist and egg production continues (although sometimes at a lesser rate) even when members of a pair are separated into adjacent pens (Gee and Sexton 1979). **Egg production** usually begins one to two years after formation of a pair bond. Laying as early as two years of age has been occasionally reported in captive Wattled, Red-crowned, Eastern Sarus, and Sandhill Cranes, and is common in Mississippi Sandhill Cranes (25% of two-year-old females; Nicolich 1993).

For most species, captive cranes achieve reproductive success earlier than wild cranes. For some species, however, captivity appears to **delay egg production**. Captive Whooping Cranes generally start laying at 5-7 years of age, even though wild birds lay as early as three years of age (Kuyt and Gossen 1987). The mean age of first breeding is decreasing as management improves (Mirande 1994 unpubl.). Until a few years ago, most captive Siberian Cranes bred at seven years of age or older. However, with improved rearing and pairing techniques, Siberian Cranes are now breeding as early as four to six years of age (Panchenko 1993 unpubl.).

Sperm production generally begins at 2-3 years of age with regular production of quality semen usually occurring the following year. Unlike females that lay eggs only when mated, even unmated males produce sperm.

Captive cranes can live long and have an extended **reproductive lifespan**. A male Siberian crane at ICF lived to be at least 78 years of age and produced sperm until at least age 75. A pair of White-naped Cranes and a pair of Demoiselle Cranes produced young when both adults in each pair were at least 60 years of age. Great longevity has important management implications (see Chapter 9).

Age and experience increase reproductive success in cranes (Kuyt 1981; Nesbitt and Wenner 1987) and some other families of birds, especially those with delayed sexual maturity (Richdale 1951; Minton 1968; Gauthier 1989). At ICF, several trends are evident which appear to apply to all cranes. During the first three years of egg laying, (1) the first egg of the season appears progressively earlier (independent t-test, $p=0.038$), (2) the number of eggs produced increases ($p<0.001$), and (3) breeding season lengthens ($p<0.001$). The same trends were noted at Patuxent, but statistical tests were not applied because of confounding variables. First time breeders at both ICF and Patuxent are also more likely to break their eggs than experienced breeders.



Management of Egg Production

Characteristics of Egg Production

In all species, egg production is strongly **seasonal** (Table 3.2). External stimuli such as increasing daylength awakens the reproductive endocrine system. These hormones stimulate the development of the gonadal and accessory reproductive tissues. Eventually, ovulation occurs and an egg is laid about two days thereafter (see Chapter 7).

Timing and length of breeding season vary by species. Table 3.2 shows the egg production season for both captive and wild cranes. Initiation of laying dates for individual females is predictable, and females within a given species generally start laying in the same order each year. However, unusual events like sickness, pen moves, or other disturbance can alter the breeding schedule.

Cranes are indeterminate egg layers capable of reneating and multiple clutching. **Renesting** occurs after eggs are removed, destroyed, or abandoned. In the wild, renesting usually involves starting a new nest at a new location. In captivity, where sites are limited, pairs frequently reuse the same nest.

The **clutch size** for most species is two eggs. Wattled Cranes often lay a single egg, while Crowned Cranes may lay up to five eggs in a clutch.

The time period between successive eggs (whether from the same clutch or a new clutch) is known as the **egg laying interval**. If successive eggs are laid within 2-4 days of the previous egg, we consider them part of the same clutch.

From long-term data kept for each female (see Table 3.3), laying patterns begin to emerge. These patterns allow managers to better predict when a female will lay her first egg of the season, the timing of successive eggs, and the AI schedule. Two general patterns have emerged: some females maintain a clutching pattern (i.e., alternating long and short intervals) while other females exhibit gradually increasing intervals between eggs.

Multiple Clutching

Inducing captive cranes to lay higher numbers of eggs in one season by removing eggs from the nest is known as **multiple clutching**. Eggs can be removed

TABLE 3.3.

Sample egg laying interval record for one Florida Sandhill Crane at the International Crane Foundation.

Year	Total Number of Eggs	Season Length	First Date	Last Date	Egg Number ¹									
					2	3	4	5	6	7	8	9		
1981	2	3	19 Apr	22 Apr	3									
1982	4	61	9 Apr	9 Jun	3	56	2							
1983	6	58	6 Apr	3 Jun	3	20	3	29	3					
1984	6	43	6 Apr	19 May	3	21	5	13	3					
1985	6	65	21 Mar	25 May	2	15	3	43	2					
1986	6	24	30 Mar	23 Apr	2	8	5	3	6					
1987	9	47	28 Mar	13 Jun	3	6	7	26	3	6	5	9		
1988	8	38	5 May	12 Jun	2	4	8	4	6	8	6			
1989	6	35	10 May	14 Jun	3	5	14	5	8					
1990	2	2	18 Apr	19 Apr	1									

¹ Number of days between successive eggs (i.e., in 1982, the second egg came three days after the first, the third followed the second by 56 days, and the fourth was laid two days after the third)

(Fig. 3.6) as laid or by clutch. Determine **egg removal procedures** before each laying season based on differences in behavior, incubation skills, laying patterns, genetic objectives (see Chapter 9), and space available for raising chicks and housing new adults.

Single-Egg Removal. When eggs were removed as laid, one captive Florida Sandhill Crane at ICF laid 19 eggs and one Greater Sandhill Crane at Patuxent laid 18 eggs in one season. Eggs were removed as laid from nine pairs of Greater Sandhill Cranes at Patuxent in a three-year study of clutch size and laying intervals. The study birds averaged 3.0 ± 0.8 days between eggs (75 eggs) in the same clutch and 10.1 ± 4.1 days between clutches (48 clutches) (Gee 1983). The number of eggs laid varied from one to four eggs per clutch with 30% being 1-egg clutches, 45% 2-egg clutches, 20% 3-egg clutches, and 5% 4-egg clutches. The study cranes showed neither a decline in egg production nor an increase in health problems that could be attributed to maximizing egg production during the three year study.

Kepler (1978) found that single-egg removal resulted in greater egg production in Sandhill Cranes than complete-clutch removal (6.4 eggs per bird versus 5.3). ICF uses single-egg removal to maximize the egg production of experienced breeders or with females designated to be surrogate incubators. Patuxent seldom uses single-egg removal.

Complete-Clutch Removal. For first time layers and to correct problems with egg breakage (see Egg Breakage section), ICF and Patuxent leave eggs until the bird completes the clutch. Disadvantages of removing complete clutches include extended intervals between clutches, reduced egg production, and a shorter reproductive season. Disadvantages to removing eggs as laid include over production by highly fecund females and reduced completion of first clutch by first time layers. Choose the method that fits your situation, bird by bird.



Fig. 3.6. Removing eggs can stimulate cranes to lay additional eggs. Here Jane Nicolich defends as Yula Kapetanagos removes eggs from Sandhill Cranes.

Photo David H. Ellis

Possible Consequences of Multiple Clutching. Multiple clutching may effect reproductive parameters. In a preliminary study, Putnam and Russman (1987 unpubl.) reported a seasonal decline in weight from first egg to last egg in 6 of 29 cranes. Patuxent and ICF have used multiple clutching with some pairs for decades with no apparent negative effects. From our records, we believe that a minor decline in egg weight over the season is common, but does not limit productivity.

Hunt (1994) noted a slight negative effect of multiple clutching in that the last eggs of the season were slightly less likely to produce fledged chicks than earlier eggs. Stated more precisely, as egg order (number of egg in laying sequence) increased, the fledging rate decreased ($P=0.023$). This minor reduction in fledging rate, however, is more than outweighed by the added productivity coming from the additional eggs. Number in laying sequence had no effect on hatching rate.

Other effects of “extended production” in cranes may include calcium depletion, post-laying “collapse,” laying of uncalcified eggs, decreased growth rate and survivability for chicks, and reduced probability of fertility or hatchability (Koga 1976; Putnam and Russman 1987 unpubl.).

In cranes, little is known about the complex relationships between multiple clutching and stress, age, experience (several differences often occur between a dam’s first and her subsequent seasons), rearing history, and physical and behavioral abnormalities (Sturkie and Mueller 1976; Putnam and Russman 1987 unpubl.; Mirande and Archibald 1990). Early studies with other species (e.g., Koga 1955, 1961, 1976) concluded that multiple clutching increased fertility.

When multiple clutching, **monitor** dams and their eggs for abnormalities; watch for changes in behavior. Record laying date for each egg, interval length, fertility, hatchability, and measurements (fresh weight, length, and width). Minimizing disturbance is especially important around these females.

Stopping Egg Production. To stop production, merely allow pairs to incubate the last clutch (eggs or dummy eggs) for a week or more. We do not recommend this method during extremely hot or cold weather when incubation may be more stressful than egg production. During extreme environmental conditions, birds stop laying naturally although incubation behavior may persist.

Egg Breakage

Egg breaking behavior is common in many species of captive birds. It has been documented in both experienced and inexperienced crane pairs. Breaking generally occurs shortly after laying but has been observed any time during incubation. **Factors** which may predispose cranes to break eggs include disturbance around the nest site, nutritional deficiencies, laying of abnormal eggs (soft shelled or undersized), inability to incubate properly, and incompatibility with mate.

Limit human activity around pairs with a history of egg breaking. **Behavioral monitoring** helps if facilities allow unobtrusive observation: video cameras can help. Observations may reveal sources of disturbance and help guide management decisions. If the male is responsible, move him to an adjacent pen 1-2 days before the female lays. Other responses may include reducing human activity, adding visual screening, moving the pair to a different pen, or removing an adjacent pair.

Continuous monitoring often makes it possible to collect eggs before they are broken and to replace them with either unbreakable (i.e., wooden or plaster-filled; Fig. 3.7) **dummy eggs** or blown crane eggs filled with a **foul-tasting** liquid (i.e., mustard, hot [tabasco or jalapino] sauce, or methyl anthranilate). Foul-tasting dummies have been used with only limited success in an attempt to create an aversion to egg breaking/eating. Further experimentation with distasteful but non-toxic substances may prove useful. Replacement with wooden or **unbreakable** dummies has proven successful in several cases where pairs have accepted and incubated these “eggs.” Four pairs of



Fig. 3.7. *Unbreakable dummy eggs are used to replace real eggs for pairs that break eggs.*

Photo Patty McCourt

TABLE 3.4.

Timing and length of breeding season in male cranes at ICF from 1978-1990.¹

Species	Sample Size	Earliest Semen	Latest Semen	Mean Season Length (days)	Peak Production	Mean Semen Volume (cc)
Blue	1	1 Apr	8 Jul	71	17 May-20 Jun	0.04
Siberian	11	8 Mar	18 May	69	10 Apr-22 Apr	0.03
Florida Sandhill	9	20 Mar	27 May	79	3 Apr-1 May	0.03
Greater Sandhill	5	24 Mar	19 May	70	16 Apr-2 May	0.05
Eastern Sarus	8	29 May	29 Jul	62	25 Jun-24 Jul	0.04
Indian Sarus	4	1 May	29 Jul	60	30 May-6 Jul	0.04
Brolga	6	27 Apr	18 Jul	81	16 Jun-17 Jul	0.08
White-naped	8	6 Mar	7 Jun	110	12 Apr-20 May	0.10
Common	4	22 Mar	22 May	66	18 Mar-14 May	0.05
Hooded	1	24 Mar	24 Mar	0	24 Mar-24 Mar	0.03
Red-crowned	11	5 Mar	24 May	82	27 Mar-8 May	0.03
Whooping ²	7	16 Mar	14 May	59	30 Mar-26 Apr	0.03
Demoiselle	3	11 Apr	20 May	39	26 Apr-24 May	—
Wattled	2	8 Mar	10 May	76	16 Mar-18 May	—

¹ *Crowned Cranes are not included because they lay throughout the year.*

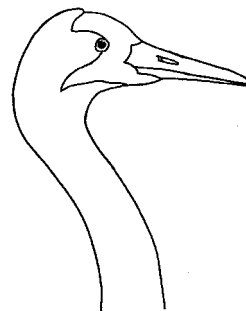
² *Data from Patuxent.*

Whooping Cranes at Patuxent broke every egg which was not immediately removed from their pens. All were subsequently given wooden dummy eggs. All pairs initially pecked, then ignored the eggs, and eventually adopted and incubated the wooden eggs. Three of the four pairs thereafter laid their own eggs and incubated without further breakage. ICF has had similar success with Wattled and Whooping Cranes. Incubation and parenting behavior is reinforced by allowing these pairs to incubate full term, and then hatch and rear a chick.

Although no research has been conducted on the relationship of diet to egg breaking, this behavior may also be linked to **nutritional deficiencies**. At ICF, we provide high protein treats (newborn mice or smelt) in food bowls or we toss them to the birds. Treats also help calm or tame the birds. Tame birds, we believe, are less prone to break eggs.

Characteristics of Semen Production

Table 3.4 summarizes crane semen production. These data provide guidelines for collecting semen. Crane semen samples are very small; the average is 0.03 to 0.10 cc for different species. Semen volume in one ejaculate can vary dramatically between males of the same species, from a smear to 0.78 cc. Measurements of semen quality and quantity are described in Chapter 11A.

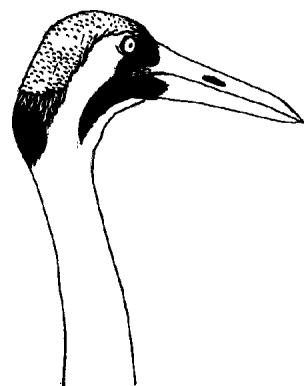


Literature Cited

- Archibald, G., and S. Swengel. 1987. Comparative ecology and behavior of Eastern Sarus Cranes and Brolgas in Australia. Pages 107-116 *in* J. C. Lewis, editor. Proceedings 1985 Crane Workshop. Platte River Whooping Crane Habitat and Maintenance Trust and U.S. Fish and Wildlife Service, Grand Island, Nebr.
- Balzano, S. A. 1989 unpubl. Climatic influences on crane production. Paper presented at the 1989 North American Crane Workshop.
- Derrickson, S. R., and J. W. Carpenter. 1987. Behavioral management of captive cranes—factors influencing propagation and reintroduction. Pages 493-511 *in* G. W. Archibald and R. F. Pasquier, editors. Proceedings of the 1983 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Einsweiler, S. 1988 unpubl. Factors affecting reproduction of the Siberian Crane at the International Crane Foundation. International Crane Foundation, Baraboo, Wis. 20 pp.
- Farner, D. S. 1986. Generation and regulation of annual cycle in migratory passerine birds. *American Zoologist* 26:493-501.
- Gauthier, G. 1989. The effect of experience and timing on reproductive performance in Buffleheads. *Auk* 106:568-76.
- Gee, G. F. 1983. Crane reproductive physiology and conservation. *Zoo Biology* 2:199-213.
- Gee, G. F., and G. W. Pendleton. 1992. Effects of extended photoperiod on Sandhill Crane reproduction. Pages 149-159 *in* D. A. Wood, editor. Proceedings 1988 North American Crane Workshop. Florida Nongame Wildlife Program Technical Report #12.
- Gee, G. F., and T. Sexton. 1979. Artificial insemination of cranes with frozen semen. Pages 89-94 *in* J. C. Lewis, editor. Proceedings 1978 Crane Workshop. National Audubon Society and Colorado State University Printing Service, Ft. Collins, Colo.
- Halibey, T. 1976 unpubl. Feed and feeding habits of captive cranes. ICF report. 19 pp.
- Hunt, M. C. 1994. Analysis of the relationship between egg order (1-15) and egg quality as determined by hatching and fledging rates in Siberian, Florida Sandhill, White-naped and Red-crowned Cranes. *Aviculture Magazine* 100(1):29-34).
- Immelmann, K. 1971. Ecological aspects of periodic reproduction. Pages 342-389 *in* D. S. Farner, J. R. King, and K. C. Park, editors. *Avian biology*. Academic Press, New York.
- Immelmann, K. 1972. Role of the environment in reproduction as source of "predictive information." Pages 121-147 *in* D. S. Farner, editor. *Breeding biology of birds*. National Academy of Sciences, Washington, D.C.
- Kepler, C.B. 1978. Captive propagation of Whooping Cranes: a behavioral approach. Pages 231-241 *in* S. A. Temple, editor. *Endangered birds: management techniques for preserving threatened species*. University of Wisconsin Press, Madison.
- Koga, T. 1955. On the artificial incubation of cranes (*Grus vipio* and *Grus japonensis*). *Journal of the Ueno Zoo* 1:1-12.
- Koga, T. 1961. Studies on the reproduction of cranes, especially on their artificial incubation and breeding. *Journal of Japanese Association of Zoological Gardens and Aquariums* 3:51-65.
- Koga, T. 1976. Increasing captive production of Japanese and White-naped Cranes. Pages 351-355 *in* J. C. Lewis, editor. Proceedings of the International Crane Workshop 1975. Oklahoma State University Publishing and Printing.
- Konrad, P. M. 1987. Rainy season ecology of South African Grey Crowned Cranes in the Luangwa Valley, Zambia. Pages 337-344 *in* G. W. Archibald and R. F. Pasquier, editors. Proceedings of the 1983 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Kuyt, E. 1981. Population status, nest site fidelity, and breeding habits of Whooping Cranes. Pages 119-125 *in* J. C. Lewis and H. Masatomi, editors. *Crane research around the world*. International Crane Foundation, Baraboo, Wis.
- Kuyt, E., and J. P. Gossen. 1987. Survival, age composition, sex ratio, and age of first breeding of Whooping Cranes in Wood Buffalo National Park, Canada. Pages 230-244 *in* J. C. Lewis, editor. Proceedings 1985 Crane Workshop. Platte River Whooping Crane Habitat and Maintenance Trust and U.S. Fish and Wildlife Service, Grand Island, Nebr.
- Lack, D. 1933. Nesting conditions as a factor controlling breeding time in birds. Pages 231-237 *in* Proceedings of the Zoological Society of London.
- Marshall, A. J. 1961. Breeding seasons and migration. Pages 307-339 *in* A. J. Marshall, editor. *Biology and comparative physiology of birds*. Vol. II. Academic Press, New York.
- Minton, C. D. T. 1968. Pairing and breeding in swans. *Wildfowl* 19:41-60.
- Mirande, C. 1994 unpubl. Genealogy of the Whooping Crane. International Crane Foundation, Baraboo, Wis. 19 pp.
- Mirande, C. M., and G. W. Archibald. 1990. Sexual maturity and pair formation in captive cranes at ICF. Pages 216-225 *in* American Association of Zoological Parks and Aquariums 1990 Annual Conference Proceedings, Indianapolis, Ind.
- Mirande, C. M., J. W. Carpenter, and A. Burke. 1989 unpubl. The effect of disturbance on the reproduction and management of captive cranes. Paper presented at the 1989 North American Crane Workshop. 18 pp.
- Morris, T. R. 1967. Light requirements of the fowl. Pages 15-39 *in* T. C. Carter, editor. *Environmental control in poultry production*. Oliver and Boyd, Edinburgh and London.
- Murton, R. K., and N. J. Westwood. 1977. *Avian breeding cycles*. Oxford University Press, Oxford, England. 594 pp.
- Nesbitt, S. A., and A. S. Wenner. 1987. Pair formation and mate fidelity in Sandhill Cranes. Pages 117-122 *in* J. C. Lewis, editor. Proceedings 1985 Crane Workshop. Platte River Whooping Crane Habitat Maintenance Trust and U. S. Fish and Wildlife Service, Grand Island, Nebr.
- Nicolich, J. 1993. Appendix 1. Patuxent captive population data. Page 97 *in* U. S. Seal, editor. *Mississippi Sandhill Crane (Grus canadensis pulla) Population and Habitat Viability Assessment Workshop*. Conducted by IUCN/SSC Captive Breeding Specialist Group.

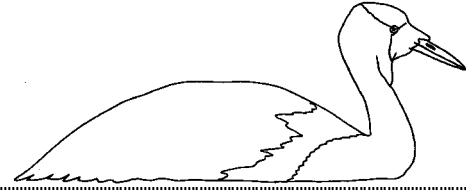
- Panchenko, V. 1993 unpubl. International Siberian Crane studbook. Oka State Nature Reserve, Ryazan, Russia. 12 pp.
- Putnam, M., and S. Russman. 1987 unpubl. Changes in egg characteristics and laying intervals in captive cranes laying multiple clutches. 42 pp.
- Richdale, L. E. 1951. Sexual behavior in penguins. University of Kansas Press, Lawrence. 316 pp.
- Sadler, R. M. F. S. 1969. The ecology of reproduction in wild and domestic mammals. Methuen and Ltd., London. 321 pp.
- Serafin, J. A., and G. W. Archibald. 1977 unpubl. Natural and synthetic diets for cranes and other Gruiformes. 7 pp.
- Sturkie, P. D., and W. J. Mueller. 1976. Reproduction in the female and egg production. Pages 306-311 *in* P. D. Sturkie, editor. Avian physiology. 3rd ed. Springer-Verlag, New York.
- Wada, M. 1984. Photoperiodism in avian reproduction. Pages 101-113 *in* K. Ochiai, Y. Arai, T. Shioda, and M. Takahashi, editors. Endocrine correlates of reproduction. Japan Scientific Society Press, Tokyo, Japan.
- Walkinshaw, L. H. 1973. Mississippi and Florida Sandhill Crane egg records. *The Cranes: a Journal Dealing with World Cranes* 1:6.
- Welty, J. C. 1975. *The life of birds*. 2nd ed. W. B. Saunders Co., Philadelphia, Pa. 623 pp.
- Wingfield, J. C. 1983. Environmental and endocrine control of avian reproduction: an ecological approach. Pages 265-288 *in* S. Mikano et al., editors. Avian endocrinology and ecological perspectives. Japan Scientific Society Press, Tokyo.





Incubation and Hatching

Robert R. Gabel and Thomas A. Mahan



This chapter compares crane egg incubation techniques and provides troubleshooting guidelines. For an expanded treatment of this subject see Stromberg (1975), Brown (1979), and Jordan (1989). The simplest approach to incubation is to permit a pair of cranes to incubate and hatch their own eggs (see Chapter 6). Circumstances may, however, require alternative methods. Genetically valuable birds may be unreliable parents that cannot be trusted with their own eggs. Eggs may also be removed from valuable birds to induce laying of multiple clutches for maximum production. Inclement weather, the threat of predators, or the unexpected death of a parent may all require that eggs be removed from their original nest. In each case, dependable substitutes for parental incubation are needed.

In planning an incubation program, consider the following: (1) size of the breeding flock, (2) reliability of electrical power, and (3) availability of space, manpower, incubation equipment, and supplies. To evaluate an incubation program, benchmarks must also be established. A reasonable hatch rate of fertile eggs is 80-85%, whether incubation is natural or artificial. Hatch rates below 80% indicate a need for improvement.

Generally, the best hatching success has been achieved for cranes using natural or a combination of natural and artificial incubation (Sullivan 1994 unpubl.). Chickens have been successfully used as surrogate incubators, although hatching rates can be lower (Mahan 1992). Chicken incubation may be the best method when crane parents and artificial incubators are not available or when power supplies are undependable.

A good hatchery manager is familiar with embryo mortality patterns, weight-loss profiles, and other egg, embryo, and hatchling characteristics that are indicative of improper temperature or humidity, or other incubation problems. This information is available in the avicultural and poultry science literature, from which many of the references for this chapter were taken.

Several factors can decrease hatchability: disease, behavioral anomalies, improper nutrition, inbreeding, and other genetic defects (see Kuehler and Good 1990; Kuehler and Loomis 1992). Consultation with veterinarians and professionals in other relevant disciplines can greatly enhance an incubation program.

Natural Incubation

The natural parents, foster parents of the same or a related species, or even unrelated species (e.g., domestic chickens) may be used to incubate crane eggs. Choose birds that are reliable incubators and whose reproductive cycle can be synchronized with that of the natural parents (see Chapter 5).

Natural incubation has several **advantages**, and hatchability for some cranes can be improved if at least the first 7-10 days of incubation are natural (Brown 1979; Erickson and Derrickson 1981; Heck and Konkel 1983; Mahan 1992). First, variation in nest temperature due to environmental temperature changes, incubation exchanges by the parents, and a temperature gradient from top to bottom of the egg are lacking in conventional artificial incubation and are believed to affect hatchability (Gee et al. 1995). Second, separate facilities are not needed for incubation and rearing if eggs and chicks remain with the parents or foster parents. Third, naturally incubated eggs are not threatened by an interruption in electrical service or mechanical failure. Finally, natural incubation, as well as subsequent chick rearing, may enhance pair bonds between birds and promote higher reproductive rates in the future (Derrickson and Carpenter 1987).

Natural incubation also has associated **difficulties and risks**: (1) contamination of eggs by feces, soil, nesting material, or other debris; (2) disease transmission from parent to egg or chick; (3) accidental or deliberate breakage of eggs by parents or foster parents; (4) predation; (5) nest abandonment (even if the

parents or foster parents have previously incubated successfully); and (6) reduced ability to monitor embryo development and egg condition. Lower production may also result if parents are used to incubate rather than recycle and produce additional clutches of eggs in the same season (Derrickson and Carpenter 1982). In addition, if foster parents are used, natural incubation involves higher costs for facilities and staff because several pairs of foster parents must be maintained year-round to care for the eggs and young of each pair of birds whose eggs are fostered. Similarly, incubation under chickens requires maintaining several hens so that at least one will be broody when each clutch of crane eggs is laid. Chickens also require a facility where the photoperiod can be controlled to stimulate egg laying and incubation to coincide with that of the cranes. A backup incubation system is needed in any event to incubate thin-shelled or cracked eggs, eggs deserted by parents, or eggs endangered by severe weather.

Parental Incubation

Each potential layer is observed two to four times each day. As the caretaker walks through the colony, he or she reviews a previously prepared form (Fig. 10.8) showing the presence and condition of eggs during the last visit and records his or her own observations. In pens with unreliable parents, new eggs, especially eggs of endangered species, are immediately removed when found. Sometimes these are exchanged for dummy eggs to stimulate incubation (see Chapter 3).

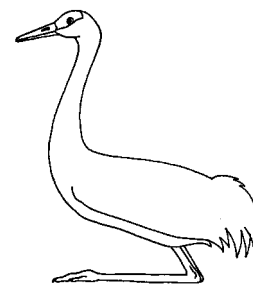
When first handling eggs, mark each with an identification (ID) number, weigh, and measure (length and width). Patuxent **disinfects** each egg by dipping in a 10% Betadine or similar povidone-iodine solution, or in quaternary ammonia or other non-toxic disinfectant at 43° C (110° F), as soon as possible after laying. ICF does not disinfect and has 77-84% hatchability (Sullivan 1994 unpubl.). Disinfection is particularly important when breeding birds are in enclosures that have been used for several years and therefore may have a burden of soil pathogens.

During incubation, observe parents from a blind or at a distance to determine nest attendance. If the parents are frequently off the nest, especially in cold weather, consider removing or replacing the eggs and using another method of incubation until conditions improve. Determine egg fertility by candling, if possible, 5 to 7 days after oviposition (see Fertility Determination section, this chapter). This is also a

good time to remove the eggs if the parents are to lay another clutch (see Chapter 3). Weight loss of the egg should also be monitored throughout incubation. Around the 20th day of incubation, Patuxent uses **flotation** to check the viability of eggs remaining with the parents (technique described later). ICF candles a second time and only uses flotation if candling is unsuccessful or embryo death is suspected. Once you are certain an embryo is not developing, remove infertile or nonviable eggs and open them for bacterial culture and examination of contents. The eggs may be replaced with artificial eggs or other viable eggs to keep the pair incubating.

One or two days before **hatching**, the chick becomes active, punctures the air cell, and becomes vocal (Hartman et al. 1987). The incubating parents communicate with the hatching chick by purring frequently, and they spend more time hock-sitting rather than lying in the nest. More frequent nest checks are advisable at this stage if the adult birds are not unduly agitated. Make at least one close inspection as soon as possible after the chick pips (i.e., when the first break in the eggshell occurs, usually indicated by noticeably louder cheeping from the nest) to see if the pip is in the correct position (at the large end of the egg). Additional inspections can be made with binoculars from a distance of 15-30 m. If an egg has been pipped for more than 48 h but has not yet opened, enter the pen to see if the inner eggshell membranes have dried and are adhering to the chick, thereby preventing hatching progress. **Chick deformities** can also prevent hatching. Problem eggs are best dealt with by moving them to the artificial incubation facility (see Hartman et al. 1987; also Assisted Hatches section in this chapter).

Check the chick as soon as possible after it hatches to determine if the yolk sac has been completely absorbed into the body cavity (see Chapter 5). If the umbilicus is closed (with the yolk sac in the abdomen), apply iodine solution to the site to prevent infection. Weigh the chick to establish a reference point for early growth evaluations.



Surrogate Incubation by Cranes

The surrogate pair must have eggs of their own that are at approximately the same stage of incubation as the fostered eggs (usually within 10 days of synchrony). This is easiest to arrange if the foster species breeds near the same time as the donor. Otherwise, the breeding cycles of one or both species must be altered by manipulating the photoperiod with artificial light, recycling the foster parents, or both. If surrogate parents are in short supply, move eggs to artificial incubators after 7-10 days. This allows pairs to incubate up to three sets of eggs in a single season (see Egg and Chick Adoptions in Chapter 5).

Surrogate Incubation by Chickens

Incubation of rare or exotic species by chickens is standard practice in aviculture, especially for the production of game birds (pheasants, quail, etc.; Brown 1979; Heck and Konkel 1983). Chickens receiving crane eggs for surrogate incubation must be in reproductive synchrony with the crane pairs. Unlike cranes, which have an incubation period of about 30 days, the normal incubation period for chickens is 21 days. Although a chicken's incubation period may be extended to 40 days, the behavior and physical condition of the hen should be closely monitored to prevent nest abandonment and to avoid impairing the health of the hen.

Use large or "standard" chicken breeds rather than bantams (which may be unable to cover the eggs completely) as surrogate incubators for cranes. Because crane eggs are much larger than chicken eggs, it is unlikely that chickens can adequately turn crane eggs. As a result, eggs under a chicken should be turned by hand at least four times per day (see Turning under Mechanical Incubation). Suggested chicken breeds for crane egg incubation are Brahmas, Langshans, and especially Cochins. Other breeds of large-bodied chickens may also be used, but commercial strains of these breeds are unlikely to be good incubators. Particularly good incubating strains are available from poultry hobbyists. Day-old chicks of these exhibition-type breeds (some of which are good incubators) are available from commercial hatcheries. Obtain chicks hatched in spring or summer for use as incubators ("broodies") the next year. All chickens should be quarantined and health tested before introducing them into the collection.

Males are needed for breeding, but are not necessary for hens to incubate. Only a few males are needed because one male can easily inseminate 7-10 hens. Producing your own replacement chicks avoids the risk of introducing disease and allows for selection of stock with good incubating qualities. Although unrelated stock must be obtained periodically to minimize inbreeding, such additions need not be frequent because healthy hens can be used for 5-7 years or longer. Indeed, experienced, older hens are the most valuable birds in a surrogate incubation program.

Maintain chickens in a well-ventilated enclosure that provides about 1 m² of floor space per adult bird (enough space for the birds to move about freely and remain unsoiled). Use hardwood chips, shavings, or other dry, relatively dust-free bedding. Prevent bedding from becoming wet or damp; immediately remove damp bedding to avoid growth of fungi and bacteria. If possible, provide an outdoor yard for exercise and a more interesting environment. This practice is believed to provide the birds opportunities for normal behavior and prevent feather plucking, cannibalism, or other destructive activities. Keep exercise areas clean and tightly enclosed to prevent the entry of predators, vermin, or wild birds that could introduce disease. Test and treat for internal parasites before the chickens are put into an outdoor enclosure so the ground does not become contaminated and serve as a source of reinfection.

Chickens are able to withstand fairly cool and warm temperatures, but should be protected from temperature extremes. Provide shade and good ventilation during warm weather and enough heat during extreme cold to prevent frostbite and freezing of drinking water (preferred minimum temperature ca 10° C [50° F]).

Controlled lighting can be used to bring hens into production and incubating condition to coincide with crane egg production. About 4 months before hens are needed for incubation, adjust the **photoperiod** to 8 h light:16 h dark/day for 8 weeks. Thereafter, change the lighting regime to 14 h light:10 h dark/day to stimulate egg production. Egg production should begin about 3 weeks later, and some hens should become broody about 2-3 weeks after the onset of laying. There is considerable variation among birds, however, and some may never become broody. Whenever the natural day length (i.e., light period) exceeds 8 h/day, eliminate access to any natural light or artificial short-day photoperiods will be ineffective. If broody hens are needed over more than 1-2 months, it may be

necessary to maintain two or more groups of chickens on different lighting schedules in separate enclosures. Prevent light leakage between such enclosures.

To prevent competition for nest sites, provide **several nests** for each group of hens. Line nest bottoms with felt-type indoor-outdoor carpeting, and fill them with chopped straw or similar material (e.g., clean, fine grass) to a depth of 5-7 cm. Hens arrange this into a cup shape for laying. Change nesting material monthly, or immediately if it becomes damp or soiled with feces or broken eggs.

Maintain the chicken flock on a high-quality **diet**, readily available from commercial livestock supply stores. Follow the manufacturer's instructions regarding the appropriate diet to feed at each age or stage of production (e.g., layer diet for hens in production). Provide feed *ad libitum* along with clean, fresh water. Scratch (i.e., cracked corn, wheat, or other grain) may be offered as a treat so that the birds become tame and are easily approached. However, because commercial poultry diets are designed to be nutritionally complete, feeding more than small amounts of scratch can lead to a dietary imbalance. Layer diets generally contain elevated levels of calcium, but crushed oyster shell can be provided as a calcium supplement for eggshell production (Brown 1979).

After laying begins, remove eggs daily to prevent breakage. Broken eggs soil the nest and hen, and may lead to habitual egg-eating. Place two or three **dummy eggs** in each nest before the onset of laying to encourage hens to use the nests and to stimulate incubation.

The combs of young and nonlaying hens are small and dull orange, whereas the comb of a laying hen enlarges and becomes bright red. In addition, the abdomen of the laying hen becomes enlarged, the vent becomes large and moist, and the spread between the pubic bones increases from about the width of one finger to two to three finger widths. Laying hens will also crouch in a sexually receptive position when approached.

Birds change in both appearance and behavior when they become broody. Their combs and other physical characteristics return to the condition of a nonlaying hen. They remain in the nest almost continuously and only leave briefly to eat, drink, defecate, and exercise, whereas laying hens are usually found in the nest only in the morning, when most laying occurs. Laying hens will generally leave the nest readily when disturbed, whereas broody hens leave the nest reluctantly and may become extremely defensive. This behavior varies with each hen, so become

familiar with each bird's idiosyncrasies to be certain that a hen is broody. When removed from the nest for egg examinations, broody hens often sit where they are placed and refuse to walk about. They elevate their hackles and squawk when approached by another bird or a caretaker, and emit an almost continual cluck when moving on their own or when disturbed.

If possible, move incubating hens to a separate room or building. Maintain this area at light and temperature conditions to which the hen is accustomed (or slightly warmer, i.e., 15° C [60° F]). Place each broody hen in a **lock-box nest** (see Fig. 4.1) where she will remain most of the day. Place an indoor-outdoor carpet liner in the nests similar to the laying nests, and form a cup-shaped straw or grass nest by hand before the hen is placed in the nest. When a hen is first moved to the broody house or room, put dummy eggs under her for 2-3 days to make sure that she continues incubating after the move. Let the hens out every morning for 30-45 min to eat, drink, defecate, and exercise. Observe the group of hens for aggression if they have not been previously housed together, and deal with serious encounters by separating the birds. Aggressive encounters may be reduced by providing the birds with multiple feeders and waterers separated by 2 m or more.

Observe broody hens in the morning when they are released into the exercise yard and again in the afternoon. These observations are especially important for hens that have just started incubating or that have been sitting for more than 3-4 weeks. Hens that defecate in their nests, or that do not sit tightly and appear eager to leave the nest, are no longer broody. If hens do not sit tightly after they are moved to the broody area, move them back to the laying house until they develop stronger incubation tendencies.



Fig 4.1. Lock-box nest for chickens used to incubate crane eggs.

Photo Patuxent

Special care is necessary to prevent egg breakage and to avoid injury when removing the hen. Lift the broody hen from her nest by gently sliding one hand under her breast and placing your other hand on her back. Slowly lift the hen up and out of the nest and set her on the floor. Be careful to prevent her from kicking eggs out of the nest. Close nest box doors while the hens are out to prevent them from flying into their own or another nest, thereby accidentally damaging the eggs. When returning hens to the nests, gently lift them to the edge of the nest and allow them to step in and settle onto the eggs. Hens often become so accustomed to this routine that they may approach the caretaker to be returned to the nest. As you handle each hen, evaluate her general body condition, and return any hen that is becoming too thin to the laying house or, ideally, to a separate area where she can lose her broodiness and regain weight.

Although crane eggs may remain under hens for the entire incubation period, the benefits of hen incubation are realized by about the tenth day of incubation. Thereafter, the eggs may be incubated artificially. Some hens recycle and are available for incubation a second time within the same season, although others molt after the first incubation cycle and do not return to production. Factors controlling these different cycles probably include ambient temperature, other environmental conditions, and the genetic make-up of the hen.

Mechanical Incubation

Critical Components

Temperature control. Reliable and consistent incubator operation requires a system with dual temperature controls consisting of primary and secondary thermostats. The primary thermostat controls temperature during normal functioning of the incubator. The secondary thermostat assumes control if the primary thermostat fails. To monitor temperature, place calibrated thermometers inside the incubator. These thermometers must be readable from outside the closed incubator.

Small temperature variations affect embryo survival because normal development occurs within a very narrow temperature range. Detrimental effects of incorrect temperatures, however, depend on duration, direction (too high or too low), and stage of development. Sharp

temperature increases of only several degrees can be almost immediately lethal. A temperature increase of only 1-1.5° C (2-3° F) may not be immediately fatal, but embryos are likely to die after only 4-5 days. Low temperatures (but still within rather narrow limits) slow development and delay hatching, but do not increase mortality substantially (Brown 1979).

Minor temperature variations exist within an incubator, especially among egg trays at different levels. Measure temperatures in different parts of the incubator annually with thermometers accurate to within 0.1° C (0.2° F). By moving the thermometers to different places in the incubator, you can actually map temperature variation within the machine (Heck and Konkel 1983). The stability of temperature conditions inside the incubator depends on ambient conditions, and therefore requires a stable temperature inside the incubator room or building. Remove or add eggs relatively quickly, but carefully, because prolonged or repeated opening of the incubator door can cause the temperature to drop significantly (Burnham 1983).

Humidity. The humidity level inside the incubator controls egg weight loss. Incubator humidity is controlled by the addition of water through evaporation from a reservoir, by misting, and by regulating air flow through the incubator or through the incubator room from outside. For incubators that are humidified by evaporation of water from a reservoir (tray), higher humidity is achieved by increasing the surface area of the water (i.e., by increasing the length and width of the tray, using rotating fins, or placing sponges in the water reservoir). Placing the fan so air blows directly across the water surface also increases evaporation. Use distilled water to avoid mineral buildup, although large incubators often have flow-through humidifying systems for which the use of distilled water would be costly.

Vent openings control air flow into the incubator and thereby reduce (vents open) or increase (vents closed) relative humidity. Except during fumigation, do not completely close the vents because developing embryos need a constant influx of fresh air. Humidity is also lost when the incubator door is opened, so open the machine only when necessary and for as brief a time as possible. Humidity losses are even more important than temperature decreases, because desired humidity levels are not restored as quickly as temperature. To minimize this problem and to rapidly restore humidity in the incubator, especially large incubators, lightly spray the incubator floor with water before closing the door.

Before eggs are set in an incubator, thoroughly clean all inside surfaces of the incubator, including egg trays, with a bactericidal and fungicidal disinfectant. The electronics and wiring are cleaned with compressed air or a light spray with the disinfectant. After cleaning, allow electrical components to dry, then turn the incubator on to raise the inside temperature to 26-37° C (80-100° F) and increase relative humidity to 50-60%.

After establishing operational conditions, the incubator may be fumigated with formaldehyde by combining 35 mL of 40% formalin with 17.5 g of potassium permanganate per 2.83 m³ (100 ft³) of incubator volume. Although it is very effective for disinfection of incubators, fumigation with formaldehyde gas requires great caution and good ventilation because formaldehyde is carcinogenic and a strong irritant to human eyes and respiratory tracts. For this procedure, we recommend the use of goggles and a mask (respirator). Be sure that the incubation facility is vented to the outside to keep fumes from escaping to other indoor areas. If appropriate handling of formaldehyde is not possible, the use of commercially available spray disinfectants is advisable to avoid health risks.

Use only porcelain, earthenware, or heat-tempered glass with the fumigation reagents because these chemicals cause a violent exothermic reaction. The reagents interact with some metals and melt or burn plastic and other flammable materials. Before fumigating, close the incubator door and vents. Place the potassium permanganate in the container first. Then place the container inside the incubator, add the formalin, and immediately close the incubator. After 20 min, remove the fumigant and leave the incubator open for a few minutes to air out. The machine is then ready to receive eggs. For incubators with a recent history of poor hatches from disease or that have had rotten eggs break or explode inside the machine, use a double-strength mixture of the chemicals.

Fumigate incubators at least every 2 weeks, even during incubation. If eggs are added to the machine frequently (every few days or less), fumigate weekly to eliminate contamination from new eggs. Only eggs in very early incubation (5 days or less) or whose stage of incubation is unknown should be removed and placed in another machine during fumigation. If another incubator is not available, any heated container that maintains the eggs at 35.0-37.2° C (95-99° F) will suffice for a short time. Otherwise, fumigate fresh (unincubated) eggs and eggs incubated more than 5 days to kill any pathogens on the shell. Clean and fumigate hatch-

ers after each use, even if only used for one chick, according to the same procedure described above. If two more eggs are in a hatcher, fumigate after all eggs are hatched and all chicks removed. If fumigation is not used, employ other disinfectants, but be sure to determine efficacy and safety for use with eggs.

The use of sterile surgical gloves or clean disposable (plastic, vinyl, or latex) gloves to handle eggs, dipping of eggs before they are placed in the incubator (see Egg Handling section below), and regular cleaning and disinfection of incubator water trays also helps to ensure good sanitation.

Record keeping. Incubator conditions (temperature, humidity, tray position, etc.) should be closely monitored and recorded at least two or three times daily. This will ensure that any trends, such as increases or decreases in temperature or humidity, are detected early and corrected. It is important that incubators with automatic turners be checked at different times during the day to determine if they are actually turning the egg. These records serve as a basis for making adjustments in incubation conditions if hatching problems arise.

Egg Handling

After an egg is collected, any dirt or fecal material adhering to the shell is wiped off with a soft cloth. Fine-grade sandpaper can be used to remove stubborn materials, but be sure not to damage the shell. To prevent the introduction of pathogens into the incubator, fumigate incubated eggs according to the procedures described previously. Do not fumigate eggs unless they are fresh or have been incubated for at least 5 days. As an alternative disinfection procedure for incubated eggs or those whose stage of development is unknown, dip them in 10% povidone-iodine solution at 43.3° C (110° F; Ernst 1975) and then allow them to dry at room temperature before setting them in the incubator.

Artificial Incubation Conditions

For all crane species, the proper dry-bulb temperature is 37.6° C (99.5-99.75° F; Putnam 1982). Preliminary research on regular **cooling** of crane eggs during incubation showed no significant effect on hatchability (Putnam 1982; Russman 1987). We recommend leaving eggs in the incubator continuously except for candling, fumigation, etc., as discussed elsewhere in this chapter.

The wet-bulb temperature used for all but two species of cranes is 30.0°C ($85\text{--}87^{\circ}\text{F}$). For White-naped Cranes, the wet-bulb temperature used is 27.0°C (80.0°F ; Putnam 1982), and Wattled Crane eggs have been successfully incubated at a wet-bulb temperature of 28.0°C ($82\text{--}84^{\circ}\text{F}$; Carol Hesch, Memphis Zoological Garden and Aquarium, Memphis, Tennessee, personal communication). At higher altitudes with lower air pressure, wet-bulb temperatures should be adjusted slightly (1°C , $1\text{--}2^{\circ}\text{F}$) upward or in areas with high relative humidity, downward to achieve the desired rate of weight loss.

During incubation (natural or artificial), carefully monitor the progress of the developing embryo by candling and flotation (see Determination of Fertility). Weigh the most valuable eggs twice weekly to determine **weight loss** during incubation. Record egg weight, and track weight loss. Optimally, eggs lose 15% (range, 13–17%) of their fresh weight over the incubation period (Rahn and Ar 1974; Ar and Rahn 1978), although eggs that lose considerably more or less than this amount often hatch, either independently or with assistance (see Problems and Remedies). Conditions that can reduce egg weight loss include high humidity, low temperature, a thicker-than-normal shell, or blocked pores in the shell. Conditions that can increase weight loss are low humidity, high temperature, or an abnormally thin or porous shell.

Candle eggs one to three days before the scheduled hatching date to determine viability and locate the air cell. Mark the lowest point of the air cell (i.e., the point that extends farthest toward the small end of the egg). The egg is placed with the marked spot up (Putnam 1982). If positioned correctly, the chick will pip near this point. Turning of the egg is no longer necessary during the last 2 days of incubation, so the egg may be placed in a depression on a styrofoam pad on the bottom of the incubator.

Alarms. An important component of the artificial incubation system is an alarm that notifies personnel of temperature extremes and power outages. Set the alarm to sound at temperatures above 38.3°C (101°F) or below 35.6°C (96°F), and when the power fails. In addition to an audible alarm (bell or siren) in the vicinity of the incubation facility, install an alarm with a telephone autodialer that notifies personnel of problems when they are off duty or away from the incubators. A flashing light can also be added to the system to serve as a visual alarm.

Artificial Hatching Conditions

Crane chicks normally hatch after about 30 days of incubation (see Table 4.1). After an egg pips, move it to a hatcher (Fig. 4.3) maintained at 37.2°C ($98.5\text{--}99.0^{\circ}\text{F}$; Putnam 1982). A hatcher is a modified incubator maintained at a higher humidity to facilitate hatching. Use a separate hatcher for each chick, or if two or more chicks are hatching at the same time, they may occupy the same hatcher if the chicks can be separated inside the machine. Do not place additional piped eggs in the used hatcher until it is cleaned and disinfected. Debris from hatched chicks provides a growth medium for bacteria and other pathogens that thrive in the heat and humidity of the hatcher. Hatching eggs in a machine separate from the incubator prevents contamination of eggs that are at earlier stages of incubation.

During hatching, eggshell membranes may become dry and adhere to the chick. Therefore, maintain the hatcher at the **highest humidity possible**. Generally, humidity inside the hatcher should yield a wet-bulb temperature of 32°C (90°F) or higher (Putnam 1982).

The chick may be heard scratching and cheeping inside the egg for up to 24 hours or more before pipping. The vigor of these activities is a good indicator of the chick's strength. Vocalization begins after the chick breaks through the inner membrane into the air cell and begins to breathe air. The egg may be moved to the hatcher at this time or after the chick has pipped. Move eggs with chicks in the air cell to the hatcher if they will not be checked again for several hours (e.g., overnight). Otherwise, the chick may hatch in the incubator and die from trauma if struck



Fig 4.3. Dan Sprague demonstrates hatcher. Photo David H. Ellis

TABLE 4.1.

Incubation Periods of Crane Eggs

Species	Number of Days (range; mean) ¹	References
Black and Gray Crowned	26-31; 28	Carthew 1966; Walkinshaw 1965; Walkinshaw 1973; Urban et al. 1986; ICF records
Wattled	32-40; 33	Conway and Hamer 1977; Johnsgard 1983; Urban et al. 1986; Breiby 1994 unpubl.; ICF records
Blue	29-33; 30	Van Ee 1966; Johnsgard 1983; Urban et al. 1986; Hartman et al. 1987; ICF records
Demoiselle	27-30; 28	Stehlik 1970; Johnsgard 1983; Urban et al. 1986; ICF records
Siberian	26-32; 29	Johnsgard 1983; Hartman et al. 1987; Breiby 1994 unpubl.; ICF records
Sandhill	27-35; 30	Johnsgard 1983; Hartman et al. 1987; ICF and Patuxent records
White-naped	30-33; 30	Johnsgard 1983; Hartman et al. 1987; ICF records
Sarus	31-36; 31	Johnsgard 1983; Archibald and Swengel 1987; Hartman et al. 1987; ICF records
Brolga	28-36; 30	Johnsgard 1983; Archibald and Swengel 1987; ICF records
Eurasian	28-31; 30	Glutz von Blotzheim 1973; Cramp and Simmons 1980; Johnsgard 1983; ICF records
Hooded	28-30; 29	ICF records
Black-necked	30-33; 30	Johnsgard 1983; Liao 1987; Breiby 1994 unpubl.; ICF records
Red-crowned	29-36; 32	Johnsgard 1983; Hartman et al. 1987; ICF records
Whooping	28-34; 29	Johnsgard 1983; Kuyt 1987; Breiby 1994 unpubl.; ICF and Patuxent records

¹ Extremely long incubation periods (e.g., 39-40 days reported for Wattled Cranes) may represent erroneous field observations or eggs exposed to extreme environmental conditions or improper incubation conditions.

by moving parts. **Place** eggs in the hatcher with the **pip up**. This position is believed to facilitate hatching by the chick (Burnham 1983). In addition, this position allows observation of hatching progress through the glass lid or door of the hatcher.

If a chick's cheeping becomes less audible or its movements less vigorous, the chick may be weakening and unable to hatch. Check the egg four times daily and record the chick's progress and condition. The chick can be stimulated by tape recordings of crane brooding calls. Also, human imitations of this purring or gargling sound may stimulate the chick to vocalize or move. Crane chicks usually hatch between 24 and 36 h after they begin cheeping or scratching (Hartman et al. 1987). Consider helping the chick from the egg if it fails to hatch within 48 h or becomes noticeably weak (see Problems and Remedies).

Compartmentalize the hatcher to prevent mixing of chicks. Vinyl-coated wire dividers are nonabrasive to chicks and allow air to circulate. Line the bottom of each compartment with a removable piece of indoor-outdoor carpeting. This provides a good substrate and prevents the chick from catching its toes in the wire floor. Be sure that the dividers are tall enough to prevent injury or chicks mixing together.

Determination of Fertility

It is important to determine whether eggs are fertile or infertile for several reasons. First, consistent infertility may indicate improper management, disease, inbreeding depression or other genetic disorders, or

physiological or behavioral problems in the breeder flock. Second, early embryo mortality attributable to pathogens, egg handling, or incubator management may be mistaken for infertility and remain uncorrected. Third, infertile eggs can be removed from nests or incubators and replaced with fertile eggs so that incubator space or incubating birds are used to full advantage. When parents incubate their own eggs, early determination of infertility allows the eggs to be removed so the birds can lay another, hopefully fertile, clutch of eggs (Brown 1979; Erickson and Derrickson 1981).

Candling

Candling is an established technique that is especially useful for determining fertility and monitoring the development of embryos in eggs with lightly pigmented or unpigmented (white) shells. It requires a lamp inside a box or tube, and a dark room. To use a candler, hold the egg with the large end slightly above the horizontal and against a hole in the box or at the end of the tube so that light from the lamp passes through the egg (Fig. 4.4). For dark-colored eggs in particular, the candler hole should fit tightly around the egg to prevent light leakage. Light that illuminates the outer surface of the egg obscures egg contents. Even with the proper equipment, some crane eggs are too heavily pigmented for candling.

Candling is only useful for determining whether or not an embryo is developing inside the egg. It is not useful for distinguishing between infertile eggs and fertile eggs that fail to develop, or for detecting embryos that die very early. **Embryo development** is detectable by candling as early as 4 days after the onset of incubation in lightly pigmented eggs, but may not be detectable until 5-7 days in some eggs. Development is first recognized as a faint, weblike network of fine blood vessels radiating from a central focus, which is the embryo. The embryo will float to the uppermost position in the egg. At this stage, check to see if the embryo moves freely by **gently** rotating the egg; if it does not move freely, it has adhered to the shell membrane and may die. Initially, the blood vessels appear in a circular pattern about 2-3 cm in diameter. As the embryo grows, the blood vessels become larger, extending around the yolk and eventually throughout the egg. As the embryo enlarges, it blocks light passage through the egg. In light-colored eggs, you may be able to detect movement, and the head, wings, and legs may be

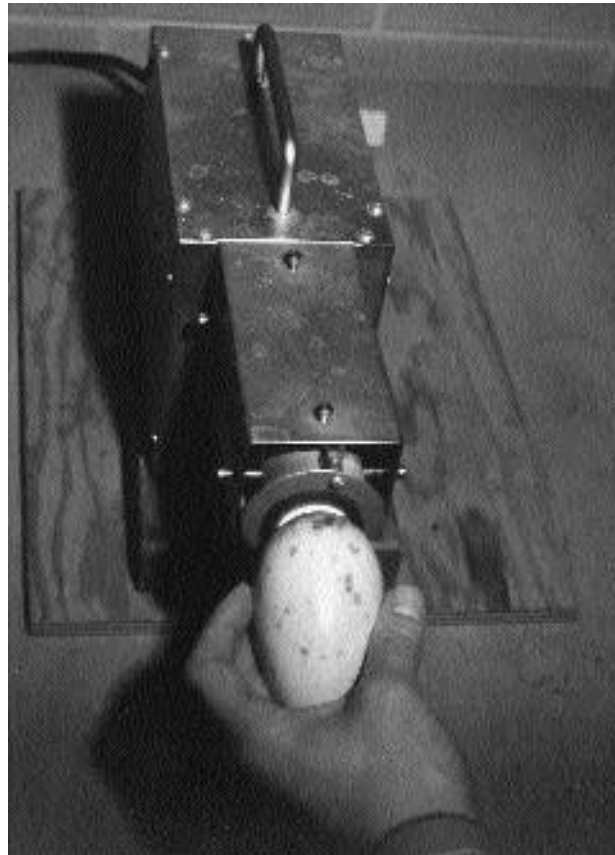


Fig 4.4. *Candling to determine fertility and viability of an egg.*

Photo Patuxent

discernible as they develop. In heavily-pigmented eggs, only light and dark areas within the egg may be distinguished, and increased opacity may be attributable to a viable, growing embryo.

Coloration of the egg contents during candling is often diagnostic for fertility and viability. A nearly clear egg with a large orange central area (the yolk) is either infertile or not incubated long enough to detect development, or contains an embryo that died in the very early stages of development. An egg with an overall reddish or pinkish hue is fertile, and the embryo is probably alive. Splotchy yellow and brown egg contents indicate a dead embryo or infected egg. For eggs in early incubation, a dark red ring indicates early embryo death or the imminent death of the embryo.

Candling can also be used to monitor the size and position of the **air cell**. The air cell should enlarge during incubation as water is lost through evaporation and respiration (Brown 1979). If eggs cannot be weighed, changes in the size of the air cell can be used to indicate weight loss indirectly. The rate of weight loss and size or shape of the air cell are not indicators

of fertility. However, an irregular or poorly defined air cell often indicates an infertile egg or an early dead embryo. Malpositioning of the air cell or a floating air cell (i.e., one that moves as the egg is turned) are often caused by jarring of the egg and may cause difficulties in development or hatching (see Problems and Remedies). Late in incubation, the margin of the air cell shifts. Near hatching, movement of the embryo in the air cell is evident.

Flotation

Flotation (Fig. 4.5) is an alternative to candling for dark-colored eggs or where an adequate light source is unavailable. This method can be used to determine fertility and viability of eggs after about 21 days of incubation, and may also be used to determine age of eggs (Fisher and Swengel 1991). At this stage, eggs float nearly vertically, with the large end up. From 21-23 days, only a slight rotational movement of the egg is noticeable. When they first appear, these movements can be obscured by movement of the flotation vessel or even a slight breeze. Nearer to hatching, stronger, twitching movements are apparent.

Float eggs in a mild **disinfectant** solution (10% povidone-iodine or equivalent) at 43° C (110° F; Ernst 1975). Observe the egg for movement for 1 min or less. Floating the egg longer risks asphyxiation or overheating of the embryo. Do not float eggs in cool water lest the egg contents contract and draw bacteria through the shell. Avoid floating eggs frequently because of potential damage to the embryo or the protective cuticle on the eggshell. Fresh disinfectant solution should be used each time eggs are floated. An initial determination of fertility and viability can be attempted after about 20-21 days of incubation. If no movement is detected, continue incubating the egg and float it again in 1-2 days. Embryos can become quiescent and fail to move when floated, so a lack of movement is not a definite indicator of embryo death. Float the egg again after a day or two if pipping does not occur when expected to determine if the chick is still alive. From about day 25, purring to the egg while it is being floated or while on a flat, sterile surface may stimulate movement.

Opening and Examining an Egg

The only way to accurately determine the fertility of an unincubated egg or one that fails to develop is to open a hole in the shell or break the egg into a dish to

reveal the germinal disk on the yolk. Do this carefully so that the yolk remains intact. In infertile eggs, the **germinal disk** appears as a small whitish spot on the yolk, about 2 mm in diameter (Fig. 4.6). In fresh fertile eggs (Fig. 4.7), this spot is a 4-mm-diameter hollow ring (donut-shaped), darker in the center. The disk enlarges as the egg is incubated. By 3 days of incubation, the heart and blood vessels become evident, even though they may be indiscernible by candling (Fig. 4.8). If non-viable eggs are to be examined more than a few hours after termination of incubation, refrigerate at 4-10° C (40-50° F).

For embryos determined to be dead by candling or flotation, opening the egg can often reveal the probable cause of death or at least the stage of development at which the chick died. Figures 4.9 and 4.10 are included for reference in evaluating embryo developmental stages. It is also useful, in evaluating necrotic tissue, to know that feather follicles first appear around day 15. Consistent mortality at a given stage of development can be diagnostic of improper incubator conditions. For further discussion of **egg necropsy** techniques, see Langenberg (1989) and Joyner and Abbott (1991).



Fig 4.5. *Egg flotation to determine viability.* Photo David H. Ellis

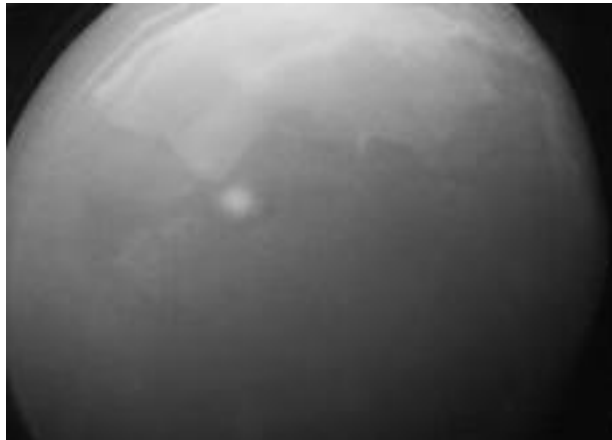


Fig 4.6. *An infertile chicken egg.*

Photo Cornell University



Fig 4.9. *A 12-day-old crane embryo.*

Photo Glenn H. Olsen



Fig 4.7. *A fertile, unincubated chicken egg.*

Photo Cornell University



Fig 4.10. *A 23-day-old crane embryo.*

Photo Glenn H. Olsen



Fig 4.8. *A 3-day-old chicken embryo.*

Photo Cornell University

Problems and Remedies

The response to incubation and hatching problems often requires innovation, creativity, and familiarity with the techniques used for other species. The following summarizes the most common incubation problems and established remedies.

Malpositions

Normally at hatching, the chick positions itself with its back parallel to the long axis of the egg, its tail in the small end of the egg, and its head turned underneath the right wing so the beak points into the air cell (Fig. 4.11). Failure of the chick to orient properly interferes with normal hatching and can lead to death of the chick.

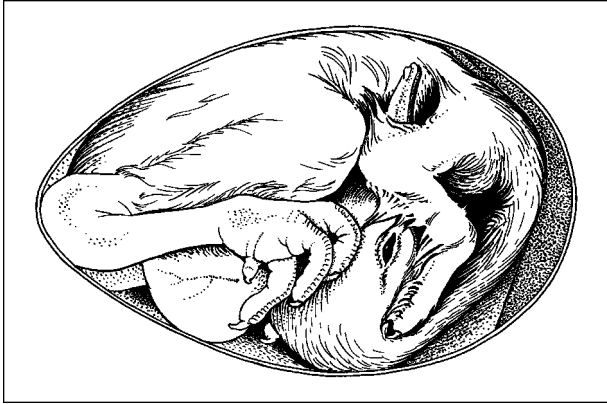


Fig 4.11. *A crane chick in normal hatching position.*

Art Kate Spencer

In chickens, malpositions have been estimated to cause 50-55% of mortality in the last 3 days of incubation and 25% of total embryo mortality (Sanctuary 1925). Embryo malpositions and their effects on hatchability have been well characterized in the poultry literature (see Landauer 1967). Although some types of malpositions are attributable to other causes, frequent occurrence of certain malpositions may be related to **egg position** and **turning**.

In chickens, elevation of the large end of the egg during incubation has been associated with an increased incidence of certain types of malpositions: (1) failure of the chick to tuck its head, (2) tucking of the head under the left wing, and (3) tucking of the head with the beak over the wing. Elevation of the small end of an egg or incubation in a horizontal position can cause orientation of the chick away from the large end so it fails to pip into the air cell (Hutt and Pilkey 1934; Talmadge 1977). The incidence of malpositions can generally be decreased by increasing the frequency of turning, especially during the last third of incubation. However, if certain types of malpositions persist, consider changing the position of the eggs.

Not all malposition types are lethal (Hutt and Pilkey 1934). Do not modify incubation methods if chicks are only occasionally malpositioned (<2%). However, the occurrence of a consistent type of malposition frequently calls for evaluation of the incubation protocol and appropriate modifications. Relatively few malpositions of crane chicks have occurred at Patuxent or ICF under the conditions specified earlier. Crane eggs may be less susceptible to malpositioning than other species due to their elongate shape. Research on chicken eggs has shown that more elongate eggs with easily distinguishable large

and small ends have higher hatchability and a lower incidence of malpositions than do eggs with indistinguishable ends (Benoff and Renden 1980). At Patuxent, eggs are positioned in incubator trays at a 20-30° lateral angle when the trays lie flat. When the tray is tilted forward, the large end of the egg is elevated 20-30°; when the tray tilts toward the back of the machine, the small end is elevated 20-30°. At ICF, eggs are incubated lying horizontally, so that the large and small ends of the egg are at the same level.

Shell Abnormalities and Weight Loss

Eggs with abnormally **thin shells** normally lose water too rapidly during incubation, whereas **thick-shelled** eggs retain too much water. Changes in egg weight can be regulated by increasing the humidity in the incubator to lower weight loss or by decreasing humidity to increase weight loss. Extremely thin-shelled eggs may also be dipped in sterile water at intervals of a few hours, or daily as needed, to maintain normal weight loss. The dip should be cooler than the egg (ca 10° C; 50° F) so the egg contents contract and draw water into the egg. Putnam and Wentworth (1986) used this technique on a Whooping Crane egg and were able to reduce the projected weight loss by 22%. Although they did not begin until two-thirds of the way through the incubation period, they slowed and even reversed the weight loss by dipping for 5 min/day in sterile water. For the first dip, 3 mg tylosin tartrate were added per liter of dip solution to prevent infection of the egg. For this procedure, adjust the duration and frequency of dipping according to the degree of weight loss and stage of incubation. Duration should be 5 min or less (shorter is preferable) to avoid stressing the embryo. Continually monitor the weight loss of eggs treated in this manner and make further adjustments in the treatment as needed.

Cracked and Damaged Eggs

Eggs that have been cracked or damaged can often be repaired. Although repaired eggs must be artificially incubated, they often develop normally and hatch.

Repair eggs with fine hairline cracks simply by applying surgical-grade cyanoacrylate (e.g., Nexaband; see Appendix) or candle wax along the crack to seal and strengthen the shell. Other non-toxic adhesives may also be suitable, but should be tested on expendable eggs, and should only be applied to the damaged

part of the shell. Extensive sealing of the shell can result in asphyxiation of the embryo. Apply wax by melting and dripping it from a burning candle. Also use candle wax to seal over larger cracks and shattered or crushed shells. If large areas of the shell have been crushed, bone cement may be used for repairs by applying a thin layer over the affected area.

To repair holes in the shell, a piece of sterilized eggshell may be glued over the hole; otherwise, parafilm, tissue, or gauze may be layered over the hole with glue (see Stoodley and Stoodley 1983; Jordan 1989). Restrict repairs to the affected area so that large areas of the shell do not become sealed and impermeable to gas exchange. In addition to asphyxiation of the embryo, sealing the egg can lead to malpositions by causing embryos to orient away from the sealed area (Byerly and Olsen 1931). Hatchability is greatly reduced in eggs with holes that penetrate the shell membranes. These eggs are likely to have been contaminated by pathogens introduced to the egg's interior, or may have received physical injury to the yolk, embryo, or blood vessels.

If a pipped egg has been damaged in some way but still contains a live chick, move it immediately from the nest to a hatcher. If it is likely that such an egg has become contaminated by soil, feces, or other material, the egg may be fumigated (see earlier precautions) to kill pathogens that could enter the yolk sac or ruptured blood vessels, or be aspirated by the chick. Although the fumigant may irritate the chick, the need to prevent infection outweighs these considerations. Studies with chickens have shown that chicks may be fumigated while still in the hatching stage (Taylor 1949). This procedure has been used for cranes at Patuxent. Alternatively, the chick may be given prophylactic antibiotic treatment while in the egg or after hatching (see Calle et al. 1989).

Floating or Malpositioned Air Cell

Occasionally, the air cell forms in locations other than the large end of the egg. In other cases, a definite air cell does not form, but bubbles form in the albumen and float loosely in the egg. Eggs with a floating air cell or bubbles are unlikely to hatch and may have bacterial or fungal infections that were introduced through holes.

Chicks hatching from eggs with stationary, but displaced, air cells can die at hatching because either the chick still orients to hatch from the large end or the chick orients to the air cell but is malpositioned

and cannot emerge from the egg. The air cell serves as a breathing space until the chick hatches, and if the chick does not pip into it, the chick must break directly out of the shell and may drown or suffocate. Immediate assistance may help such chicks (Jordan 1989), but they are often lost. Incubating these eggs in a vertical position (large end up) and hand-turning may result in successful hatching (C. M. Kuehler, Peregrine Fund, Volcano, Hawaii, personal communication).

Contaminated Eggs

Egg contamination by pathogens may occur in the oviduct before laying, in the nest, or in the incubator. Little can be done once pathogens enter an egg: the embryo dies or the egg begins to deteriorate. However, dipping the egg in a disinfectant solution or injecting antibiotics directly into the egg is sometimes effective in preventing infection (Kuehler and Loomis 1992). To avoid contamination of additional eggs, immediately remove eggs from the nest or incubator if they contain known dead embryos or show signs of infection (odor or discoloration of egg contents). Due to gases generated during decomposition, eggs can actually explode, contaminating the incubator and other eggs with debris.

Attempt to culture pathogens from obviously contaminated eggs and some or all eggs with dead embryos or eggs that are presumed infertile (i.e., show no development). If microbial infection is a problem, investigate the cause or origin of infection. Persistent egg contamination warrants obtaining cultures from incubators, egg handling equipment, and birds (cloacal swabs). To minimize the incidence of infection, wear sterile surgical gloves to handle eggs and scrupulously disinfect all equipment and surfaces that eggs contact. If cultures of dead embryos are negative for bacteria and fungi, more sophisticated testing for viruses, nutritional problems, and genetic problems may be needed.

Assisted Hatches

If an egg fails to pip when expected, or takes longer than 48 h to hatch after the initial pip, consider assisting the chick in emerging from the egg. If an egg has not pipped the inner membrane when expected and there are signs of weakening, a malposition should be suspected. **Malpositions** can be diagnosed by three methods: (1) candling may reveal the bill tip near the

air cell, (2) radiographs may indicate the position of the embryo (Ensley et al. 1994), and (3) opening the shell and moistening the membrane with sterile saline may reveal the bill tip.

If the chick's head is pointing away from the air cell, carefully peel away the shell without disturbing the inner membrane. When the bill is located, select a small area free of active blood vessels, make a small incision, and pull the bill through far enough to expose the nares and allow respiration. Fluids may need to be removed from the airway of the chick. Once the chick is breathing, allow a day or two for the blood vessels to dry and the yolk sac to retract. Keep humidity high, cover the missing shell loosely with plastic or tape, and moisten membranes to keep them from drying. The chick will likely need assistance to free itself when ready to hatch.

Base your decision to assist on the strength of the chick's vocalization and movements. A tape recording of an adult crane's brooding call (purring), cheeping of other chicks, or an imitation of these sounds by the aviculturist usually elicits loud cheeps (if the air cell has been entered) and struggling from the chick. Although chicks enter a quiescent period prior to hatching (Hamburger and Oppenheim 1967), a chick that remains quiet and motionless may be too weak to finish hatching on its own.

Failure to hatch after pipping may result from malposition, dehydration of the shell membranes (so the chick adheres to the membranes), an abnormally thick shell, or weakness of the chick due to improper incubation conditions. Assist the chick cautiously in case the yolk sac is still exposed (see below) or the external blood vessels are still functional. Remove the eggshell over the air cell to expose the chick without disturbing the membranes around the chick. Moisten the membrane with sterile water or saline to release the chick and reveal blood vessels. If the blood vessels appear empty (i.e., pale and small in diameter), gently and slowly peel the membrane away from the chick's bill a little at a time. Do this gradually to allow the chick to emerge by itself, if possible. Membrane blood vessels can also be tied off surgically. A chick that continues to be weak, shows edema around the back of the head and neck, and does not progress on its own should be carefully pulled from the egg. **Weak chicks** can be treated with fluids, glucose, and steroids by injecting either while still in the egg or immediately after assisted hatching. Additional information on time intervals between hatching events and assistance of chicks is provided by Hartman et al. (1987).

Exposed Yolk Sac

An exposed yolk sac (i.e., one that has not been retracted within the abdominal cavity) usually results from early hatching (caused by high incubation temperature, excessive behavioral stimulation, or an assisted hatch). Handle such chicks carefully because rupture of the yolk sac can increase susceptibility to infection and bleeding, and deprives the chick of a nutrition source immediately posthatching. See Chapter 5 for further discussion and information on the treatment of this condition.

Transporting Eggs

Fresh, unincubated eggs can be transported relatively easily as long as they are protected from physical damage (cracked shells, freezing, or overheating). For incubated eggs, a rapid transfer (less than 5 min) between nests and/or incubators at the same site requires no special equipment except that eggs should be cushioned to prevent damage from sudden or rough movement. The amount of cooling that occurs in the egg in a few minutes has no appreciable effect on the egg unless the weather is extremely cold or the egg is exposed to rain or chilling winds.

To transfer over substantial distances (e.g., from a wild nest to a captive-rearing facility), a portable **suitcase incubator** (Fig. 4.12) was developed at Patuxent (Erickson 1981). Hatchability of transported eggs is usually very high with these incubators, which are constructed by lining a reinforced cardboard suitcase with polyurethane foam. Supply heat using hot-water bottles filled with water at 51.7° C (125° F) and placed in the bottom of the suitcase. Above the hot-water bottles, place polyethylene foam inserts that will contain the eggs. Egg shaped cut-outs (pockets, one half of which is in the lid and half in the base) are interconnected with air channels running between egg pockets and extending to the ends of the inserts to allow convection and to equilibrate temperature throughout the incubator. When the incubator suitcase is filled with eggs, close the cover and insert a thermometer through a hole in the side or cover so that it extends into the area near the eggs (i.e., into one of the air channels between eggs). Monitor the temperature continuously, and regulate it within 34.4-37.2° C (94-99° F) by keeping the incubator lid closed to retain heat and by opening and fanning the

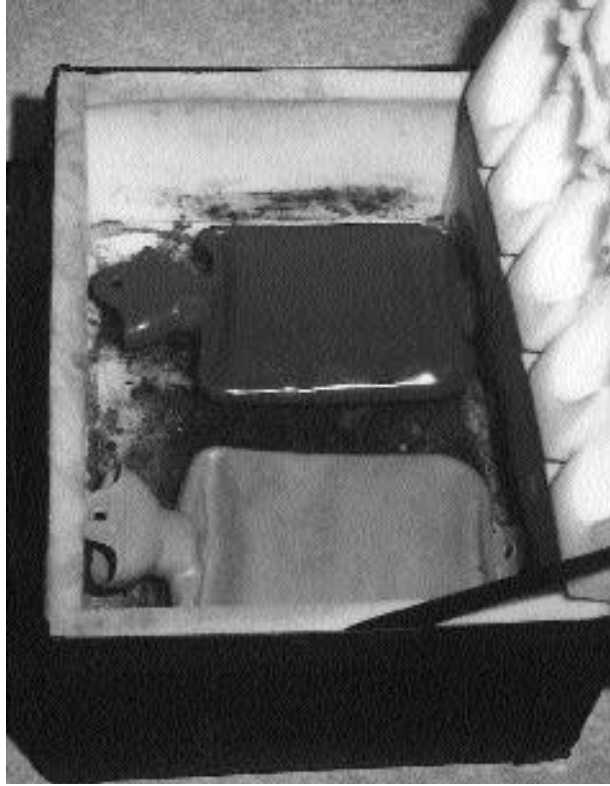


Fig 4.12. *Portable incubator suitcase: Left, hot water bottles now empty but in place; Right, polystyrene liner with cavities for eggs and channels for convection and thermometer.*

Photo Patuxent

lid to release heat if the temperature is too high. Do not allow eggs to enter or remain in an incubator hotter than 37.2°C (99°F). Replenish the hot-water bottles at approximately 2-h intervals or when the incubator temperature falls below 34.4°C (94°F). Hold the incubator suitcase in your lap or lift it above your lap to protect eggs, especially those in early stages of incubation, from sudden movements or stops, bumps, or vibrations, which would otherwise cause blood vessels or embryonic membranes to rupture.

For **international** shipments lasting 24 h or more, ICF has developed a **wooden box** with separate egg and water bottle compartments so eggs are not disturbed when changing water. Temperatures are kept between 36.1 and 37.2°C (97 and 99°F).

Closing Statement

Incubation is part art and part science. Understanding the principles of incubation alone will not guarantee success. The aviculturist must become experienced in recognizing subtleties revealed during candling

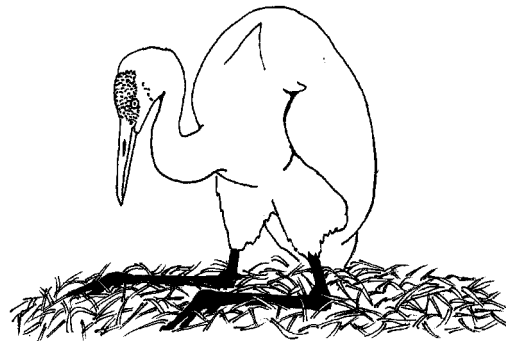
and general handling of eggs, and in dealing with problems with the incubators, whether they are machines or birds. In the beginning, experiment with less valuable eggs, and expect to lose a few of these, but learn from your losses and avoid them. The information presented here and in the references cited provide a basis for conducting an incubation program, but the novice should also seek the advice of experienced persons when problems arise.

Literature Cited

- Ar, A., and H. Rahn. 1978. Interdependence of gas conductance, incubation length, and weight of the avian egg. Pages 227-235 in J. Piiper, editor. *Respiratory function in birds, adult and embryonic*. Springer-Verlag, Berlin.
- Archibald, G. W., and S. R. Swengel. 1987. Comparative ecology and behavior of Eastern Sarus Cranes and Brolgas in Australia. Pages 107-116 in J. C. Lewis, editor. *Proceedings 1985 Crane Workshop*. Platte River Whooping Crane Habitat Maintenance Trust and U.S. Fish and Wildlife Service, Grand Island, Nebr.

- Benoff, F. H., and J. A. Renden. 1980. Broiler breeder egg shape. 2. Hatchability of pole distinguishable and pole indistinguishable eggs. *Poultry Science* 59:1682-1685.
- Breiby, T. E. 1994 unpubl. Incubation and hatching intervals of cranes with an analysis of the relationship of captive and wild laid Whooping Crane eggs. International Crane Foundation report.
- Brown, A. F. A. 1979. The incubation book. The World Pheasant Association, Reading, U.K. 246 pp.
- Burnham, W. 1983. Artificial incubation of falcon eggs. *Journal of Wildlife Management* 47:158-168.
- Byerly, T. C., and M. W. Olsen. 1931. The influence of gravity and air-hunger on hatchability. *Poultry Science* 10:281-287.
- Calle, P. P., D. L. Janssen, C. M. Kuehler, and J. Oosterhuis. 1989. Gentamicin injection of incubating avian eggs. Pages 83-89 in J. H. Olsen, editor. Proceedings of the American Association of Zoo Veterinarians.
- Carthew, W. R. 1966. Breeding of Grey-necked Crowned Cranes (*Balearica regulorum*). *Avicultural Magazine* 72:1-3.
- Conway, W., and A. Hamer. 1977. A 36-year laying record of a Wattled Crane at New York Zoological Park. *Auk* 94:786-787.
- Cramp, S., and K. E. L. Simmons, editors. 1980. Handbook of the birds of Europe, the Middle East and North Africa. The birds of the western Palearctic. Vol. 2, Hawks to bustards. Oxford University Press, Oxford, U.K. 687 pp.
- Derrickson, S. R., and J. W. Carpenter. 1982. Whooping Crane production at the Patuxent Wildlife Research Center, 1967-1981. Pages 190-198 in J. C. Lewis, editor. Proceedings of the 1981 Crane Workshop. National Audubon Society, Tavernier, Fla.
- Derrickson, S. R., and J. W. Carpenter. 1987. Behavioral management of captive cranes—factors influencing propagation and reintroduction. Pages 493-511 in G. W. Archibald and R. F. Pasquier, editors. Proceedings of the 1983 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Ensley, P. K., B. A. Rideout, and D. J. Sterner. 1994. Radiographic imaging to evaluate chick position in California Condor (*Gymnogyps californianus*) eggs. Pages 132-133 in R. E. Junge, editor. Proceedings of the American Association of Zoo Veterinarians.
- Erickson, R. C. 1981. Transport case for incubated eggs. *Wildlife Society Bulletin* 9:57-60.
- Erickson, R. C., and S. R. Derrickson. 1981. The Whooping Crane. Pages 104-118 in J. C. Lewis, editor. Crane research around the world. International Crane Foundation, Baraboo, Wis.
- Ernst, R. A. 1975. Hatchery and hatching-egg sanitation. University of California Division of Agricultural Sciences Leaflet 2629.
- Fisher, I. J., and S. E. Swengel. 1991. A guide for aging Sandhill Crane eggs. *Wildlife Society Bulletin* 19:494-497.
- Gee, G. F., J. S. Hatfield, and P. W. Howey. 1995. Remote monitoring of parental incubation conditions in the Greater Sandhill Crane. *Zoo Biology*. In press.
- Glutz von Blotzheim, U. N., K. M. Bauer, and E. Bezzel, editors. 1973. Handbuch der Vögel Mitteleuropas, Band 5. Akademische Verlagsgesellschaft, Wiesbaden, Germany.
- Hamburger, V., and R. Oppenheim. 1967. Prehatching motility and hatching behavior in the chick. *Journal of Experimental Zoology* 166:171-204.
- Hartman, L., S. Duncan, and G. Archibald. 1987. The hatching process in cranes with recommendations for assisting abnormal chicks. Pages 387-397 in J. C. Lewis, editor. Proceedings 1985 Crane Workshop. Platte River Whooping Crane Habitat Maintenance Trust and U.S. Fish and Wildlife Service, Grand Island, Nebr.
- Heck, W. R., and D. Konkel. 1983. Incubation and rearing. Pages 34-76 in J. D. Weaver and T. J. Cade, editors. Falcon propagation: a manual on captive breeding. The Peregrine Fund, Fort Collins, Colo.
- Hutt, F. B., and A. M. Pilkey. 1934. Studies in embryonic mortality in the fowl, V. Relationships between positions of the egg and frequencies of malpositions. *Poultry Science* 13:3-13.
- Johnsgard, P. A. 1983. Cranes of the world. Indiana University Press, Bloomington. 257 pp.
- Jordan, R. 1989. Parrot incubation procedures. Silvio Mattachione, Pickering, Ontario. 142 pp.
- Joyner, K. L., and U. Abbott. 1991. Egg necropsy techniques. Pages 148-152 in Proceedings of the Association of Avian Veterinarians, Chicago, Ill.
- Kuehler, C., and J. Good. 1990. Artificial incubation of bird eggs at the Zoological Society of San Diego. *International Zoo Yearbook* 29:118-136.
- Kuehler, C. M., and M. R. Loomis. 1992. Artificial incubation of non-domestic bird eggs. Pages 1138-1141 in R. W. Kirk, editor. Current veterinary therapy XI: small animal practice. W. B. Saunders, Philadelphia, Pa.
- Kuyt, E. 1987. Management and research of Whooping Cranes 1965-1982. Pages 365-369 in G. W. Archibald and R. F. Pasquier, editors. Proceedings of the 1983 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Landauer, W. 1967. The hatchability of chicken eggs as influenced by environment and heredity. Monograph 1 (Revised), Storrs Agricultural Experiment Station, University of Connecticut-Storrs.
- Langenberg, J. 1989. Pathological evaluation of the avian egg. Pages 78-82 in Proceedings of the American Association of Avian Veterinarians, Greensboro, N.C.
- Liao Yanfa. 1987. The Black-necked Cranes of Longbaotan. *ICF Bugle* 13(1):1, 4-5.
- Mahan, T. A. 1992. Incubation of crane eggs by Cochon hens. *Avicultural Magazine* 98(3):126-130.
- Putnam, M. S. 1982. Refined techniques of crane propagation at the International Crane Foundation. Pages 250-258 in J. C. Lewis, editor. Proceedings of the 1981 Crane Workshop. National Audubon Society, Tavernier, Fla.
- Putnam, M. S., and B. C. Wentworth. 1986. Reducing excessive weight loss in a Whooping Crane egg by rehydration. *Avicultural Magazine* 92:161-165.
- Rahn, H., and A. Ar. 1974. The avian egg: incubation time and water loss. *Condor* 76:147-152.
- Russman, S. R. 1987. The effects of artificially cooling crane eggs at the International Crane Foundation. Pages 535-537 in G. W. Archibald and R. F. Pasquier, editors. Proceedings

- of the 1983 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Sanctuary, W. C. 1925. On the cause of dead chicks in the shell. *Poultry Science* 4:141-143.
- Stehlik, J. 1970. Beitrage zur Biologie der Jungfernkraniche (*Anthropoides virgo*). *Freunde des Kölner Zoo* 3:115-119.
- Stoodley, J., and P. Stoodley. 1983. Parrot production. Bezels Publications, Portsmouth, U.K. 108 pp.
- Stromberg, J. 1975. A guide to better hatching. Stromberg Publishing, Pine River, Minn. 100 pp.
- Sullivan, K. 1994 unpubl. Achieving the greatest hatching rates among populations of captive cranes: an analysis of incubation techniques and their success. International Crane Foundation, Baraboo, Wis. 11 pp.
- Talmadge, D. W. 1977. The effect of incubating eggs narrow end up on malposition II and hatchability. *Poultry Science* 56:1046-1048.
- Taylor, L. W. 1949. Fertility and hatchability of chicken and turkey eggs. John Wiley and Sons, New York. 423 pp.
- Urban, E. K., D. H. Fry, and S. Keith. 1986. The birds of Africa. Vol. 2. Academic Press, Orlando, Fla. 552 pp.
- Van Ee, C. A. 1966. Notes on the breeding behaviour of the Blue Crane, *Tétrapteryx paradisea*. *Ostrich* 37:23-29.
- Walkinshaw, L. H. 1965. One hundred thirty-three Sandhill Crane nests. *Jack-Pine Warbler* 43:136-143.
- Walkinshaw, L. H. 1973. Cranes of the world. Winchester Press, New York. 370 pp.



Chick Rearing

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Rearing method has a profound and lifelong effect on a crane's behavior. Because cranes sometimes live longer than seventy years in captivity, it is imperative to rear according to need. Cranes reared for display should be reared differently than breeding stock or birds for release to the wild.

There are two primary crane chick rearing methods. The first, parent-rearing, is when a chick is reared by one or both of its own parents, or surrogates of the same or another species. These chicks are usually correctly imprinted and can be released into the wild or serve as natural breeders. The second, hand-rearing, is when the crane chick is reared by humans with or without the use of costumes and with or without live conspecific crane imprinting models (see Chapter 11D for details on raising hand-reared chicks for reintroduction).

The amount of preparation, time, labor, and expense incurred rearing chicks will vary greatly depending on the method chosen, and must be carefully considered to ensure success. Husbandry practices applicable to both rearing methods are discussed first, followed by the techniques specific to each method and veterinary techniques for chicks.

General Husbandry for Chick Rearing

Facilities

Parent-rearing facilities are modified breeding pens (see Chapter 12). Facilities that allow caretakers to lock adult cranes in or out of a shelter while capturing the chick reduce the risk of injury to birds and keepers. Young crane chicks are surprisingly mobile, and can easily pass through typical chain link fence. To prevent this, a smaller (0.56 cm or 0.25 in) plastic coated mesh, or solid material, should be added to the fence from

just below ground level to a height of 30 cm (12 in). Install the "chick proofing" on the inside surface of the chain link to prevent chicks from getting trapped between the two fencing materials.

Optimally, chicks should be reared in flight-netted pens. If this is not possible, the flight capabilities of chicks must be monitored closely around fledging (60-100 days depending upon species) and appropriate flight restraint measures must be taken before the chicks escape (see Chapter 11E).

Hand-rearing facilities should include brooder boxes (Fig. 12.1) or commercial incubators and predator proof indoor/outdoor covered pens large enough to provide chicks with adequate exercise (see Chapter 12). The indoor pen should provide a controlled environment with food and water. Each should be equipped with at least one heat lamp of adjustable height. If outdoor runs are large enough (ca 3 × 7 m) to allow growing chicks adequate space for exercise until they are fully grown, they are also large enough to house calm adult conspecifics in neighboring pens to serve as imprinting models. If a parent-reared chick requires intensive care, the runs can also serve as adequate housing for a dam and her chick. Of course, use caution when bringing sick animals into the rearing facility.

To promote strong imprinting on conspecifics, visual contact with an adult or subadult crane in a neighboring pen is beneficial during the first days after hatch. Older chicks can be housed adjacent to adults or subadult conspecifics, which we call socialization models, that are housed in large pens built adjacent to outside chick runs. These birds provide an opportunity for chicks to observe social behavior within a group, something the imprinting model does not provide.

At Patuxent, chicks young enough or sick enough to be kept in a brooder box are not usually exposed to live conspecifics because of the added stress associated with trying to watch and follow the adult. Instead, taxidermic heads are left in sight of the chick and pelt fragments of gray or white feathers are left for

the chick to cuddle. Because chicks readily respond to the heads and pelts, when the chick is strong enough to be returned to a rearing pen, recognition of the live adult is usually immediate.

If brooder boxes are used, they should have an exercise area of at least one square meter, be easily cleaned, and allow easy access to the chick. Because of aggression, chicks must often be housed in individual pens and brooder boxes. Ideally, partitions between brooder boxes, incubators, and indoor/outdoor pens or runs should be made of a material (e.g., plexiglass) which allows chicks visual contact with neighboring chicks and adult imprinting models while preventing injuries. Wire barriers or flexible, plastic mesh (0.45–0.56 cm, 0.2–0.25 in) also provide for visual contact with neighbors. Even with these barriers, there remains some risk of eye or beak injuries if the chicks fight through the fences.

Adult cranes housed next to chicks as imprinting models may be curious or may perceive young chicks as prey. Install an adequate barrier between chicks and adults to prevent adults from jabbing through a wire mesh barrier or digging under the plexiglass. Plexiglass must be carefully secured and buried at least 2 inches in the ground. If adult cranes are not housed next to chicks, partitions made of flexible, plastic mesh (0.45–0.56 cm, 0.2–0.25 in) can be used to separate chicks. However, as chicks get older and fight more aggressively, they may need plexiglass partitions to prevent permanent beak damage or eye injuries.

Indoor pens are easier to clean and maintain if they have concrete floors with substrate such as 5–6 cm (2 in) of wood chips, shavings, or sand. Wood shavings should be dust free, laboratory grade if possible (e.g., Beta-chips), to reduce respiratory problems. In addition, carpet pads or rubber matting may be placed over the concrete and under the wood shavings to help prevent chicks from slipping on the smooth surface. Using 2.5 cm (1 in) deep sand bedding diminishes the chances of slipping and decreases pathogens which might grow in wet shavings, but sand can fill the air with dust when sifting out feces. Sand and shavings can cause eye injuries or irritation when trapped under the lid. Placing a carpet on top of the sand or shavings is highly recommended for the first two weeks.

Natural turf is the best substrate for outdoor runs and provides a stimulating environment for the chicks, but is difficult to disinfect. To reduce parasite and pathogen loads, pens can be used on alternating years, lime can be tilled into the soil, or sod can be

removed and replaced. Another option in temperate climates is to leave the pen fallow for 4–5 months and maintain imprinting models in the same pen every year. Some institutions also treat the ground with One Stroke Environ (see Chapter 2 and Appendix).

Outdoor runs can also have concrete floors covered with a sand (2.54 cm, 1 in) deep. Sand is easily removed at the end of the season. The concrete slabs can be scrubbed and disinfected.

A hand-rearing facility may also include an exercise yard (where chicks can be walked or allowed to run freely and are socialized under supervision) and a swimming pool at least three feet deep for hydrotherapy (see Exercise, this chapter). Flight netting outside runs ensures that older chicks cannot fly out and protects them from avian and terrestrial predators.

Protocols and Record Keeping

Current protocols for specific rearing methods should be available in chick rearing facilities. Detailed records should be kept on each bird (see Chapter 10). Important milestones in the chick's life, such as when the chick begins eating and drinking on its own, must be carefully noted. Physical problems, medical treatments, weight gain, changes or supplements to the diet, amount and type of exercise, socialization with other chicks, exposure to imprinting models, and behavioral changes should all be recorded.

Diet

Crane chicks must be provided with a nutritionally balanced diet suitable to the needs of a rapidly growing animal with a high metabolism. Specially formulated crane chick (starter) diet should be fed from hatching (day 0) through fledging (day 70+) or until all primaries are completely grown (up to 4 or 5 months).

Serafin (1980, 1982) recommended a diet containing no more than 24% protein and 0.73% sulfur amino acids for slowing growth of hand-reared cranes and thereby reducing the risk of abnormal leg development. Higher protein levels, especially animal protein, increase the incidence of leg and wing problems.

Pelleted, commercially prepared food is a convenient, reliable alternative to mixing special diets. Different feed formulas are needed for growing chicks, non-breeding adults, and breeding adults (see Table 2.2). Local feed producers may be able to

TABLE 5.1

Vogelpark, Walsrode crane chick starter diet.

Ingredient	% Composition
"Insekt-futter" (Insect-feed, red) ¹	50
Beef Hearts (finely ground)	25
Quark (yogurt-like dairy product)	10
Mealworms (1/2 quick-boiled, 1/2 live)	10
Green feed (lettuce, other greens)	5
Yeast powder (fortified)	trace
"Osspulvit-pulver" (calcium supplement)	trace
Crickets (Family Gryllidae) (fresh-killed)	(5-8 per bowl)

Mix all ingredients for a moist, but not saturated, mixture. Water can be added if too dry. Approximately 50-100 g per bird are fed twice daily. Crickets are placed on top of each food bowl. Pelleted food is also always provided. As the chicks grow, less of the fresh mixture is provided and they eat more pellets. After 6-8 weeks, the young bird eats only pelleted food.

¹ Type I (rot) Trocken-Weichfutter (mixture for small birds and quail) from: Claus GmbH, Spezial-Futtermitt Postfach 100, 6703 Limburgerhof, Germany.

manufacture feed when provided the formula, or prepared crane feed may be purchased from Zeigler Feed Company (see Appendix).

Food must always be recently milled (within three months), dry, intact, and free of contaminants including mold and vermin. Crumbles are fed from hatching to 2-3 weeks of age. As the chick begins eating on its own, pelleted starter ration (diameter 5 mm or 0.1875 in, 24% protein) is mixed into the crumbles. The percentage of pellets is slowly increased until the chick is eating only pellets by three to four weeks. Parent-reared chicks can be fed a mixture of crumbles and pellets from day 1.

Many zoos feed a poultry (usually turkey) starter ration augmented with insects, fish, rodents, or other protein. At Vogelpark Walsrode (Walsrode, Germany), young cranes are fed a combination of the pelleted diet and a mix similar to a "soft bill" diet (Table 5.1).

Ideally, any institution raising crane chicks will have access to a complete, balanced diet. However, if this is not feasible, or if the diet available is questionable, a standard dose of water soluble poultry vitamins and electrolytes can be added to the water. The poultry additive should be discontinued as soon as a balanced diet is available.

After fledging (day 70+) or when primaries are fully grown, chicks are taken off starter ration and put on maintenance ration (protein 15-19%).

Supplementary Feeding

For very young chicks that are ill or otherwise slow to learn to eat, supplementary feeding may be necessary. Of the two methods available (i.e., force feeding pellets and gavage [intubation or tube feeding a liquid diet]), gavage is preferred. Instructions for supplemental feeding and tube feeding diets are found under Veterinary Techniques in this chapter. Tube feeding, unless done excessively, usually will not discourage a chick from eating on its own. In fact, for neonatal chicks, tube feeding small quantities 2-3 times a day may help stimulate their appetite while it also staves off dehydration.

Water

Fresh water should be kept constantly available and replaced daily or whenever contaminated. Non-spillable bowls must be deep enough to enable the chick to drink, but still allow it to escape should it stumble in (Fig. 5.1). Standard one gallon plastic poultry water jugs with red lids work well. Shallow bowls with a large, open surface area require more maintenance, because they are more easily contaminated by the chick's droppings.

Because cranes are wading birds, it seems reasonable that teaching the chick to drink would be a simple matter, however, it is not. Videotapes of wild Mississippi Sandhill Cranes show that the adults



Fig. 5.1. Chick feeder and waterer should be “non-tipable.”
Photo ICF

spend hours coaxing the chick to take its first sip, even while the chick sits in open marsh water.

Dehydration is a significant health concern when raising chicks. It reduces the desire to eat *and* drink, and may cause the chick to act dazed and lethargic. Chicks that have been eating well may stop entirely when dehydrated. Both hand-reared and parent-reared chicks must be carefully monitored in their first week for dehydration, and receive fluids when necessary. For clinical signs associated with dehydration and treatment, see Table 5.2.

Handling

Crane chicks are very fragile. Improper handling can cause lacerations, broken or damaged limbs, and ruptured yolk sacs, all of which can end in death. Some amount of handling is necessary in order to evaluate the health and growth of the chicks. Whenever deciding to handle chicks, consider the

amount of information gained versus the amount of stress to the cranes.

At hatching, crane chicks weigh between 100 and 130 grams and can fit in the palm of a hand. When picking up such a small, delicate animal, the “scoop” method is the safest (Fig. 5.2). Chicks can be “scooped” up from either the front or from behind. One or two fingers are slipped between the chick’s legs, and its body is held gently in the palm, while the legs dangle between the fingers or over the side of the hand. The other hand covers the chick’s back to prevent it from jumping off the palm. The legs are left unrestrained, but must be prevented from clawing the chick’s neck or face. When releasing chicks, support



Fig. 5.2. Scoop method of carrying newly hatched chick.
Thom Lewis pictured.

Photo David H. Ellis

TABLE 5.2

Clinical Signs and Treatment of Dehydration

Dehydration	Clinical Signs	Treatment
>5%	Not detectable	No treatment may be required
5-6%	Slight loss of skin elasticity. Some tenting of skin (over hocks or elsewhere). Dull appearing eyes. Tacky mucous membranes.	Subcutaneous fluids
7-9%	Some loss of skin elasticity with distinct tenting of skin possible, but not pronounced.	Subcutaneous fluids
10-12%	Mucous membranes dry. Chick dull and depressed. Extremities cool to the touch. Heart rate increased.	Intravenous bolus therapy and subcutaneous fluids; warmth, other supportive care (antibiotics, etc.)
12-15%	Chick extremely depressed and near death.	Intravenous fluids, warmth, antibiotics

the body until the legs support the bird's weight. Be especially careful to prevent the chick from falling onto its back. A supine chick will flail with its legs and can easily tear its own neck or injure its eyes with its nails.

A safe method of carrying birds over 10 days old is the bouquet method. One palm supports the bird's chest or keel while the legs are gently restrained by the other hand (Fig. 5.3). The legs are held apart with one or two fingers between them. The bird's body is held horizontally with the legs held back out of the way so the chick cannot claw itself. Legs must not be twisted, crossed over, or allowed to rub together. Two versions are available: either the bird is held horizontal or more upright.

As chicks grow older, care must be taken during handling so that emerging feathers are not damaged or broken. As wings and primaries grow, wings must be carefully restrained to reduce the risk of injury and feather damage.

Chicks over six weeks of age are normally carried like small adults. The body is held to the side like a football tucked under the arm, with the forearm holding the bird's body against the caretaker's side and the fingers of that same arm holding the legs (Fig. 5.4).



Fig. 5.3. Lorie Shaull demonstrates "bouquet" method of carrying a mid-sized chick.

Photo David H. Ellis



Fig. 5.4. Jane Nicolich holding an adult Sandhill Crane using the same "football carry" that is used on large chicks

Photo Glenn H. Olsen

Weighing

Monitoring a chick's growth (expressed as percent weight gain per day) is the primary factor in determining the chick's health. Weight gain can also be plotted and compared to a normal growth chart for the individual species (Fig. 5.5).

It is advisable to weigh a chick in a box on a standing scale (Fig. 5.6) rather than a hanging scale to reduce the risk of leg injuries. The scale should have an accuracy of 1% until chicks are over 2000 g. The floor of the box should be covered with a non-slippery material (i.e., carpet). To reduce human contact, chicks may be placed in a closed box during weighing.

Hand-reared chicks, when larger than 1 kg, can be guided or trained to walk onto a platform scale to reduce the chance of injury during weighing. However, many walk-on scales provide only 20 g increments.

Ideally, the first weighing should occur as soon as possible after hatching. Chicks should be weighed at the same time each day, until it is determined that the chick is no longer losing weight. The frequency of weighing the chick will depend on the chick's health and the rearing method used. Normal weekly weight gains for six species are summarized in Table 5.3.

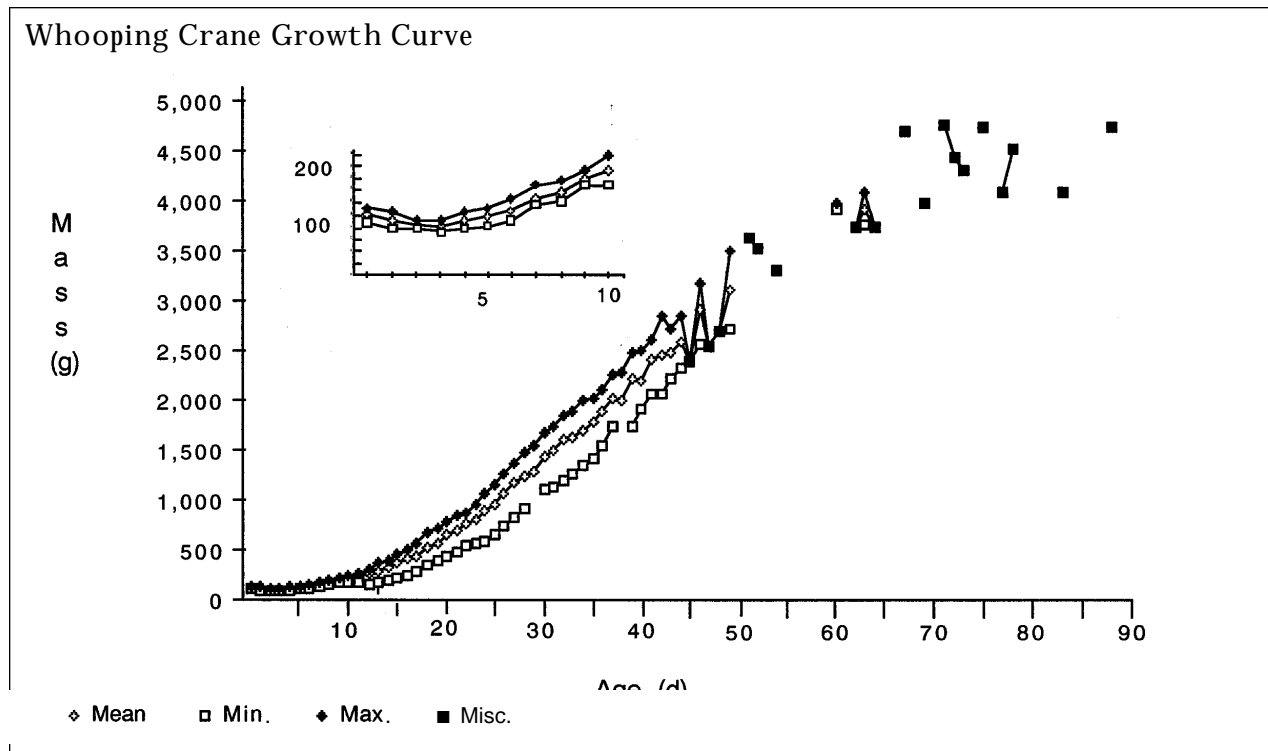


Fig. 5.5. Growth curve for Whooping Crane chicks. The brief weight decrease following hatching is normal as are sizeable fluctuations around the mean.



Fig. 5.6. Kathy O'Malley weighing a chick in a box.

Photo David H. Ellis

Growth Problems

A 10-15% weight loss in the first 3-5 days is normal as the chick absorbs its yolk sac (Fig. 5.5). Chicks that lose more than 15% of their body weight should be monitored closely and encouraged to eat (see Training Chicks to Eat and Drink under Hand-rearing in this chapter). If weight loss continues or lethargy sets in, support by subcutaneous injection of fluids or by gavage feedings (see Veterinary Techniques in this chapter or Chapter 8).

Excess weightgain (and resulting legrowing problems) is a major concern in rearing crane chicks (see Veterinary Techniques section). This problem, present in all rearing methods, is more common in hand-rearing. Chick weight should be monitored carefully during the period of most rapid growth, approximately days 10-40. Weight gain must be considered over the course of several days, but continuous weight gains exceeding 10% to 15% per day can cause problems. However, even chicks under ten days of age, or whose weight gains are less than 10%, occasionally suffer leg deformities. Therefore, daily monitoring is critical.

At Patuxent, leg problems seldom occur with the small, Mississippi Sandhill Crane, but are common

TABLE 5.3

Percent weight change¹ over one-week periods for six species of cranes.²

Week	Species ³					
	Siberian Crane	Sandhill Crane	Sarus Crane	Brolga Crane	White-naped Crane	Red-Crowned Crane
1	49	34	60	35	48	50
2	93	47	112	85	99	67
3	90	73	88	61	89	80
4	47	65	63	60	67	67
5	43	42	39	46	54	53
6	35	35	31	45	35	41
7	26	23	25	38	29	37
8	24	20	18	32	19	31
9	14	11	10	18	10	22
10	13	10	9	13	9	17
11	11	8	5	8	6	15
12	9	6	4	5	4	11
13	7	6	3	4	4	8
14	5	4	1	3	3	7
15	4	4			3	6
16	4	4			2	
17	3	3			1	
18	3	2			1	

¹ For example, during week 10, Siberian Cranes increase in weight by an average of 13%.

² Source: 1982-1988 data from ICF compiled by Ian Fisher.

³ Species and number of chicks weighed: Siberian Crane, $N = 15$; Sandhill Crane (Florida), $N = 46$; Sarus Crane (Eastern), $N = 19$; Brolga, $N = 7$; White-naped Crane, $N = 23$; and Red-crowned Crane, $N = 22$. Chicks were eliminated from this presentation if they developed leg rotations problems. Not all chicks were weighed every day.

with the larger Whooping Crane. Parent-reared chicks grow faster than hand-reared chicks, but in spite of their rapid growth, they rarely suffer from leg problems. Several factors probably contribute to this lack of leg deformities. First, parent-reared chicks typically have their diet supplemented by their parents with live food captured in the pens, and they are continuously fed over the course of the day. Second, parent-reared chicks are never on concrete, and, perhaps most importantly, the quality and quantity of exercise the chicks receive following their parents contribute to the low prevalence of leg problems.

To monitor for leg deformities, check each chick's legs daily. When the chick walks on a flat surface, the middle toes should be parallel, pointing straight for-

ward. When the chick stands, legs should be evenly spaced, perpendicular to the ground. If the chick's middle toes begin to point either outward or inward, the bird may be showing the first signs of a leg rotation. When a chick has bowed legs, another common deformity, its middle toes may still be parallel, but the legs are splayed either inward or outward at the hock.

To prevent leg problems, exercises should be encouraged (see Exercise under Hand-rearing Methods, this chapter). Chicks raised near active, live imprinting models may not need supplemental exercise if they spend a lot of time following the adult. If any bird shows signs of leg deviation or too rapid weight gain, respond with a combination of exercise, food rationing, and/or taping of legs (see Veterinary Techniques).

Food Rationing for Hand-reared Chicks.

Some birds gain excessively even with regular exercise. In such cases, the following food withholding techniques can be used to limit weight gain.

1. Remove food only at night. Usually chicks do not consume much food at night so removal limits only the amount of food available to them in the early morning hours when cranes normally feed.
2. Provide food four times a day for 15-60 minute intervals, then leave it in the pen overnight. This is the preferred method for most chicks, because the chick still has access to enough food to grow properly, to stem its hunger, and to prevent it from developing vices such as eating bedding or feces.
3. If the chick is eating pelleted food, provide either crumbles only or a mixture of crumbles and pellets so the chick has to expend more time and energy to eat the same amount of food.
4. Remove food at night and provide it three or four times a day for an hour at a time. On this regime, chicks may become frantic or consume bedding in which case using one of the other options must be implemented.

Regardless of which technique is used, food rationing ends as soon as the chick's weight gain slows for several days or abnormal behavior develops. Weight gain should be monitored daily, however, until the period of rapid growth is over (ca 40 days of age).

In a group-rearing situation, if even one chick is showing excessive weight gain, the entire pen should be rationed or the bird of concern can be temporarily removed to limit its feeding opportunities. However, carefully monitor the social interactions of the chicks because deprivation of food can result in increased aggression.

Parent-rearing Crane Chicks

The parent-rearing process involves a pair of cranes, or occasionally a single bird, raising their own or an adopted chick. The process closely parallels the rearing of a chick in the wild. From its parents, the chick learns to drink, forage, avoid humans and predators, and learns how to interact with other cranes. Cranes reared by their own species imprint properly and make good candidates for release into the wild. With some taming, they can also be good birds for captive breeding with the advantage of having reduced need for artificial insemination (AI). Without taming, they

are wary of humans and rarely attack caretakers during routine interactions, but unless tamed, they sometimes injure themselves or caretakers during handling or other disturbances.

Taming can best begin as soon as a chick is independent of its parents (ca 5 months of age), but can also be accomplished with older birds. When possible, pen the wild, parent-reared cranes with tamer, hand-reared conspecifics of similar age. Prior to taming, most of a parent-reared crane's interactions with people have been negative. In taming, gradually increase the amount of positive or neutral experiences with people. For example, provide treats such as corn, pinkies (baby mice), or smelt on a daily basis (see also Chapter 6). Toss treats to the birds, then move far enough away for them to approach and eat the treats. Purr, avoid sudden movements, and crouch down (i.e., decreasing height decreases threat). Through time, the birds' retreat distance gradually decreases and eventually some birdswill approach. Parent-reared cranes will usually remain somewhat aloof and will not approach closer than 3-5 m. However, some parent-reared breeding adults will attack to protect eggs or chicks.

Another taming technique is to merely linger in the open in a non-threatening way. Begin at the distance beyond which cranes no longer pace the far fence (ca 30 m or more), then move closer as the birds grow accustomed to your presence. After the cranes are tame toward their caretakers, they, with time, will accept other humans as well.

Parent-rearing is less labor intensive than conventional hand-rearing, but requires more extensive facilities to maintain breeding pairs and replacements. Parent-reared chicks are subject to more danger than chicks reared by hand (e.g., inclement weather, parasites, and greater risk of predation). Predator proofing the perimeter fence and flight netting the enclosure will help reduce the mortality due to terrestrial and aerial predators (see Chapter 11F).

Choosing Parents

In choosing pairs to raise chicks of endangered species, evaluate the previous parenting experience of the pair. All captive cranes do not make good parents; some kill or neglect chicks. Before a pair is allowed to raise a genetically valuable or endangered crane, we recommend that the pair be closely monitored and have at least one successful year in raising non-endangered cranes or even a chick of

some other large-bodied species of precocious fowl (e.g., Anseriformes or Galliformes). Preferred pairs tolerate routine disturbances (such as caretakers feeding and administering treatments). They do not redirect aggression to eggs or chicks, nor do they neglect chicks when disturbed. Good pairs are attentive to their chicks (feeding, brooding, defending, and sheltering them), and both members participate in incubation.

Cross-fostering, the rearing of a chick by parents of another species, results in near-normal behavior development, however, the chick may be sexually imprinted on the foster species and thus may experience difficulty pairing with its own species when sexually mature (see Chapter 6). In captivity, this imprinting may be altered or reversed by removing the chick from the foster parents before fledging, socializing it in a juvenile cohort of conspecifics, and then force pairing it at two years of age.

If a chick will be reared by foster parents, careful planning is required to have a suitable pair ready at the hatch date. Normally, eggs of two or more potential surrogate pairs are manipulated to ensure that a suitable pair is ready.

Adoption Methods

Five different adoption methods have proven successful. In the preferred method, a pair hatches an egg they have been incubating and raises the resulting chick. The egg can be their own or one from another pair. A second alternative is to introduce a piped egg in exchange for an egg that has been incubated at least 21, but preferably 25-30, days. (ICF has had pairs hatch eggs after as little as one week of incubation in the pen.) To decrease the chances of the pair rejecting or destroying the egg, place the egg with the pip-hole down. This method is used when a pair's incubation performance has been poor or is unknown.

In the other three fostering methods, small chicks are introduced to surrogate parents. Because of high risk to the chick, these methods should not be used routinely nor should they be used with chicks of an endangered species. The success of these techniques depends on the behavior of the chicks as well as the parents. Only chicks that have had previous exposure to live cranes or taxidermic brooder models and heads should be used. No matter how attentive the adults are, if the chick is afraid of or unresponsive to live cranes, the attempt will likely fail.

In one approach, if a piped egg is unavailable or if a pair has a history of problems during hatching, a young chick is introduced in place of an egg or dummy egg. At Patuxent, this has been successful in three out of six attempts with Sandhill Crane chicks and parents, and four of four attempts with Sandhill Crane chicks and Whooping Crane parents. Generally, only experienced parents will tolerate such abrupt changes. If this method is tried with an inexperienced pair, only expendable chicks should be involved.

In the fourth method, the pair's chick is replaced by another chick. The chicks being exchanged should be similar in age, weight, appearance, and activity level. This technique is often used when a chick becomes sick or dies. Replacement of a sick chick with a healthy one utilizes the parent rearing capabilities of a valuable pair while allowing more intensive care to be given to the sick chick.

In the final method, a chick is fostered to a pair without eggs or another chick. Patuxent has fostered Sandhill Crane chicks to Whooping Crane pairs that had never laid or had not recycled after an earlier clutch and were not sitting on dummy eggs. Four of twelve attempts were successful (i.e., chicks survived at least two weeks). Chick ages in successful adoptions ranged from 1-12 days. ICF attempted to introduce 2-hour- to 3-day-old Florida Sandhill Crane chicks to five pairs of Whooping Cranes. Some adults showed extreme interest and/or aggression toward the chicks, while others completely ignored them. Chicks that were initially reared by surrogate crane parents (versus hand- or isolation-rearing) showed the most normal interactive behaviors with the adults. ICF uses special pens (which are placed in the pair's enclosure prior to the breeding season to acclimate the pair to the new structure) to introduce the chick to the adoptive parents and assess their responses before actually releasing the chick in their pen.

Wing tags or leg bands should be removed from chicks before adoption is attempted because parents are likely to peck or pull at these objects and injure the chicks. If either parent behaves aggressively toward the chick, if both ignore the chick, or if the chick flees from the parents, the adoption should be terminated.

Egg adoptions are useful in stimulating parental behavior and to increase chick rearing in the colony (for details see Chapter 3). Patuxent attempted egg adoptions with nine pairs of non-productive birds. Seven did not adopt the eggs. Of the two pairs that accepted the eggs, one later adopted a chick

exchanged for the egg and one hatched the egg. Both females (ages 5 and 6) laid the following season for the first time. Patuxent has also attempted egg adoptions eight times with breeding pairs that either did not lay in that particular year or did not re-lay after an earlier clutch was removed. Five of these attempts were with the same Whooping Crane pair in consecutive years. This pair adopted the eggs and raised chicks in two years. In the other three years, they rejected the dummy eggs, but later accepted foster chicks. Another Whooping Crane pair which did not lay one year rejected an introduced dummy egg. Finally, a Mississippi Sandhill Crane pair which did not lay for two years, but had in previous years, was given dummy eggs. Each year they immediately adopted the dummy egg and subsequently hatched or adopted and reared a Sandhill Crane chick.

ICF has attempted six egg adoptions with Whooping Cranes, three with non-layers who did not exhibit signs of egg laying and three with post-molt layers. None were successful. Two of two egg adoption attempts with Siberian Cranes were successful. One was to a female who exhibited intense nest building and bill-down behavior, but did not lay an egg. Another was to a pair in which the male had prior incubation and chick rearing experience. He immediately accepted the egg, built a nest around it, and began incubating. Over the following 9 days, the female showed complete disinterest and never joined in incubation. On the ninth day, the egg was replaced with a pipped egg. The male successfully hatched, brooded, and fed the chick, but after several hours it was found dead (presumably killed by the female). The pair laid eggs and raised a Siberian Crane chick two years later.

To adopt an egg, first watch for **signs of laying**. To avoid disrupting natural reproduction, egg adoptions should not be attempted if a pair seems likely to lay. If towards the latter part of the expected laying season, birds show no clear signs of laying or show a decrease attention given to the nest, an egg adoption may be attempted.

Proceed by surreptitiously **placing a dummy egg** in a handmade nest in an area of the pen where the pair seems most likely to lay. If the pair initially ignores or attacks the egg, continue the adoption. It sometimes requires a week for the pair to accept the egg and begin incubating. After incubating for at least 21 days (longer if possible), exchange the dummy egg for a pipped egg. If all goes well, allow the pair to hatch or adopt and rear the chick.

Routine Care of Parent-reared Chicks

A special protocol for parent-reared chicks outlines the schedule and methods for daily care, routine examinations, health care, and weight monitoring. Diagnostic tests (such as fecal parasite screening) and prophylactic treatments should be scheduled if disease history of the flock warrants (see example, Fig. 10.3). Chicks should be examined and weighed once a week after the first week until 40-50 days old.

All chicks, regardless of treatment schedule, are visually inspected from a distance daily to detect abnormal behavior (e.g., gait problems, impaired respiration, lethargy, or wing abnormalities). Avoid handling birds in temperature extremes. Frequency of handling should be determined by the health of the chick, the need to monitor growth, the treatment schedule, and the tolerance of the family to stress.

Once the down of a newly hatched chick is dry, the chick is removed from the pen, weighed, checked for general physical condition (including examination and disinfection of the umbilicus), and given prophylactic treatments prescribed by a veterinarian. After treatment, the chick is placed back in the pen at the hatching location. When returning a chick to its pen, make sure the parents can see the chick and that the chick is never placed between caretakers and aggressive parents. Otherwise, the pair may accidentally step on the chick while rushing after the caretakers as they withdraw from the pen.

The flight capabilities of chicks housed in unnetted pens must be monitored closely after about 55 days. A juvenile that flies into a neighboring pen could be killed by its occupants; one that flies from the facility could become exposed to predators. Appropriate flight restraint methods are discussed in Chapter 11E.

Natural foods (e.g., insects) provided by the parents are supplemented with commercially prepared crumbles or pellets. Fresh food and water is placed near the nest until the chick is mobile (at 2-3 days). If possible, make food and water containers accessible to both the parents and the chick. This will reduce the number of containers needed and allow the parents to teach the chick where to locate food and water. After the first few days, the food and water bowls are placed near the adults' feeder (Fig. 5.7). If separate chick feeders and waterers are used, they can be removed when the chick is large enough to use those of the adults (i.e., at ca 30-40 days of age). Some parents redirect aggression by knocking over waterers or food bowls. If this is a consistent problem, secure the vessels in place.



Fig. 5.7. After about 10 days, the chick's food and water are moved near its parents' feeder.

Photo Jane M. Nicolich

In the wild, juvenile cranes leave their parents on spring migration or at the onset of the next breeding season. If a captive pair is intended to breed again, it is best to remove the juvenile from their pen at least three months before the planned egg-laying date. Upon separation from the foster parents, juveniles are normally penned with same-aged conspecifics to form social groups.

Working with Parents

Working around parent cranes requires care and training. Previously shy pairs become aggressive and aggressive pairs become very dangerous when they have a chick to defend. A crew of three caretakers is often required to tend a chick: two caretakers fend off the adults while the third person provides fresh food and water and, when necessary, captures the chick. When the chick becomes larger and faster, a fourth person sometimes participates in the capture. Use extreme caution to avoid stepping on a hidden chick. While servicing the pen, brooms and flexible plastic shields (made from toy sleds) are useful in fending off aggressive parents. Hold the broom so that the brush end parts in the middle and is held at the base of the crane's neck. This keeps the crane at a distance, minimizes the crane's ability to rush left or right around the broom, and also reduces the chance of injury to the crane. Be constantly prepared to grab the crane because some birds leap over or slip around the broom.

Hand-rearing Methods

Hand-rearing cranes has some advantages over parent-rearing: many chicks can be reared without the need for a large colony of surrogate parents; the chick's health and growth can be more easily monitored; the environment, including temperature and sanitation, can be controlled; and chick mortality can be considerably reduced.

Disadvantages to hand-rearing include the cost of building the facility, complete with pens large enough for adequate exercise, room for adults to encourage proper imprinting, pools, offices, equipment storage areas, laundry rooms, etc. Hand-rearing is also laborious, results in more leg and toe problems than parent-rearing, and is more likely to result in imprinting problems.

General Requirements for Hand-Rearing

The Newly Hatched Chick

A crane chick being hand-reared should be moved from the hatcher to its properly heated pen or brooder box once it has dried (4-24 hours after hatching). The hatchling should be weighed and examined. The umbilicus should be viewed and swabbed or sprayed with betadine (a povidone iodine solution). Some institutions administer prophylactic antibiotics during the first few days of the chick's life (see Veterinary Techniques this chapter). All pertinent information, including identification numbers, parental information, hatching history, and medical information is recorded on the chick's individual record (Fig. 10.3).

Temperature

For the first week, hand-reared chicks should be maintained in ambient temperatures between 35-37° C (95-98° F). Monitor not only the temperature, but also the chick's behavior. Cold chicks shiver and call; overheated chicks pant and/or hold their wings away from their body. Temperatures can be decreased by 3° C (5° F) each week for healthy chicks, but should not drop below 21.5° C (70° F) until the chicks are at least

three weeks old. Chicks can have access to cooler areas throughout the day, but should be coaxed into or returned to the warmer area when chilled and for the night.

Indoor/outdoor pens with one or more heat lamps allow chicks to walk away from the heat source and thus self-regulate body temperature. Placing a taxidermic crane brooder model or food and water bowls near the heat source will encourage the chick to return to the heat source. Once accustomed to returning to the heat lamp for warmth, chicks will investigate their pen, including outdoor runs, and still return to the warmth of the heat lamp to sleep just as they would brood under a parent. Heat lamps can be removed once the chick is thermocompetent (i.e., at approximately 40-50 days).

Chicks are locked in the indoor pen (controlled environment) at night, especially when weather is cold or wet. After a chick exceeds 1250 g, it can be allowed out all night if weather conditions permit and if pens are predator proof. Adjust these guidelines according to the chick's health and weather conditions.

Substrate

Because hand-reared chicks often collect debris in their eyes during the first 7-10 days, they are not usually kept on sand or wood shavings during this period. However, smooth flooring can also cause problems (e.g., splayed legs, hock rotation, joint damage, or slipped tendons). Be aware that these leg problems may also be due to genetic flaws or incubation problems. To improve footing, use outdoor carpeting without backing or foam padding. Choose carpet that dries quickly, does not unravel or fray, and does not have loops that can catch small, sharp toenails.

During cleaning, replace soiled or wet carpet pieces with clean ones, or replace wet bedding with dry. Allowing carpets to dry in sunshine helps destroy bacteria and fungi. To sift feces and spilled food from bedding, use cat litter scoops or scoops constructed of wire mesh.

Training Crane Chicks to Eat and Drink

In the wild, parent cranes teach their young what to eat by offering food in their bill tips. When hand-rearing a chick, similar methods must be used. Tests on color and shape preference have shown that most crane chicks respond best to long, thin, red shapes (Kepler 1978). Red plastic spoons, red-tipped dowels, or red tape attached to the bill of a puppet,

taxidermic head, or feeding syringe can all be used in training crane chicks to eat and drink. Not all chicks respond to red. Mississippi Sandhill Cranes and African Crowned Cranes respond better to a black bill tip. It is important to accommodate the individual needs of each chick.

Chicks are introduced to food within a day of hatching. Many chicks are exhausted after hatching and spend most of the first day resting. Offer food to chicks when they are alert and active.

When offering food to chicks, the caretaker either imitates a crane's "purr" or plays a tape recording of a parent brood call. The caretaker then offers food in a feeding spoon in the puppet's bill (Fig. 5.8) or dips the tip of the feeding utensil in water, then dips the wet tip in dry crumbles, and offers the adhering food to the chick. Even newly-hatched chicks will usually stab at the food. If they successfully "hit" it, they will get some crumbles in their beaks and will swallow them. Feed the chicks until they lose interest, which may be in as few as five minutes for newly hatched chicks.

As the chick grows more coordinated and has better eyesight, move the feeding utensil closer to the food bowl. Within 2-3 days, the chick will begin to peck at the food where the utensil dips into the bowl. In several days, the chick will eat the crumbles from the bowl anytime the feeding utensil is moved around in the food. The puppet, taxidermic head, or dowel can be suspended on a string passing through an eyelet in the ceiling and tied to the pen wall, so the handler can purr and "bob" the puppet or dowel by flexing the string without entering the pen. Chicks should be offered food five or six times a day until they are gaining weight and routinely eating on their own (usually 4-14 days). At this time, the chick no longer needs training.



Fig. 5.8. Feeding chick with puppet head.

Photo David H. Thompson

Most newly hatched chicks will readily eat crumbled food, however, chicks that have been parent-reared often much prefer live food and may completely reject crumbled feed. They may also fail to respond to the color red. Sick chicks may also reject crumbled feed.

Several techniques can be used on chicks reluctant to accept crumbles:

1. Add mealworms, waxworms, or other enticing insects to crumbles. For chicks that have been parent-reared, this may be the only way to get them to eat. If chicks have not been parent-reared, mealworms or other live food should be used with caution because the introduction of live food may further decrease the chick's willingness to eat crumbles. A diet of live insects is nutritionally incomplete, and provides excessive levels of methionine and cystine which have been correlated with bone growth problems (Serafin 1982). To avoid this problem, crumbled feed should be incorporated as soon as possible.

When introducing live insect food, place moving insects on top of the crumbles so the chick sees them. Mealworms will quickly burrow to the bottom of the bowl. Chicks accustomed to live food will start digging through crumbles within 2-3 days. For chicks unaccustomed to live food, it may be necessary to offer insects using a puppet or to place mealworms in their mouths.

2. Offer moistened crumbles. This has several benefits. Moistened food stays on the feeding utensil and increases the chances of the chick getting a "good bite." Moisten food just prior to feeding and discard it after the feeding session to avoid proliferation of bacteria and mold. Moistened food also provides the chick with some fluids during the initial training period.

3. Offer liquid food in a red-tipped feeding syringe. A drop of liquid food, suspended from the end of the syringe, is consumed by the chick as it pecks at the tip. Eventually, the moistened syringe tip can be dipped in crumbles and offered to the chick.

4. Dip the chick's beak in crumbles to accustom him to the food bowl.

5. Place crumbles in mouth to accustom chick to texture.

Although methods 4 and 5 above have been used successfully, chicks are occasionally so disturbed from this handling that they become frightened of the keeper and/or bowl. Thereafter feeding sessions are even more time consuming. We recommend using techniques 1 to 3 to limit handling.

Sometimes it is best to tube feed reluctant chicks, especially if dehydration is a concern. Tube feeding provides nutrition as well as fluids and often stimulates the chick's appetite thus promoting self feeding (see Veterinary Techniques in this chapter for formulas and methods).

The following are **techniques to encourage drinking**. Patience is required because it often it takes a combination of techniques over several days before the chick is observed drinking on its own.

1. Use the mounted head, puppet or dowel, to lure the chick to the water. Stir the water, allow water to drip from the tip, or move the tip under water to stimulate the chick to pursue it.

2. Attract the chick's attention to the water bowl by placing marbles or other shiny objects such as marble-sized stones in the water. Vogelpark Walsrode (Walsrode, Germany) uses live insects for this purpose. Once the chick is drinking on its own, remove any inedible objects.

3. A red-tipped syringe or a gavage tube can be filled with water and held with a drop suspended from the tip. As the chick grabs the red tip, the drop will fall into its mouth. To avoid aspiration, water should never be squirted from the syringe into the chick's mouth.

4. Water can be dripped from a height of several feet so it splashes into the chick's water bowl at regular intervals. Birds are naturally attracted to moving water, and many chicks will investigate.

5. If there is no evidence of drinking, the handler should lift the chick above the water at an angle so the chick's bill dips into the water once or twice. Repeat the process, then set the chick down. Alternately, the bill may be gently dipped into the water, but this must be done cautiously to prevent the chick from aspirating water, and to avoid making it fear water. This technique is effective for both hand-reared and parent-reared chicks.

6. Provide the chick with a small pool. They will sometimes drink after wading into the water.

Dehydration can be detected by noting the signs listed in Table 5.2. Determine elasticity by gently pinching the bare skin of the leg just above the hock. If significant dehydration is evident, administer fluids subcutaneously. Injecting fluids sometimes leads a chick to drink on its own. Once the chick's activity level increases, it becomes easier to teach the chick to find and use water.

Exercise

Regular exercise is necessary for normal development and growth of strong, straight legs. Unfortunately, an exercise program does not seem to prevent toe problems. Wild and parent-reared crane chicks are on the move much of the day. Human caretakers can hardly produce an equivalent amount of exercise for hand-reared chicks, but several techniques can partially substitute.

Healthy chicks are fairly active by one or two days of age. Depending upon facilities and the species of crane, the chicks may get enough exercise if given a large pen (ca 400 m²) with lots of stimuli (e.g., insects, pools, plants, and toys) and a live adult crane next door. If pens lack stimuli and are <20 m², conduct 20 min exercise periods at least twice a day.

Chicks can be **taken for walks** from 10 minutes to all day depending on the age of the chick and on manpower availability. Avoid exercising chicks on smooth, slippery surfaces such as concrete or blacktop. Walking in natural areas provides good footing and exposes the chick to new experiences and new foods. At ICF, chicks are exercised and socialized under supervision in cohorts of two to four beginning a few days after hatching. As chicks get older and their aggressiveness decreases, more chicks can be exercised together. Terminate walking before a chick becomes exhausted, pants, grows frantic, or becomes overheated. Excessive exercise can cause the same leg problems as lack of exercise.

Disadvantages of walking are that it is labor intensive and may encourage excessive attachment to caretakers. For birds with leg deviation problems, walking can adversely affect the legs. Chicks that are walked too long can suffer joint injuries. The veterinarian can advise if walking or swimming (aqua therapy, discussed next) will better correct leg problems.

Vogelpark Walsrode has experienced few leg abnormalities in chicks reared in small pens lined with corrugated cardboard and covered with 13-18 cm (6-8 in) of woodwool (excelsior). The chicks receive little exercise other than walking through this thick carpet of woodwool. The diet used at Vogelpark (Table 5.1) may also contribute to their success.

Swimming (Fig. 5.9) is a useful method of exercise for all chicks and is especially important for birds with certain leg problems (e.g., rotated or bowed hocks, and traumatic injury of leg joints). To more efficiently use caretaker time, swim two to several compatible



Fig. 5.9. *Whooping Crane chick swimming for exercise.*

Photo David H. Ellis

chicks at the same time. Chicks should not swim in cool weather.

Disadvantages of swimming include the expense of purchasing and maintaining the pool, and the intolerance of some chicks to the stress of forced swimming. Many chicks protest swimming, and some may injure themselves while clambering to escape from the pool. By contrast, others become so accustomed to the technique that they are content to gently float on the water and fail to exercise. Reluctant swimmers may be encouraged to be more active by providing insects scattered on top of the water for them to catch, and caretakers purring to encourage chicks to follow or gently nudging them. Sometimes swimming chicks in groups of 2-4 will keep them moving. If several chicks are swum at once, control aggression by keeping them separated with brooms, long-handled brushes, or your hands.

A caretaker should always be present to observe the chicks in the pool. Young chicks are often not buoyant enough to swim for more than a few minutes and will sink if not rescued. Swimming sessions should range between 5-20 minutes and be terminated before chicks become chilled or sink. Chicks with leg problems can be swum either more frequently, or for longer periods (upto 30 minutes), depending upon their behavior. When chicks are removed from the pool, place them indoors near heat lamps unless it is above 27^o C (80^o F) and sunny outside. A chick may be unsteady after swimming, so use care when placing it back in the pen.

Whether chicks are walked, swum, or put into a program that combines both techniques, they must be introduced to exercise slowly. Swimming is less stressful to most chicks and requires less time than walking for similar benefits.

Chicks that are sick may need to have their exercise regime modified or restricted until they are fully recovered. Sick chicks routinely develop leg problems if exercise is not provided.

Imprinting and Socialization

Penning hand-reared chicks in close visual and acoustical contact with a live, conspecific crane (i.e., imprinting model) helps reduce imprinting on humans. Observing interactions of a group of adults (i.e., socialization models) may also facilitate the development of normal behavior. This is critical for hand-reared chicks that are to be used as captive breeders or release birds. Socialization is also encouraged by penning small groups of fledged chicks together.

Provide adults or subadults in pens within sight of the chicks' outdoor runs, or place individual conspecific models in adjacent pens. These model cranes should be selected based on their behavior including their ability to adapt to the chick facilities. Cranes which are nervous or call constantly should not be used. Loud calling can terrify the chicks, and may incite other models to call, creating a stressful environment.

Frequently, hand-reared subadults make the best models because they are young and curious, have not yet become aggressive to humans, adapt more readily to the pen (especially if raised there), and may actually interact with the chick by purring and tapping on the plexiglass. The model's interest in the chick may be curiosity or aggression. It should be assumed that a model will kill a chick if given the opportunity. Older hand-reared birds may be difficult, even dangerous, to use as models because of their aggression toward caretakers.

Different species show different propensities to serve as models. At Patuxent, male, female, subadult, and mature Whooping Cranes have consistently proved to be good imprinting models. They have shown interest in chicks, vocalizing to them, interacting with them continually, and protesting when caretakers handled the chicks. It is not unusual to see Whooping Crane models feeding chicks through the fence. Sandhill Cranes, on the other hand, have consistently been uninterested in the chick's welfare and have shown aggression toward, or predatory interest in, the chicks. At ICF, some Whooping, Wattled, and Siberian Cranes are interested in the chicks and some are not.

Chicks may also be socialized with chicks of their own or another species. Socialization can be combined with exercise by taking two or three chicks on walks together. Younger chicks are often aggressive and must be closely supervised to prevent fighting. As chicks grow, aggression decreases so cohorts of pre-fledged birds can be formed, but until fledging age, chicks are housed singly at night. Closely monitor any newly formed cohort to prevent injuries. In a new cohort, a dominance hierarchy will be established and aggression or dominance at feeding stations frequently occurs. Provide 2-3 feeding stations to reduce chances of injuries while ensuring adequate nutrition for all cranes. Caution is especially important when combining parent-reared and hand-reared crane chicks in one cohort.

If no other chicks or conspecific adults are available for imprinting models, mirrors can be added to the chick's pen. Taxidermic cranes (brooder models) and taxidermic heads (feeding models) can help in imprinting very young or sick chicks. These can be left in sight of the chick and pelt fragments of crane feathers in the appropriate colors can be left with the chick for "cuddling." Chicks may respond strongly enough to the heads and pelts that, when placed in a pen near a live conspecific, acceptance of it is immediate. For more information on imprinting see Chapter 6.

Types of Hand-rearing

Conventional Hand-rearing

These chicks are raised by humans without imprinting models (alive or taxidermic). Talking is not excluded from the rearing area, and chicks are housed singly or in small groups. These birds can be used for captive exhibition, breeding, and for non-behavioral research, but may not be suitable for behavioral or reproductive studies.

At one time, this was the most common hand-rearing method. Many institutions have modified conventional hand-rearing to reduce labor, aggression to caretakers, and the risk of these cranes becoming sexually imprinted on people.

Group-rearing. Chicks of some species can be successfully housed and reared together from hatching. At Patuxent, Florida Sandhill Cranes have been

successfully reared in groups of up to four, and Mississippi Sandhill Cranes have been reared as pairs and kept together in groups of up to four for as long as two weeks. Greater Sandhill Cranes and Whooping Cranes are too aggressive to be put together, even briefly, when very young.

Group-rearing is more likely to be successful when lighting is reduced in the indoor pen and in the spring when it is cooler. When attempting group-rearing, all chicks should be of similar age (i.e., within one day), and be placed together at the same time. Because chicks spend most of their first 48 hours sleeping, this is a safe period for shared housing.

Chick aggression often seems related to hunger, so chicks housed together should receive more frequent feedings, and have more than one feeding station available. Introducing live food (insects) can also reduce aggression. Aggression usually diminishes naturally around fledging time. However, chick aggression is unpredictable, and chicks raised together may be amiable for weeks or months, but suddenly commence fighting and seriously injure or even kill each other. Move the most aggressive chicks to separate pens.

Some species of cranes may be group-reared by including turkey or chicken poults to receive much of the aggression. Patuxent has used broad-breasted bronze (not white) turkeys and Cochin chickens because these breeds are placid birds. These aggression targets help crane chicks learn to feed, and they appear to stimulate the crane chicks to chase, move around, and get more exercise.

Crane chicks should not be allowed to kill poults. At least one poult should be used for every two crane chicks, though a 1:1 ratio may be better. Although this method reduces the risk of cranes injuring each other, some crane chicks are occasionally injured by other crane chicks and poults. Here again, the most aggressive chicks will still need to be moved to separate pens.

Using poults increases caretaking needs because pens are fouled more quickly with the additional birds. Poults should be tested and determined to be clean of any disease or parasite which could be transmitted to the crane chicks. Patuxent has used this method only when it was necessary to group-rear chicks for research, and has discontinued using poults with endangered chicks in favor of housing the chicks singly. Although this method has been used successfully, it is not highly recommended by the authors.

Conventional Hand-rearing with Imprinting Cues

Precautions are taken to reduce the risk of the chicks sexually imprinting upon people. These birds are hand-reared by uncostumed caretakers, but with exposure to various imprinting cues (e.g., puppet heads, taxidermic heads, brooder models, live conspecific imprinting models, and tape recordings of crane calls; see Imprinting in Chapter 6 and Fig. 11D.3). Talking is not excluded from the rearing area but is generally discouraged when interacting with the chicks. When interacting with chicks, caretakers either play recordings of crane vocalizations or imitate the appropriate crane call.

The newly hatched chick has access to a taxidermic conspecific crane mounted in a brooding posture (brooder model) with its carapels extended and its neck arched downward so its beak almost touches the ground (Fig. 5.10). Sometimes the model's beak is placed in water or food to encourage the chick to drink or eat. A small, portable tape recorder placed near the model provides prerecorded brood calls during feeding sessions. Alternately, the caretaker feeding the chick can imitate the crane brood purr and better coordinate the vocalizations with food presentation. However, playing the recordings during feeding sessions may help the chick differentiate between the imprinting cues (i.e., brooder model and puppet head) and humans. Vocal cues should be used selectively. Observations of cranes rearing cranes indicate that parents tend to decrease the frequency of vocalizations after two weeks of age (Hartup and Horwich 1994). After the chicks have learned where



Fig. 5.10. *Two Sandhill Crane chicks with brooder model and head used to promote imprinting.*

Photo Kathleen O'Malley

to locate food and water, vocalizations should be used only to attract the attention of the chick.

The brooder model can be left in the pen until the chick loses interest in it. Many chicks at Patuxent cuddle the model and sleep beside it even after thirty days of age. Models should be removed from any chick that tries to tear it up or from chicks that refuse to leave it for exercise.

Chicks are housed separately in visual contact with one another. They may be exercised and socialized in groups under supervision. At ICF, we have observed that if more than four chicks, preferably of the same species, are raised together, their interest in the human caretaker is reduced.

Around 10-12 weeks of age, the chicks are grouped (2-8 birds depending on pen size and other needs) and moved to pens next to adults of the same species. Chicks are allowed to see these socialization models until the following breeding season when the adults are sometimes screened off for to promote breeding (see Chapter 6).

Rearing in Isolation from Human Contact

For brevity termed **isolation-rearing**, this method involves hand-rearing crane chicks while minimizing the visual and auditory interaction with humans. Use of this technique produces birds suitable for captive breeding and release.

Three variations of isolation-rearing are screen-rearing, costume-rearing, and strict isolation-rearing. From early experiments in strict isolation-rearing, the other two methods evolved. Each method differs in the props (i.e., equipment, costumes, and adult cranes) required, and the amount of human contact with the chicks.

Screen-rearing (Fig. 5.11) describes the situation where chicks are fed by an uncostumed caretaker concealed by a portable screen. Talking may or may not be eliminated in the facility, but the caretaker remains silent when weighing or medicating the chicks. Visual contact with imprinting models is maximized. The chicks are imprinted on cranes, but remain tolerant of humans. Adults reared by this method are good display animals that breed readily in spite of considerable human contact.

In **costume-rearing**, the chick is reared with all of the imprinting and socialization techniques, models, and equipment discussed earlier, but with uncostumed humans visible only during stressful activities. For all positive interactions, humans wear

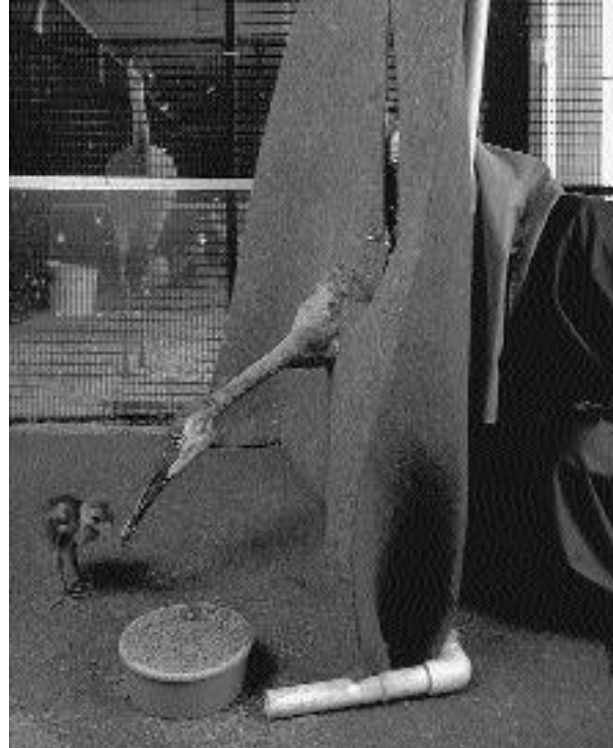


Fig. 5.11. *Screen-rearing. Note imprinting model (adult Sandhill Crane) in adjacent pen.*

Photo Vickie Lewis

a loose fitting hood and mantle that conceals the human form (Fig. 5.12; see also Chapter 11D). Birds so reared are suitable for captive breeding or release. If a strong bond between the costume and chick is not required, the routine medical management can be done by costumed personnel, otherwise all negative experiences are given by uncostumed humans. If the costume will be used at the release site, all capture episodes are by uncostumed humans except those



Fig. 5.12. *Costume-rearing Sandhill Crane chicks.*

Photo David H. Ellis

occurring when the chicks are only 1-2 weeks of age. Disadvantages to costume-rearing are that it is labor intensive, and birds may need to be acclimated to humans if they are to remain in captivity.

Facilities used for costume-rearing should prevent chicks from seeing uncostumed humans entering or leaving the facility, and should limit motor vehicle traffic noises and distant human voices. Solid fencing, tennis netting, or vegetation can be used to isolate a facility from its surroundings. Inside the facility, solid opaque walls, tennis netting, and portable screens can be used to restrict the chick's ability to see caretakers. A one-way viewing window and puppet hole can be installed in the main door (Fig. 5.13). Feeding can be accomplished with the hand puppet from outside the pen or the costumed caretaker can enter the pen and interact with the chick.

The costumes are specially made or can be a modified Hindu sari. They should be constructed of opaque, breathable material, loose fitting, and cover the caretaker from head to knee. The purpose is simply to disguise the human figure. The head covering or hood should be made of the same material as the costume body, with a face made of camouflage screen fabric to hide facial features. Some costumes are made to look more crane-like by sewing a scattering of feathers to a separate piece of material which can be attached to the wing sleeves with velcro or snaps, allowing removal when the costume is laundered.

Costume-rearing begins two days before an egg hatches at which time human voices are no longer



Fig. 5.13. Caretaker uses a one way window to observe and feed the chick while remaining out of sight.

Photo K. R. Langford

permitted in the incubator room. Tape recorded crane brood calls are played during routine checks. (Patuxent protocol for release birds calls for 15 min tape bouts, four times per day until hatching; ICF bouts vary in length from 30 sec to 2 min). Excessive use of the tape may stimulate chicks to hatch too quickly and suffer an exteriorized yolk sac or other problems. Once the egg pips and is moved to the hatcher, caretakers checking the egg wear the costume so the emerging chick will not see an uncostumed human. A chick needing assistance hatching has its head covered if it has emerged or is assisted by costumed caretakers. The welfare of the chick is paramount, and the ability to assist the chick has priority over concern for it seeing people. After the chick has hatched and dried, it is removed from the hatcher by a costumed caretaker and transported to the chick-rearing facility in a closed box (see Chapter 11D for details on rearing cranes for release).

Strict isolation-rearing was the term originally (and appropriately) used to describe the method of rearing cranes with minimal contact with both humans and most other living stimuli. Chicks were housed and reared in visual (but no physical) contact with other chicks. Live imprinting models were not used. A puppet head fed the chick through a hole in the pen door. Chicks were captured by a person covered by a sheet and placed in a box while pens were serviced.

Around fledging time the chicks were introduced to humans in an abrupt manner. People entered the pen to capture the chicks to do physical exams and to move them to larger pens amidst the flock.

The results of isolation rearing varied greatly by species (Putnam 1981). Some Sandhill Cranes were first hesitant upon seeing people, then willingly followed people and were quite curious. These birds acted like typical hand-reared chicks. Sandhill Cranes reared at Patuxent and Red-crowned Cranes at ICF, by contrast, were nervous and flighty like wild caught birds.

Human Avoidance Conditioning.

For those chick-rearing methods where exposure to uncostumed humans is minimized, deliberate negative exposure called Human Avoidance Conditioning is often provided, especially if chicks appear too tame for release to the wild. Two different types of conditioning have been used successfully. Both are intended to train costume-reared chicks to differentiate between the costumed and uncostumed humans.

The first type of training involves normal handling. All positive and parental interactions (e.g., feeding and protecting) are done by a costumed parent. Slightly stressful interactions are done costumed, but with the chick hooded. For extremely negative procedures (e.g., taking blood from large chicks), the human is uncostumed. If the chick does not appear upset, upon release we chase it, yell, and clap our hands.

The second type of training (detailed below) involves mock attacks on the chicks. During these sessions, the live imprinting and socialization models are usually present so that their alarm calls verify the “danger” to the chick. Tape recorded alarm calls are played if no adults are available or if there is concern that the activity will not sufficiently alarm the adults. If facilities permit, visually isolate the birds that are about to be trained from the rest of the chicks (i.e., lock target chicks into their outdoor runs and lock non-target chicks indoors).

For costume-reared Mississippi Sandhill Cranes destined for release, training bouts began at about twenty days of age and were staged once or twice a month for chicks that were slow to develop wariness (Ellis et al. 1992). Once the chicks and adults are in place, one or two uncostumed humans surprise the chicks by bursting into view and racing through the chick area while shouting and making loud noises, sometimes banging on pots and pans to deliberately frighten the chicks. If a chick does not show fear (either by fleeing, freezing in an erect position, or squatting and hiding in the grass), the noisy humans pursue and grab the chick roughly then release it. The humans leave as abruptly as they appeared.

During Human Avoidance Conditioning, the costumed parent may or may not be present. If the costumed parent is with the chicks, the parent should flee from the humans or may turn and chase them away, thus protecting the chicks. If costumed parents are not present during the avoidance session, it is often helpful to have the costumed parent interact with the chicks shortly after the session in order to assess the effect of the training session.

Scheduled bouts of Human Avoidance Conditioning are probably not needed for already wary birds, but appear necessary for calmer, tamer birds. Nervous chicks or adult models can injure themselves by running or flying into fences, so caretakers should rush in quickly, end the activity as quickly as possible, and discontinue the “attack” if birds appear likely to injure themselves. Because all chicks receive a dozen or

so negative contacts with uncostumed humans during the rearing process, it is normally unnecessary to conduct more than 1-5 bouts of actual Human Avoidance Conditioning least the chick grow accustomed to the activity.

Veterinary Techniques for Rearing Crane Chicks

contributed by Glenn H. Olsen and Julia A. Langenberg

Each rearing method has advantages and disadvantages from a medical viewpoint. Survival of crane chicks averages higher in a more controlled environment (i.e., the hand-reared chick survival rate, from hatching to fledging, is often higher than for parent-reared chicks), however, medical management is only one factor in choosing which rearing method to use.

Good medical management of the crane chick begins with the care of the parents, especially the female. Her nutritional deficiencies or debilitating diseases may adversely affect the developing embryo (Olsen 1989; Olsen et al. 1990). In addition, infectious diseases and parasites carried by the adults may be passed to offspring either in the egg or directly to a hatched chick.

Preventative Health Program

Chicks should receive regular veterinary examinations especially during the critical first week (see Fig. 10.3 for health care schedules). The type and frequency of health problems seen in crane chicks will vary between species, between collections, and often between hand-reared and parent-reared chicks. A veterinarian should also review the chick’s weight gain and nutritional program.

If, in your environment, neonatal infections are rare, prophylactic antibiotic injections are not advised. Where advisable, give gentamicin (5 mg/kg) or amikacin (8 mg/kg) injections (Fig. 5.14) for the first 3 days. Frequent parasite examinations are part of a good preventative medicine program. Prophylactic treatment for parasites may be necessary if parasites are common in your collection (see Fig. 10.3). Screening for other infectious diseases (such as *Salmonella*) that are carried by adults and dangerous to chicks is also recommended.



Fig. 5.14. *Antibiotics and antihelminthics can be administered if infections are prevalent in a colony.* Photo David H. Ellis

Yolk Sac Problems

Exteriorized Yolk Sac. The yolk sac, a diverticulum of the intestine, is a major source of nutrition for the developing embryo and the newly hatched chick for the first 3 days of life. The yolk sac should be drawn into the abdominal cavity through the umbilicus prior to hatching. This retraction normally occurs with the spasmodic contractions of the abdominal muscles during hatching (Olsen 1989). Too high humidity during incubation, incorrect incubation temperatures, or pulling the chick from the egg too soon may all contribute to an exteriorized yolk sac (Fig. 5.15).



Fig. 5.15. *Exteriorized yolk sac.* Photo Glenn H. Olsen

If the yolk sac remains exteriorized, it may become torn or infected. If possible, gently manipulate an exteriorized yolk sac into the abdominal cavity after cleaning the area with a 10% povidone-iodine solution. The remaining umbilical opening can be closed using fine, absorbable sutures such as 4-0 Dexon (braided polyglycolic acid suture). Use a purse string pattern or 1-2 simple interrupted sutures. If the yolk sac cannot be manipulated into the abdominal cavity, it should be surgically removed using a ligature around the stalk (Hartman et al. 1987). Follow up care should include applications (twice daily) of povidone-iodine solution to the umbilicus or incision site and antibiotic injections (gentamicin, 5 mg/kg subcutaneous) twice daily for the first 3 days.

Another condition occasionally seen in chicks is delayed closure of the umbilicus, sometimes accompanied by a small (<5 mm diameter) yolk sac protuberance. When first seen, these small protrusions are often starting to dry and turn black as the knob is strangulated by the sealing umbilicus. In such cases, do not attempt to force the yolk sac remnant into the abdominal cavity. Rather, bathe the area 2-3 times daily with 10% povidone-iodine solution and maintain the chick on gentamicin or amikacin. Within 2-4 days the necrotic yolk sac remnant falls off and no further treatment is required. Until it sloughs off, these chicks should not be housed with other chicks to prevent penmates from pecking the umbilicus stump.

Omphalitis. Gram negative organisms, especially *Escherichia coli*, cause infection in the umbilical area or in the yolk sac (Flammer 1986). As a preventative measure, the chick should be hatched and reared in a clean environment. The umbilicus should be swabbed or sprayed with a dilute solution of povidone-iodine soon after hatching. If loss of young chicks from bacterial infections is common in your colony, maintain such chicks on antibiotics (such as gentamicin, 5 mg/kg daily) for the first 72 hours.

When an infection develops, clinical signs often include poor appetite, failure to grow, depression, a swollen abdomen, or reddening of the umbilical area. Culture the site to identify the organism and place the chick on antibiotics. Use fluid therapy if the chick is dehydrated, and swab the umbilical area with 1% solution of povidone-iodine. Surgical removal of the infected yolk sac has been used as an alternate treatment in other avian species (Kenny and Cambre 1992). This has been attempted several times in cranes, and the chicks often survive several days after the operation but succumb to peritoneal infections.

Yolk Sac Peritonitis. Occasionally, trauma or infection results from a rupture of the yolk sac within the abdominal cavity. Clinical signs include depression, poor appetite, abdominal distension, respiratory distress, weight loss, and sudden death. If peritonitis lasts several days, adhesions (scarring) within the abdominal cavity frequently occur. Supportive care (fluids, tube feeding, etc.) and treatment with antibiotics are recommended, but are often unsuccessful.

Respiratory Disease

Typical **signs** of respiratory disease in young chicks include open-mouth breathing, raspy breathing, a respiratory click, lethargy, reduced appetite, and cyanotic (blue) or pale mucous membranes. At Patuxent, parent-reared chicks are especially vulnerable to respiratory diseases following cool, rainy weather, especially if the parents are ineffective or inexperienced. At Patuxent, respiratory disease is more common in chicks raised by Florida Sandhill Cranes than in chicks raised by Greater Sandhill Cranes.

In chicks under 20 days of age, bacterial respiratory infections are most common following stress or chilling. In older chicks, disseminated visceral cocci-diosis and fungal infections are the most important causes of respiratory disease. Cultures and a cytology workup from the anterior choana (roof of the mouth) and trachea can be important in arriving at a diagnosis.

Initial treatment will include **antibiotic therapy**: gentamicin, amikacin, piperacillin sodium, and enrofloxacin are all good choices prior to receiving antibiotic sensitivity test results (see Table 8.1). Supportive care is important including fluid therapy for dehydration and tube feeding if anorexia is a problem.

Nebulization therapy is often beneficial to chicks with respiratory infections. Nebulizing oxygen itself is helpful for a dyspneic or cyanotic chick. Therapy should not exceed 1 hour daily, and should be divided into 2-3 equal time periods. Antibiotic medications used include: erythromycin (200 mg in 10 mL saline), gentamicin (50 mg in 10 mL saline) (gentamicin is not absorbed by respiratory epithelium and therefore does not effect concurrent injectable doses), polymyxin B (333,000 U in 5 mL saline), sulfadimethoxine (200 mg in 15 mL saline), and tylosin (100 mg in 10 mL saline) (Spink 1986). A mucolytic agent, such as acetylcysteine, can be added to the nebulizing

solution to help reduce thick mucous secretions. Acetylcysteine is given at 1 mL (2% solution) in 15 mL saline or saline/antibiotic combination.

Nebulization of chicks is done with the same equipment as recommended for adults (see Chapter 8). An Ultra-Neb 99 nebulizer or similar product producing a small particle-size mist is most effective. The small disposable cups with 30 cc maximum capacity are most effective, as small quantities of medicine can be mixed for each nebulization. The only difference is that the cage for the chicks is smaller. At Patuxent, we use a Snyder oxygen cage (see Appendix).

Fungal infection, specifically aspergillosis, is another cause of respiratory disease. Aspergillosis can also occur as a secondary infection in a chick already compromised by bacterial pneumonia and long-term antibiotic therapy. The diagnosis of aspergillosis can be made by radiography, respiratory cytology, culture, or serology. One effective antifungal treatment used in a variety of bird species is to nebulize with amphotericin B (Olsen 1991; Olsen et al. 1995). A solution is prepared by adding 2 cc of stock solution (5 mg/cc amphotericin B) to 15 cc sterile water. Amphotericin B has the potential for forming a precipitate with saline, therefore sterile water is preferred. Birds are nebulized for 20 min twice daily. Amphotericin B can also be administered intratracheally or intravenously (1 mg/kg 2-3 times a day). In addition, fluconazole is given orally at the rate of 100 mg/kg twicedaily for up to 30 days, oritraconazole (6 mg/kg) twice daily for up to six months.

Another product used for nebulization is clotrimazole. This product is nebulized in a small pediatric nebulizer. We use 3 cc per nebulization, using O₂ to produce the mist. Birds remain in the nebulizer 20-30 min twice daily with a schedule of 3 days on nebulization, 2 days off for up to one month.

Diarrhea and Cloacal Prolapse

Diarrhea. Crane chicks, both parent-reared and hand-reared, sometimes develop diarrhea around day 6. Cultures often yield heavy growth of *E. coli* suggesting it as the causative organism. However, *E. coli* is normally found in healthy chicks. Therapy includes antibiotics (see Table 8.1), oral kaolin/pectin, or bismuth subsalicylate (Pepto-Bismol) to reduce diarrhea, and subcutaneous or intravenous fluid supplementation to correct dehydration (use lactated Ringer's solution).

In older chicks, several causes of diarrhea have been identified including bacterial infections, parasites, reactions to medications, and gastrointestinal foreign bodies. Symptomatic treatment similar to that described above is used until laboratory results indicate a specific diagnosis.

Cloacal prolapses occur in crane chicks. Some are secondary to diarrhea and some are associated with chronic vent dermatitis. Most prolapses respond to topical treatment with lubricants (petroleum jelly), steroids, or Preparation-H, but a few need surgical replacement (see Chapter 8). Using gentle finger pressure or a moistened, cotton-tipped applicator, the prolapsed cloaca is carefully reinserted and then held in place using a purse-string suture (Fig. 8.18d) in the skin around the vent opening.

Dehydration. Dehydration, associated with many diseases, often results in death if left untreated (see Table 5.2). Weighing sick chicks daily, or even twice daily, allows the clinician to monitor fluid loss. Packed cell volume, plasma total solids, BUN (blood urea nitrogen), and uric acid can also be used to monitor hydration (see Table 8.3) because all are elevated in dehydrated birds. Without proper hydration, other therapeutic measures are not as effective.

In the severely debilitated chick, intravenous bolus therapy (Redig 1984; Harrison 1986) is the most effective initial treatment. For a less severely ill bird, oral or subcutaneous administration can be used. The best site for subcutaneous fluids is along the sides just behind the wings.

The fluid needs of a bird can be estimated by calculating the daily maintenance need (44 ml/kg body weight) plus the dehydration deficit (5-10% is average, see Table 5.2). Generally, 50% of the calculated deficit should be replaced in the first 12 hours. During the next 24-hour period, 25% of the deficit plus maintenance should be given.

The rapid restoration of fluid balance in the debilitated chick is probably more important than the type of solution used, providing that the fluid is isotonic. Lactated Ringer's solution (similar in composition to avian plasma [Redig 1984]), normal saline, or half-strength lactated Ringer's solution (mixed 50:50 with 2.5% dextrose) are often used. Fluids should be administered at body temperature, so a supply of warm fluids (37-39° C; 97-102° F) can be kept in an incubator or each bolus can be heated in warm water.

Heat stress. During weather with high temperatures and high humidity, some birds exhibit symptoms of heat prostration or heat stress. Whooping and Siberian Crane chicks appear to be especially susceptible. Signs of heat stress include open-mouth breathing, panting, wings held away from the body, and staggering. If no action is taken, a chick can suffer brain damage or die. The bird should be immediately moved indoors or into the shade, and should be cooled with cold water. Fluids should be given intravenously or subcutaneously to counteract stress and shock.

Heat stress is often associated with handling birds in hot weather. If a bird must be handled when ambient temperatures exceed 32° C (90° F), move the chick to a cool, shaded, or air-conditioned environment or handle only in the cool early morning hours.

Nutritional Support

Insufficient intake of calories leads to cachexia and emaciation. The bird will first mobilize body fat and then will catabolize muscle. Because young crane chicks do not have large fat reserves, loss of muscle tissue can occur rapidly and early in disease processes. Signs of emaciation in birds include a prominent keel and translucent skin due to lack of dermal fat (Lowenstine 1986). Crane chicks do not have well-developed pectoral muscles prior to flight. Therefore, assessment of pectoral muscle mass (Body Condition Index, Fig. 8.5), even though a valid technique in adult-sized cranes, is not used in chicks. Rather, the muscles surrounding the caudal, thoracic, and lumbosacral spine (palpated as a soft flat mass lying between the shoulders and to the side of the dorsal processes of the anterior portion of the synsacrum) are evaluated. These muscles are depleted in the emaciated crane chick.

Daily maintenance energy requirements for the crane chick should be calculated. Approximate caloric maintenance requirement is determined by finding the basic metabolic rate (BMR), $BMR = K(W_{kg})^{0.75}$, where K equals a theoretical constant for kilocalories and W_{kg} is the bird's weight in kg (Quesenberry et al. 1989). For cranes, $K = 78$, therefore $BMR = 78(W_{kg})^{0.75}$. The daily energy requirements in kilocalories (Kcal/day) are normally at least 1.5 times the BMR (see also Nutritional Support of a Sick Crane section of Chapter 8).

Blood glucose levels are useful in determining the degree of nutritional depletion. Normal Florida Sandhill Crane chicks at ICF maintained blood glucose levels over 200 mg/100 mL from hatching through fledging. No critical levels have been determined in cranes, though levels as low 80 mg/100 mL have been documented in emaciated chicks. Values less than 50 mg/100 mL are considered critical in birds of prey and lead to hypoglycemic convulsions and coma (Lowenstine 1986). Immediate correction of low blood glucose is best accomplished with intravenous or subcutaneous administration of 2.5% dextrose in half-strength lactated Ringer's solution.

Any bird suffering from cachexia and emaciation should receive a thorough examination to determine the cause of the condition (disease, diet, and management problems are all possible). If the chick is failing to gain weight, supplemental feeding should be initiated. Long-term caloric and nutritional support of debilitated chicks is best accomplished with oral alimentation (tube feeding and gavage are synonyms) using a flexible rubber tube (Fig. 5.16) made from a French urinary catheter (size 5 to 12 depending on the chick's age) mounted on the tip of a 5-60 cc syringe. Pass the tube over the tongue and down the esophagus to the level of the thoracic inlet. Palpate the neck to locate the tube and to assure that you are not in the trachea. If the tube is in the esophagus, you will palpate two cylindrical structures, the tube and the trachea. Delivering food in the trachea will be fatal for the chick.

Chicks under 10 days of age can be tube fed every 2-3 hours if necessary. For safety, start with about 3 cc for a hatchling; use larger amounts (up to 100 cc) for older chicks. Chicks weighing less than 100 g may be unable to receive 3 cc/feeding; administer the liquid diet slowly and watch responses. If the chick starts to regurgitate, stop the tube feeding, clean out the mouth, and gently stroke the neck in a downward motion. On subsequent tubings, decrease either the rate or the amount of formula to prevent further regurgitation. Because tube feeding also contributes to fluid balance, adjust total fluid therapy accordingly.

For extremely debilitated chicks of any age, Lafeber's Emerald I (see Appendix) is very helpful. This product contains only carbohydrates and is used for chicks too sick to digest anything else. It also helps elevate dangerously low blood glucose levels. A second product, Emerald II, has protein, fat, and fiber for crane chicks that can tolerate more nutrition (i.e., chicks that are not affected by gastrointestinal stasis).



Fig. 5.16. Glenn Olsen tube feeding a Whooping Crane chick.

Photo David H. Ellis

Another formula, known affectionately as Mother O'Malley's Crane Stew, is listed in Table 5.4 with two variations. The basic formula is used for severely debilitated adults or chicks. If a crane can benefit from complex nutrients, crane pellets are added using starter pellets for chicks or maintainer pellets for adults. The fine solids in this tube-food will give the chick's digestive system something substantial to process, and are believed to stimulate the chick's appetite and normal digestive processes better than a more easily digestible food. At Patuxent, young chicks have gained weight when fed solely on this tube feeding diet.

Severely debilitated adults should be fed the original formula (Table 5.4); however, most others can be fed one which includes adult pellets. Several cranes at Patuxent have survived solely on this diet, and even gained weight over the course of a month.

In addition, *Lactobacillus* products (1.4 tsp/kg; 7.0 g/kg) have been given to both young and adult cranes to promote digestion and to restore normal gastrointestinal flora. However, there have been no studies in cranes documenting the effectiveness of this therapy.

TABLE 5.4

Mother O'Malley's Crane Stew—Basic Formula

4 cups (946 mL) of warm water

2 tablespoons (30 mL) Vionate (or other vitamin powder)

4 heaping tablespoons (80-100 mL) Prosobee, Isomil, or other soy-based powdered infant formula

1/3 tube of Nutri-Cal (concentrated food for debilitated animals, sold in a 4¼ oz [120 g] tube)

1/4 cup (59 mL) vegetable oil

2 cups (274 mL) dry baby cereal, preferably mixed style

Water

Mix all ingredients in a blender (minimum 5 cup [1,200 mL] container) and process on high speed until smooth. If the formula seems too thick, add a small amount of water. Mix well before drawing up, and bring to about 21° C (70° F) or warmer before feeding. This formula can be divided into small containers and frozen for up to three months. After defrosting, the formula should be mixed thoroughly before use.

Variation 1: For chicks that can benefit from complex nutrients, add 2 cups (ca 250 g) crane starter pellets to the original recipe. Put the starter pellets in a 4-cup (946 mL) container, and add hot water to the top. Allow pellets to soak until fully expanded and soft (5-20 min). Place half of the pellet mixture into a 5-cup (1,200 mL) blender. Add enough water to blend the pellets easily (1-2 cups, 137-274 mL) and blend on high speed. Strain all the material through a fine sieve. Discard the solids. (This is a tedious process involving straining and constant stirring to enable the fine solids to pass through the mesh. Without this step, however, none of this food could pass through the small tube needed for young chicks.) To flush fine solids through, you may occasionally need to add more water. Once the solids are strained out, use this for the base and add the rest of the ingredients in the original formula.

Variation 2: For tube feeding sick older chicks (after all primaries are grown) and sick adults, substitute adult crane pellets for the starter pellets and eliminate the straining step. This food should be thick and able to pass through a large tube, although it may block occasionally when some of the coarser solids swell.

Ophthalmic Conditions

Eye injuries ranging from traumatic conjunctivitis to punctures of the cornea have been observed in crane chicks. A common cause is one chick pecking at another's eyes. Other causes of traumatic eye lesions include sharp objects (e.g., wires and thorns), abrasive cage materials such as sand substrate or wire partitions, and self-inflicted injuries caused by the chick flailing with its sharp toenails. Fluorescein dye is used to determine the presence and extent of corneal defects. Extensive lacerations, including corneal lacerations, can be sutured using fine (5-0 to 7-0) suture material. Antibiotic ophthalmic drops or ointments are also used.

Ocular discharges can result from debris (especially sand or wood chip bedding) under a lid, traumatic injury, a respiratory infection, or infection of the eye. The preferred treatment for debris in the eye is to pull the lid away from the eye and flush the debris out with a saline eye wash squirted from a plastic squeeze bottle or syringe (without needle). After flushing, apply antibiotic ophthalmic drops to the eye every 2-4 hours or ointments 2-3 times/day. Because ointments are

more viscous than drops, they are more likely to allow pieces of bedding to cling to the eye area. As a result, drops are often preferable to ointments in young birds.

Severe corneal infections associated with *Pseudomonas aeruginosa* have been seen in Whooping and Siberian Crane chicks (Miller et al. 1994). These infections are probably secondary to minor corneal trauma, but rapidly progress to complete corneal destruction and perforation. If ocular discharge does not decrease quickly with topical antibiotic treatment, a culture should be evaluated and intensive topical and parenteral treatment with an aminoglycoside antibiotic should be started.

Orthopedic Problems

Beak Deformities. Chicks should be observed daily for signs of abnormal beak growth which can lead to permanent beak crossing or malocclusion (popularly known as wry bill or screw bill). Most beak deformities are seen at a young age, and some can be corrected with careful beak trimming or by applying a splint to the beak for 2 to 6 hours each day for several

days. Beak deformities have also been observed following exposure to mycotoxin contaminated feed (Olsen et al. 1995).

Wing Problems. When the rapid growth of the primary and secondary feathers exceeds the development of the muscles and other support tissues in the wing, the wing may rotate outward at the carpus or may droop. This condition, called **angel wing**, can also be associated with excessive protein in the diet or too-rapid growth. Larger species are more prone to this condition. Supporting the affected wing in a normal position, using elastic bandage (Vetwrap) or adhesive tape in a figure-8 bandage around the carpus and radius/ulna or elbow, is the best means of correcting the problem (Fig. 5.17). To use adhesive tape, tear off a 2.5 cm (1 in) wide strip 30-60 cm (12-24 in) long, depending on the bird's size. Fold the adhesive strip over longitudinally leaving only about 5-10 cm (2-4 in) at one end with the adhesive side exposed. Wrap the folded tape around the hand (metacarpals) and forewing (radius-ulna), securing the wing in a normal folded position. Continue to wrap the tape around the wing eventually sticking the exposed adhesive section to the folded section of tape. No adhesive tape should be allowed to stick to the down or feathers.

The wing should be bandaged for **two days only** in any one treatment. Tape left longer than 2 days can alter feather (or even bone) growth, and can constrict blood vessels in the rapidly growing wing. Usually one treatment is sufficient to correct angel wing, however, additional bandaging may be required if the problem is still evident. Generally, a 2-4 hour rest without a bandage is allowed between successive two-day bandaging episodes.

During development, feathers or blood quills, especially the large primary or secondary feathers, can



Fig. 5.17. *Sandhill Crane chick taped for angel wing.*

Photo Glenn H. Olsen

break due to trauma, aggression, excessive grooming, or with handling. Blood loss from damage to a large feather may be significant and can lead to shock. Immediate treatment is to remove the bleeding feather by grasping the shaft with hemostats or pliers at the base and pulling it directly out. In most cases, the hemorrhage will stop immediately. When hemorrhage continues, the hole from which the quill was removed can be packed with gel-foam or hemostatic powder and the wing bandaged with a figure-8 wrap so pressure is applied to the point of hemorrhage. The bandage can be removed a few hours after treatment. If the bird is in shock or if the blood loss has been excessive (>10% of blood volume), fluid therapy should be given immediately (see Dehydration).

Foot and leg problems. Foot and leg problems are common during captive rearing. Their frequency can be reduced through careful attention to diet, adequate exercise, proper substrate, controlled weight gain, and proper handling methods (Olsen 1994). When these problems do occur, early treatment is critical to normal chick development.

Curled toes are seen immediately post-hatching and may result from incubation or genetic problems. Curled toes range in severity from mild cases that respond to treatment with splints to severely club-footed chicks that fail to respond to any therapy and must be euthanized. Recognizing the problem can, at times, be difficult because neonatal cranes normally have edematous (swollen) legs and feet. The toes may appear curled, but are normal when the edema subsides. Splinting of toes is usually not done until 1 day post-hatching, unless the chick is unable to stand or eat (Olsen 1994).

Deviated or **crooked toes** are frequently encountered with hand-reared chicks at any age prior to fledging. The deviations appear to be due to laxity of the ligaments and tendons of the toes. Whether this laxity is due to problems with substrate, exercise, nutrition, or a combination of all three, is not clear. The most commonly encountered toe deformity is a single bent or curved digit. Hand-reared chicks under 10 days of age frequently have one or two bent digits. Older chicks of larger species frequently have curved middle toes.

To detect the problem, observe the gait of each chick daily; the middle toe should point straight forward with the other toes pointing ca 45° to each side. Toe misalignment can be corrected with a splint (Olsen 1994); sometimes only 1-2 days of support may be adequate. Splints made of small wooden dowels

(1.5-4.0 mm dia) or popsicle sticks are taped to the crooked toe using low-tack tape such as filament packing tape (Fig. 5.18) (Olsen 1994). For chicks under 3 days of age if two or more toes are crooked, the foot can be taped in a normal walking position to a thin cardboard or plastic “snowshoe” splint (Fig. 5.19). Snowshoe splints cause problems in older chicks (i.e., the birds fight the snowshoe, the snowshoes collect feces and dirt, and they are slippery), but if used they can be made using moldable cast material (Orthoplast or Roylan Polyflex II, see Appendix). The splint should be removed every 1-2 days and the foot reevaluated. Leave the snowshoe off for several hours before replacing it. Snowshoes are usually helpful for chicks under three days of age. After that age, applicator sticks and tape work better.

When using wooden applicator sticks as splints, cut the stick to lie parallel to the chick's toe from the



Fig. 5.18. *Taping for crooked toes using dowels and filament tape.*

Photo Glenn H. Olsen

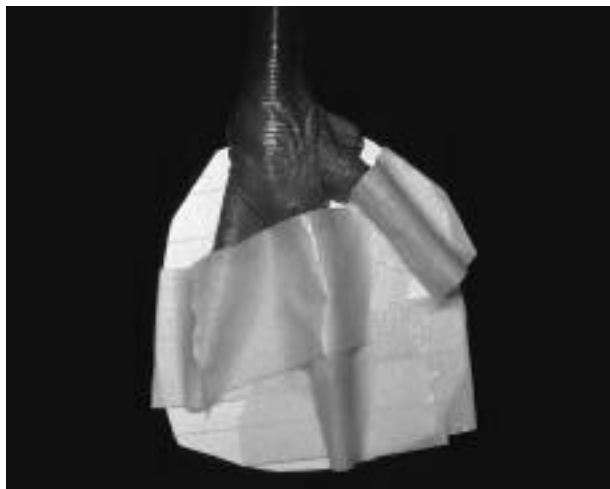


Fig. 5.19. *“Snowshoe” splint for crooked toes.*

Photo Glenn H. Olsen

junction of the foot to the toenail. Sand the cut ends of the stick or cover the ends with tape to avoid abrasion. Place the applicator stick against the outside of the curve on the side of the toe. Do not place the stick above or beneath the toe. If the toe is rotated as well as laterally bent, try to twist it gently back to the normal position.

When chicks are older than 14 days, their toes cannot be adequately straightened by one small applicator stick. Instead tape two sticks together as the brace, or use one stick on each side of the toe. Using pre-cut strips of low-tack tape, wrap the tape around the toe. Do not pull tape tightly, but rather place the tape against the toe and loosely wrap it. Use the minimum amount of tape to do the job, but the toe should be completely encased from toenail to foot (Olsen 1994). Leaving part of the toe exposed can cause circulatory problems. Leaving the splint on young chicks longer than two days can cause constriction of blood vessels and damage the toe.

Some chicks limp when a toe splint is applied. When this occurs, make sure the tape is not too tight, and observe the chick for other leg problems which can occur if a chick does not adjust to a toe splint. Crooked toes, if uncorrected, can eliminate birds from release programs, can leave adults severely deformed (Fig. 5.20), may inhibit natural breeding, and may lead to arthritis and bumblefoot as birds age.

In newly hatched chicks, deviation of the legs from the hip area, called **splayed leg**, is associated with improper incubation or chicks raised on slippery surfaces, but it may also occur spontaneously. Hobbling the legs above and/or below the hocks for 1-2 days in a normal position (Fig. 5.21) using adhesive tape or elastic bandage can be helpful (Olsen 1994). In some parrots, this condition responds to vitamin E and selenium injections (Harrison 1986), but the effects on cranes are unknown.

Hand-reared crane chicks sometimes develop other deviations of the legs (e.g., **leg rotation**, **angular limb deformities**, or **bowed legs**). The most common form is angling or rotation of the leg below the hock, although inward rotation is also occasionally seen. A chick's leg position and gait should be closely observed and checked daily by caretaker staff. The middle toes should be parallel and point forward; if one deviates, examine that leg carefully for changes at or below the hock. The causes of this problem are not known, although improper diet, excessively rapid growth, inadequate exercise, and genetics probably all play a role (Serafin 1980, 1982). Treatment is much more



Fig. 5.20. Severely deformed toes in an adult Sandhill Crane.

Photo David H. Ellis



Fig. 5.21. Whooping Crane taped to control leg rotation.

Photo Patuxent

successful when the problem is detected early and at a young age. Tension taping of the rapidly growing side of the hock (i.e., outside of the curve) using a strip of adhesive tape, can be effective in slowing growth on one side of the growth plate to allow the leg to straighten out (Haefner 1988). Tape hobbles or splints have also been used with limited success to reposition deviating legs. For severe cases, surgical correction using techniques like periosteal stripping and wedge osteotomies have been tried by ICF, but with limited success.

All splinting/taping methods must be accompanied by correction of the contributing causes. If lack of exercise is suspected as a contributing cause, the safest way to increase exercise once a deviation has developed is hydrotherapy, swimming the chick at least twice daily for at least 20 min each time. Supervised walking can also be helpful. Excessive weight gain can be controlled by limiting food availability, increasing exercise, and monitoring weight carefully. Food can be

withheld for part of the day, or food and water can be placed at opposite ends of the enclosure.

Fractures can occur in crane chicks and are usually associated with trauma. Fractures at growth plates are commonly seen and are the most difficult to manage. The same external splints and internal surgical correction techniques are used for chicks and adults. Wing fractures often heal, but with some loss of wing function. This is usually not a problem for captive birds, though it can affect natural fertility if the bird is a male. Fixation of leg fractures is only occasionally successful. Chicks frequently die from complications post-surgery such as premature closure of growth plates, osteomyelitis, and stress-related diseases. Survival depends on intensive care, and even then, survivors are difficult to resocialize with penmates after healing.

Parasites

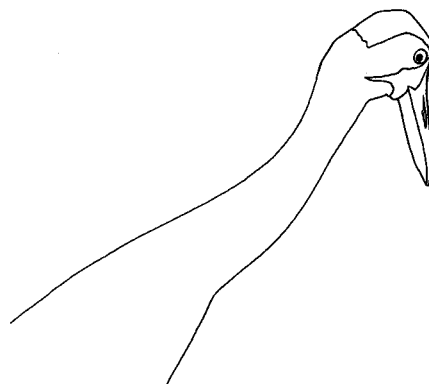
Helminth parasites can severely debilitate crane chicks. Gapeworm (*Cyathostoma* sp. and *Syngamus* sp.), capillarids, and ascarids are the common nematodes of captive cranes. *Acanthocephala* sp. can cause intestinal perforation and peritonitis. Close monitoring of chicks for parasites is essential. Weekly laboratory testing for parasite eggs in the feces is recommended (see Chapter 8). However, feces can test clear even when chicks are infested, especially with gapeworms. Infected chicks can be treated with ivermectin (1% solution 0.02 mg/kg subcutaneously or orally), fenbendazole (100 mg/kg orally), or pyrantel pamoate (4.5 mg/kg orally). Often two treatments 7-10 days apart with one of these anthelmintics is needed to clear the parasites; repeat fecal examinations should be done 10-14 days after treatment to insure that the parasites are gone. Prophylactic doses of these medications can be given if there is a history of parasite problems in the flock. (See Fig. 10.3 for an example of a parasite screening and prophylactic medication schedule.)

Coccidiosis, from infection with *Eimeria gruis* or *E. reichenowi*, is a particularly devastating disease in crane chicks. In cranes, coccidiosis is not just a gut parasite; it can also be visceral (i.e., the organisms invade the internal organs including the heart, liver, lungs, and kidneys). Because coccidiosis is a clinical problem mostly in young cranes, it is recommended that a coccidiostat be used in the food and/or water of crane chicks. Very often parent-reared chicks initially consume mostly insects, so the coccidiostat may need

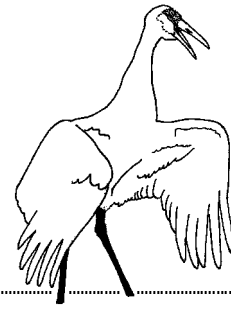
to be supplied in the water. Amprolium in food and water (0.0175%) or monensin sodium in food (90 g/ton) can both be effective coccidiostats. However, if a treatment regime using one of these two drugs is used long-term without alternation, it becomes less effective as resistant strains of the parasite develop. Adults should be monitored for the presence of oocysts, and treated as appropriate. This will reduce pen contamination and the exposure of the chicks. Chicks and adults with coccidia can be treated with trimethoprim-sulfamethoxazole, sulfadimethoxine, metronidazole, nitrofurazone, or pyrimethamine (see Chapter 8 for details).

Literature Cited

- Ellis, D. H., G. H. Olsen, G. F. Gee, J. M. Nicolich, K. E. O'Malley, M. Nagendran, S. G. Hereford, P. Range, W. T. Harper, R. P. Ingram, and D. G. Smith. 1992. Techniques for rearing and releasing nonmigratory cranes: lessons from the Mississippi Sandhill Crane program. *Proceedings North American Crane Workshop* 6:135-141.
- Flammer, K. 1986. Pediatric medicine. Pages 634-650 *in* G. J. Harrison and L. R. Harrison, editors. *Clinical avian medicine and surgery*. W. B. Saunders, Philadelphia, Pa.
- Haefner, S. 1988. Correcting leg and joint abnormalities in long-legged precocial birds, with notes on prevention. Pages 563-550 *in* *Proceedings of the Regional Conference of the American Association of Zoological Parks and Aquariums*, Pittsburgh, Pa.
- Harrison, G. J. 1986. What to do until a diagnosis is made. Pages 356-361 *in* G. J. Harrison and L. R. Harrison, editors. *Clinical avian medicine and surgery*. W. B. Saunders, Philadelphia, Pa.
- Hartman, L., S. Duncan, and G. Archibald. 1987. The hatching process in cranes with recommendations for assisting abnormal chicks. Pages 387-396 *in* J. C. Lewis, editor. *Proceedings 1985 Crane Workshop*. Platte River Whooping Crane Habitat Maintenance Trust and U.S. Fish and Wildlife Service, Grand Island, Nebr.
- Hartup, B. K., and R. H. Horwich. 1994. Early prenatal care and chick development in a cross-fostering trial with White-naped (*Grus vipio*) and Greater Sandhill (*Grus canadensis tabida*) Cranes. *Bird Behavior* 10:21-27.
- Kenny, D. and R. C. Cambre. 1992. Indications and techniques for the surgical removal of the avian yolk sac. *Journal of Zoo and Wildlife Medicine* 23:55-61.
- Kepler, C. B. 1978. Captive propagation of Whooping Cranes: a behavioral approach. Pages 231-241 *in* S. A. Temple, editor. *Endangered birds: management techniques for preserving threatened species*. University of Wisconsin Press, Madison.
- Lowenstine, L. J. 1986. Nutritional disorders of birds. Pages 201-212 *in* M. E. Fowler, editor. *Zoo and wild animal medicine*. W. B. Saunders, Philadelphia, Pa.
- Miller, P. E., J. A. Langenberg, L. A. Baeten, and C. P. Moore. 1994. *Pseudomonas aeruginosa*-associated corneal ulcers in captive cranes. *Journal of Zoo and Wildlife Medicine* 25:449-454.
- Olsen, G. H. 1989. Problems associated with incubation and hatching. Pages 262-267 *in* *Proceedings of the 1989 Annual Meeting of the Association of Avian Veterinarians*, Seattle, Wash.
- Olsen, G. H. 1994. Orthopedics in cranes: pediatrics and adults. *Seminars in Avian and Exotic Pet Medicine* 3(2):73-80.
- Olsen, G. H., J. M. Nicolich, and D. J. Hoffman. 1990. A review of some causes of death of avian embryos. Pages 106-111 *in* *Proceedings of the 1990 Annual Meeting of the Association of Avian Veterinarians*, Phoenix, Ariz.
- Olsen, G. H., J. W. Carpenter, G. F. Gee, N. J. Thomas, and F. J. Dein. 1995. Mycotoxin caused disease in captive Whooping Cranes (*Grus americana*) and Sandhill Cranes (*Grus canadensis*). *Journal of Zoo and Wildlife Medicine*. In press.
- Putnam, M. S. 1981. Refined techniques in crane propagation at the International Crane Foundation. Pages 250-258 *in* J. C. Lewis, editor. *Proceedings 1981 Crane Workshop*. National Audubon Society, Tavernier, Fla.
- Quesenberry, K. E., G. Mauldin, and E. Hillyer. 1989. Nutritional support of the avian patient. Pages 11-19 *in* *Proceedings of the 1989 Annual Meeting of the Association of Avian Veterinarians*, Seattle, Wash.
- Redig, P. T. 1984. Fluid therapy and acid base balance in the critically ill avian patient. Pages 59-73 *in* *Proceedings of the International Conference on Avian Medicine*, Toronto, Ontario. Association of Avian Veterinarians.
- Serafin, J. A. 1980. Influence of dietary energy and sulfur amino acid levels upon growth and development of young Sandhill Cranes. Page 30 *in* *Proceedings of the Annual Meeting of the American Association of Zoo Veterinarians*, Washington, D.C.
- Serafin, J. A. 1982. The influence of diet composition upon growth and development of Sandhill Cranes. *Condor* 84:427-434.
- Spink, R. R. 1986. Aerosol therapy. Pages 376-379 *in* G. J. Harrison and L. R. Harrison, editors. *Clinical avian medicine and surgery*. W. B. Saunders, Philadelphia, Pa.



Behavior Management



Scott R. Swengel, George W. Archibald,
David H. Ellis, and Dwight G. Smith

Management of crane behavior varies with use, age, and reproductive condition. We discuss five management classes: chicks, subadults, breeding birds, cranes for public display, and release birds. Cranes are individuals, and management practices that are beneficial to one crane may be harmful to another. Because management of these classes and individuals overlap, a combination of behavioral management techniques often works best for individual cranes.

This chapter emphasizes crane **social behavior** and how it affects captive management. Ellis et al. (1991) described the non-social (maintenance) behavior of all species. Because most body maintenance behavior patterns are superficially alike for both calm and highly stressed cranes, we discuss below only the few maintenance actions that suggest a crane is stressed. Studies of crane social behavior that include illustrations of displays are Allen (1952) on the Whooping Crane; Archibald (1974a) and Katz (1979) on the Hooded Crane; Masatomi and Kitagawa (1975) on the Red-crowned Crane; Poulsen (1975) on Siberian and Common Cranes; Voss (1976, 1977), Nesbitt and Archibald (1981), and Tacha (1981) on the Sandhill Crane; and Ellis et al. (*In prep.*) on all species. Previous papers describing behavioral management of captive cranes include Archibald (1974b), Kepler (1976, 1978), Archibald and Viess (1979), and Derrickson and Carpenter (1980, 1987).

Techniques in Behavior Observations

Observing cranes in the same context and at the same time each day reveals seasonal changes in social patterns and abnormalities suggesting health problems. Data collection can be as simple as merely noting abnormalities or social problems, or as complex as a bird-by-bird tabulation of behavior details on form

sheets. **Daily observations** are essential in monitoring the pairing and social interaction of cranes in the same pen or adjacent pens.

Blinds (Fig. 6.1) set up near cranes allow for longer-term observations of relatively undisturbed (by humans) behavior. Cranes behave more normally when people are not in view, so blinds are a valuable supplement to daily observations not just for research, but also in the management of pairs. Because some cranes remain disturbed by observers in nearby blinds, it is crucial to locate blinds with care and use one-way glass if necessary to enable the observer to be invisible to the crane.



Fig. 6.1. Brian Clauss enters an elevated observation blind.

Photo David H. Ellis

Remote monitoring (Fig. 6.2) via closed-circuit TV (CCTV) is valuable for observing pairing, breeding, or egg laying of cranes without risking them noticing the observer. Cranes seem to adjust quickly to cameras mounted high in a pen corner (although they notice cameras that move or make noise). CCTV allows for the most undisturbed watching of cranes and is ideal for making videotapes that can be replayed at high speed and searched for significant behavioral patterns, such as copulation and nest building. For cranes that break eggs, long-term egg vigils are made easier by CCTV. CCTV allows for a second look to determine if first impressions can be confirmed by review of videotapes.



Fig. 6.2. *Closed circuit television camera monitors Whooping Crane.*

Photo David H. Ellis

Aggressive Displays

Cranes that have red caps or bare skin on their heads expand the cap (**Crown-expansion**), extend their wattles (**Bare-skin-expansion**), or suffuse the bare skin with brighter color to supplement their displays (Fig. 6.3). Cranes with completely feathered heads elevate head plumage to increase the apparent size of their heads. These head displays are low-intensity displays when they appear alone, but they are also used as elements of many other aggressive displays.

Cranes have several low-intensity displays that are used to intimidate or repel intruders. Most of the displays described below apply to all cranes. The nomenclature for these activities follows Ellis et al. (*In prep.*). We will describe the displays in increasing order of intensity. In perhaps the lowest intensity display, a crane raises its head to full height and extends its neck upward and slightly forward in an

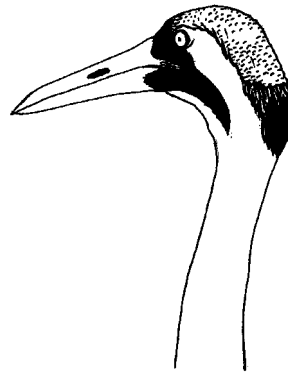


Fig. 6.3. *Crown expansion: Whooping Crane.*

Art Billi Wagner

Alert-posture. When in this posture, and during many of the social displays that follow, most cranes perform **Crown-expansion** or **Bare skin expansion**. The crane may then begin a ritualized display walk (**Horizontal-strut** or **Vertical-strut**) with the bill slowly bobbing up and down in time with exaggerated, rhythmic steps (Fig. 6.4). The toes

are rigidly fanned and extended during the **Vertical-strut**. The crane may either turn its bill away from the intruder (**Crown-present**) or direct its bill downward toward the intruder. In higher intensity strutting, the crane either increases the speed of the walk or lowers its head (sometimes nearly to the ground). Common, Hooded, Whooping, Black-necked, Sandhill, and Siberian Cranes (listed in descending order of feather elevation) raise their tertial feathers into a bustle during display walks (Fig. 6.4).



Fig. 6.4. *Strut (vertical): Eurasian Crane.*

Art Billi Wagner

Another low-intensity threat is the **Ruffle-bow**, in which the crane elevates its feathers and slowly, at first, ruffles its plumage until at length the whole body is rapidly shaking (Fig. 6.5). This display is much like the maintenance **Ruffle-shake**. All species perform the Ruffle-bow, and the Blue, Demoiselle, and Wattled Cranes use this as their principal display. Sandhill Cranes end the Ruffle-bow by throwing the head downward, then preening the breast or tibiotarsus. The Whooping Crane tucks the bill high at the end of its Ruffle-bow (Fig. 6.6). All cranes have a **Ritualized-preen** display in which they place the bill between the back and one wing (Fig. 6.7). Siberian Cranes lower the primaries of one wing and do not move the bill during the display. Other cranes do some rudimentary preening movements and periodically raise their heads to look around. Several species stamp their feet (**Stomp**) a few times in conjunction with the Ruffle-bow or Ritualized Preen. The Red-crowned Crane often Stomps, then arches its head back and raises its wings above the back until its head is nearly between the wings over the back (**Arch**). The Whooping Crane version of the Arch we call the Butterfly (Fig. 6.8). The Red-crowned Crane and its nearest relatives (see Chapter 1) sometimes raise their wings slightly during more intense versions of the Vertical-strut. The Brolga, Sarus, and White-naped Cranes have an exaggerated downward bow as part

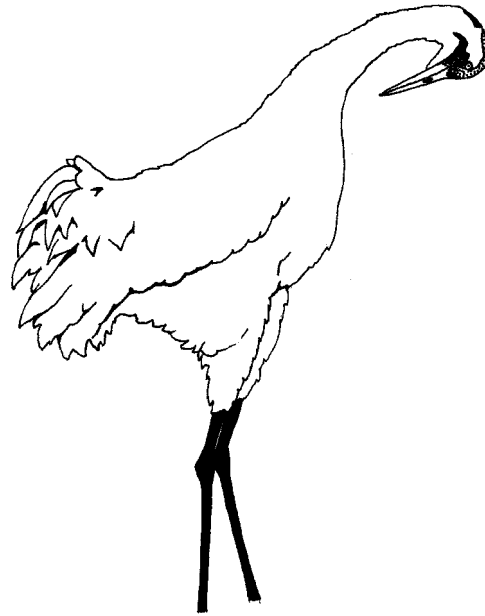


Fig. 6.6. *Ruffle-bow-up: Whooping Crane.* Art Billi Wagner

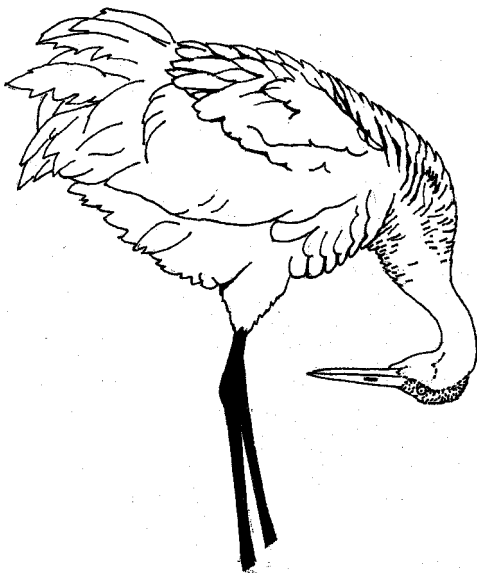


Fig. 6.5. *Ruffle-bow-down: Sandhill Crane.* Art Billi Wagner



Fig. 6.7. *Ritualized Preen: Whooping Crane.* Art Billi Wagner

of the Ruffle-bow. After the ruffle, they usually bow deeply forward, then do a Ritualized Thigh Preen or elevate the head far over the back.

We believe that the most aggressive display is the **Crouch**. In this display, the crane lowers to lying posture with its wings slightly to mostly spread and its bill in front, usually touching or probing at the ground. After performing the Crouch, cranes sometimes **Rush** an intruder by springing up suddenly and charging headlong, flapping their wings and gliding over the ground rapidly if the intruder fails to depart. The crane terminates the Rush with either a Stomp and Ruffle-bow, or by **Attacking** the intruder. Otherwise, cranes usually end the Crouch display with an Arch (Red-crowned) or Ritualized-preen (Siberian, Sandhill and others).

Methods of **Attacking** (Fig. 6.9) include spearing the opponent with the bill (**Bill-stab**), **Leaping** in the air and slashing the intruder with the inner toenails (**Jump-rake**), and thrashing with the wings (**Wing-thrash**). Cranes sometimes use stealth to approach an intruder: they circuitously walk closer and closer while feeding, then abruptly Rush the intruder (crane, human, or other animal). Rushes can also lead to aerial pursuit as a territory owner drives away an intruder, sometimes slashing at the intruder with its feet even during flight.

Aggressive cranes may also bill-spar with one another, spreading their wings and Bill-stabbing at their opponents while standing erect. This may

or may not lead to Jump-raking. Cranes also **Hiss** loudly while bill-sparring. A minor form of Attack is to peck at or grasp the wings or tail of a subordinate to displace it.

Vocalizations can indicate fear or aggression, and like the Unison-call, help drive away intruders and maintain pair bonds (Archibald 1976a, 1976b). Pairs **Unison-call** (Fig. 6.10) after repelling intruders. **Guard-calls**, which are short blasts separated by several seconds, also help to defend the territory (Archibald 1976b). See Archibald (1976a) and Chapter 11C, especially Fig. 11C.1, for more details of these vocal displays.

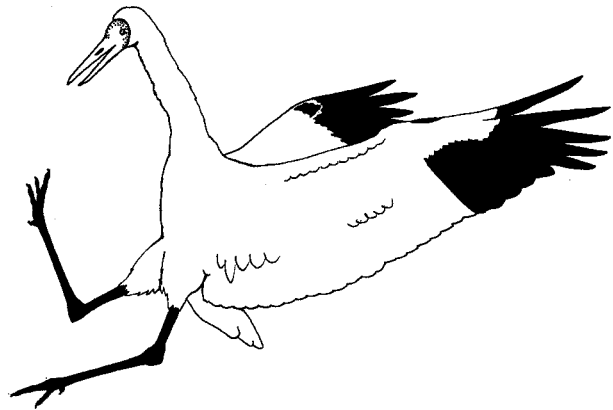


Fig. 6.9. *Jump-rake: Siberian Crane.*

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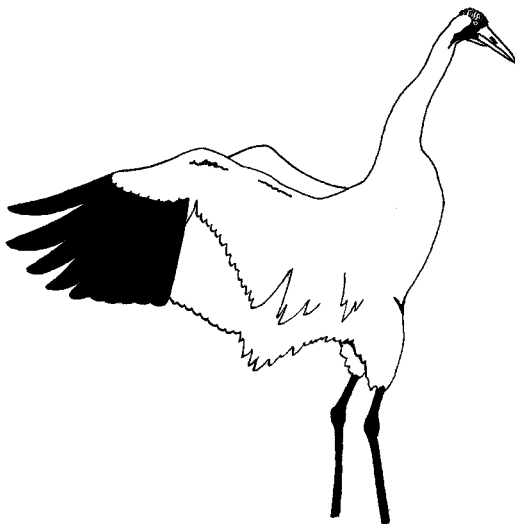


Fig. 6.8. *Butterfly: Whooping Crane.*

Art Billi Wagner



Fig. 6.10. *Unison-call: White-naped cranes.*

Art Billi Wagner

Dancing consists of bouts of Rushing, Flapping, Leaping, Tuck-bobbing (Fig. 6.11), gaping, and feather or stick tossing. Dances last from several seconds to a few minutes and are usually pair-related. However, young cranes often dance in apparent appeasement of dominant cranes. Sometimes Dancing includes sham Bill-stabs and Jump-rakes, but in stable pairs, physical contact is usually not present. Many other elements of aggressive displays (e.g., Crown-expansion) are evident in Dances.

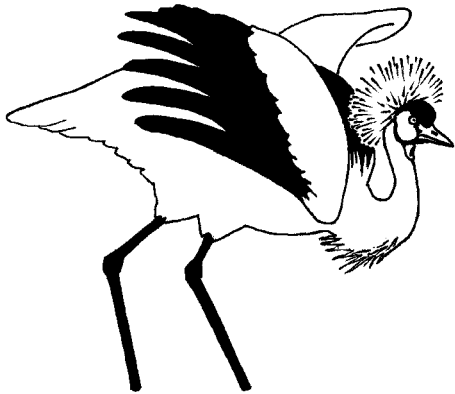


Fig. 6.11. *Tuck-bob: Gray-crowned Crane.* Art Bill Wagner

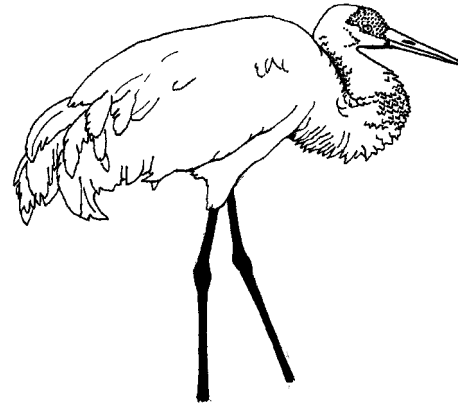


Fig. 6.12. *Cower: Sandhill Crane.* Art Bill Wagner

wings slightly (the elbow is lifted away from the flank), and give purring calls. Chicks often perform this display. When adult cranes Cower, they are probably reverting to chick behavior to placate a dominant crane or person. Often a submissive crane will spread its wings with the trailing edges drooped and turn its back to a dominant crane (or human) as in the adult female's **Pre-copulatory Display** (Fig. 6.13). This behavior is not limited to one sex and is often seen in chicks and subadults.

Submissive Behavior

Submissive displays and fleeing behavior are useful indicators that a crane is stressed by its environment. Some captive cranes run away when a person or dominant crane approaches and may push at the fence as if to walk through it. The crane may also nervously pace the fence, rake its feet along the fence in a climbing manner, and drag its neck and bill along the fence. These disturbance-related activities can cause physical damage, especially abraded wrists, and even more seriously, broken bills.

A submissive crane (Fig. 6.12) usually retracts its neck and adopts a hunched posture (**Cower**) with its head and neck feathers fluffed, and its crown, wattles, and/or bare skin patches contracted. Frequently, subordinate cranes lower their heads, spread their

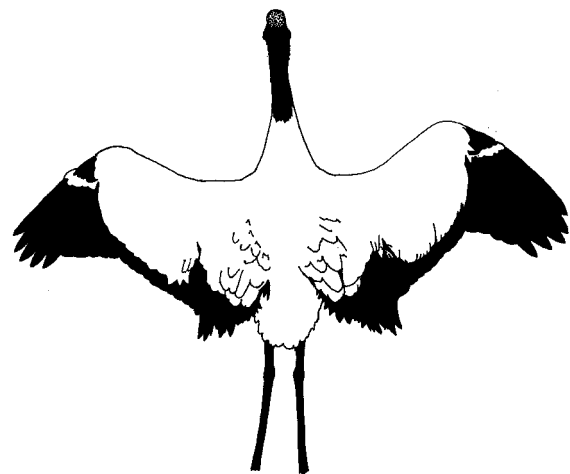


Fig. 6.13. *Pre-copulatory display: female Black-necked Crane.* Art Bill Wagner

Behavioral Management of Chicks and Subadult Cranes

This section provides a summary of the management of young chicks; other details are provided in Chapter 5. Chicks have strong social needs. Most species are gregarious, and nearly all cranes flock during the non-breeding season. Flocking probably results in increased survivorship and in foraging advantages. Cranes three months to three years old should be socialized with conspecifics so that they develop normal social behavior. Combine cranes in groups of two or more in pens that are at least 100 m² for two cranes and proportionately larger for larger groups. The group will establish a dominance hierarchy (Derrickson and Carpenter 1980) based on the size and sex of the cranes, with males and larger cranes typically being more dominant (Kepler 1976).

Parent-reared cranes will try to establish **vocal contact** with their parents after separation and will also try to reunite if allowed. Place such cranes at least 200 m away from and out of sight of their parents until they integrate into a social unit with other colts. Parent-reared crane chicks are typically fearful of people. As such, they should be habituated to humans if they are to remain in captivity (Archibald and Viess 1979). Frequent, nonstressful encounters with humans will help calm them down (e.g., provide a food treat when entering the pen). Placing wild colts with tame cranes will also help.

In grouping colts, avoid penning cranes together that are likely to be paired later. Cranes treat their penmates as siblings and may refuse to pair with them later. It is wise to keep intended mates separate until they are at least 18 months of age.

Cranes that are 6 months old or older often become aggressive towards their penmates, particularly those of the same sex. Groups should be reorganized when excessive aggression appears. However, it will help to place two food/water stations at opposite ends of the pen so submissive cranes can eat and drink. Even then, watch for cowering cranes that are afraid to go to the food. In large pens with 10-15 birds, a third station may be needed. Submissive cranes that are regularly attacked by penmates should be removed.

Chicks spend much time practicing flying when they are two or more months old and need an unobstructed area at least 15 m long for exercise.

They may be wing clipped after they fledge. Opinions vary on whether permanent flight restraint (see Chapter 11E for details) can prevent males from balancing properly during copulation and thus limit fertility. Many pinioned and tenotomized males fertilize their mate's eggs. However, in captive Red-crowned Cranes (the heaviest crane), full-winged males had higher rates of natural fertility than pinioned males (Belterman and King 1993).

Keep subadult cranes in genetically and behaviorally compatible groups (2 to 16 individuals) until they are paired. When pairs form in group pens, extreme aggression often appears, with the dominant pair often monopolizing food or water and occasionally killing pen mates. If the new pair in a group pen is desirable, move the pair to a breeding pen. If the pair is unwanted, remove one or both cranes to reduce aggression.

If sex is known, juveniles can be grouped by gender, however, same sex groups can also be aggressive, and homosexual pairs can form in such groups (Archibald 1974b; Kepler 1978; Derrickson and Carpenter 1987). Early sexing increases the chances of early reproduction through behavioral management.

Behavioral Management of Breeding Cranes

Cranes have pronounced species specific variation in nesting phenology (see Chapter 3). Their captive management should reflect species differences and allow for individual variation as well.

Annual Behavior Cycle of Breeding Cranes

Cranes exhibit seasonal cycles in social displays especially in activities related to pairing and those involved in rearing young. It would be helpful to have data on seasonal trends for all species of cranes in your colony, but such is available only for the Whooping Crane and Mississippi Sandhill Crane as described below. Also, Katz (1979) presented somewhat similar data for two pairs of Hooded Cranes from late February through early June.

For boreal species, display intensity ebbs in winter, but on warm days in late winter and early spring, display frequency and intensity increase. As the breeding season approaches, vocal and visual behavior

related to pair-bonding, territorial defense, and breeding is increasingly evident. There is also good evidence for decreases in performance of some displays during the molt and during the incubation and chick rearing period when the adults tend to be more secretive. The intensity and frequency of several vocal and visual displays again increases in fall, suggesting an autumnal recycling perhaps in response to a photoperiod mirroring that for spring. Thereafter, performance tendency wanes through the early months of winter.

In the following paragraphs, we will show some general trends in this annual cycle and compare behavior trends for the migratory Whooping Crane with those of the nonmigratory Mississippi Sandhill Crane. Male-female differences will also be emphasized. These generalizations are presented because of their usefulness in crane husbandry. Also there are seasonal cycles in non-social behavior such as the increase in fall food intake (or at least body weight) especially evident in northern latitude breeding cranes (Swengel 1992).

Our data on social displays derive from a 15-month period during which we conducted a standard morning "walk-through" in the crane colonies at Patuxent and recorded the responses to an approaching human. Because hand-reared cranes responded to the approaching human as though he was an intruding crane, for hand-reared pairs we were able to evaluate seasonal trends merely by recording all social displays as we walked through the colony. Our walk through the colony did not elicit social displays in cranes reared by crane foster parents (except for fleeing behavior) so parent-reared cranes were eliminated from the comparisons shown below. Data were used for 12 male and 12 female Whooping Cranes and 9 male and 7 female Mississippi Sandhill Cranes. All data were taken on form sheets as an observer (who was familiar to the cranes, but who avoided entering their pens) approached to within ca 2 m of the pen between 0.5 and 3.5 hours following sunrise.

As for all species with a red cap, Whooping Cranes, and to a lesser degree Mississippi Sandhill Cranes, expand the red, bare-skin areas and present this area toward approaching intruders (Fig. 6.3). For both species, Crown-expansion and presentation values are high throughout the year,

except during incubation or molt when a crane is trying to be inconspicuous. Crown contraction at other times signals fear or illness. The Whooping Crane exhibited two peaks. Females of both species cycle in concert with their mates, but they, on average, expand to a lesser degree than males.

The Unison-call (Fig. 6.10), a form of antiphonal duet given by both members of a pair, can be performed throughout the year and is given primarily in response to conspecific intruders (Walkinshaw 1973a; Archibald 1976b). Well-coordinated, frequent Unison-calling signals that a pair is properly bonded. Duetting is also believed to be important in the synchronization of the crane breeding cycle (Voss 1977). In the Whooping Crane pairs studied at Patuxent, the Unison-call was heard with some frequency all months of the year (Fig. 6.14), but was highest between October to April and lowest during the July molt. Unlike our Whooping Cranes which did not breed during the study year, our Mississippi Sandhill Cranes exhibited a strong peak from June through August when they did breed. This periodicity suggests the importance of the Unison call as a territorial display while breeding (Mississippi Sandhill Crane data) and also in reinforcing the bonding of a pair during migration (Whooping Crane data).

Contact- or Flight-calling was entirely absent in our pairs of the non-migratory Mississippi Sandhill Cranes, but our Whooping Cranes exhibited a strong peak in March and April and a minor peak in September (Fig. 6.15). The Contact-call communicates a bird's disposition to fly and is probably important in synchronizing long distance movements of the pair. Contact-calls may be given synchronously

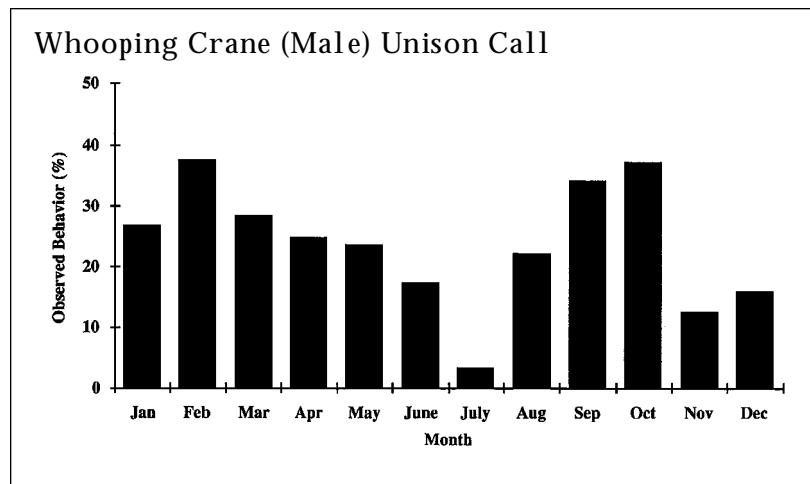


Fig. 6.14. Seasonal pattern in Unison-call performance for Whooping Cranes.

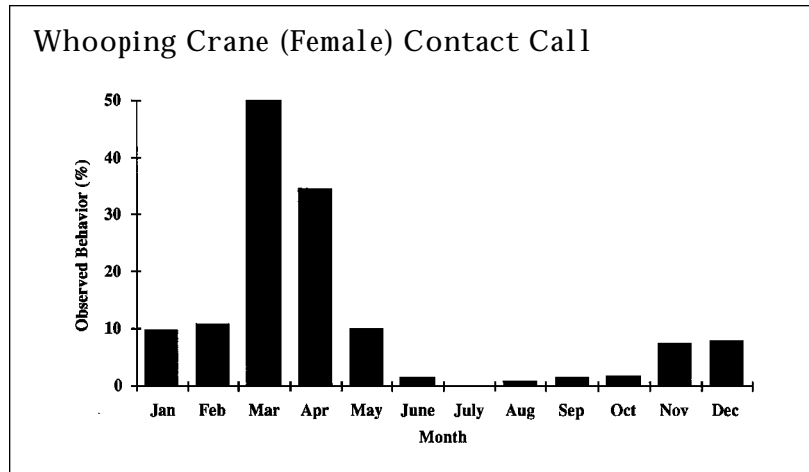


Fig. 6.15. Contact-call performance by month for female Whooping Cranes.

by males and females or singly by either sex. The male trend (not shown) closely paralleled the female trend (Fig. 6.15).

Contact-calling is often accompanied by a highly stereotyped Pre-flight posture (Fig. 6.16) wherein the neck is extended up and far forward. Both ethons had similar performance trends in both species.

We have observed, but not quantified, that the intensity of flight related behavior patterns is directly related to the length of the migration; being greatest in Siberian Cranes, followed by Whooping, Hooded, Red-crowned, White-naped, and Greater Sandhill Cranes. In captivity, the intensity of migratory restlessness seems also to decrease with age.

The Guard-call or Alarm-call is normally given in response to a distant disturbance such as an unfamiliar human or other alarming stimulus animal. In both Mississippi Sandhill and Whooping Crane pairs, the Guard-call is given less often than the Unison call, and seldom during the early part of the breeding season.

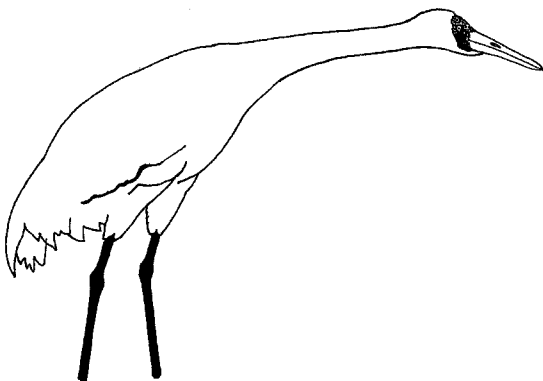


Fig. 6.16. Pre-flight Posture: Siberian Crane. Art Billi Wagner

The average distance between members of a pair is another indicator of the strength of the pair bond, especially during migration and nesting. During the incubation period, however, the non-incubating parent seems to avoid the vicinity of the nest causing a peak in the average distance between mates.

Both sexes of both species of cranes performed the exaggerated, rigid-toed, high-stepping that we call Strut. When Strutting, the body axis is either rotated down and forward (Horizontal Strut) or elevated anteriorly (Vertical Strut, Fig. 6.4). During a Strut performance,

Crown-expansion and presentation also occur. Although both species and both sexes Strut, in the Mississippi Sandhill Cranes, the males normally Horizontal Strut with their mates walking (Unison walk) in tow. Male Whooping Cranes typically Vertical Strut while their mates remain stationary and perform ritualized Preening or some other social display. For the male Mississippi Sandhill Crane, Strutting exhibited a single peak (Fig. 6.17) in midsummer. Strut was without peaks and much less frequently performed in the Whooping Crane.

Crouch is the display that shows the strongest male-female and species differences. For Crouch, a crane flops onto the ground and lies as if brooding young while aggressively billing the surrounding vegetation. Female Mississippi Sandhill Cranes may perform Crouch any time of the year (Fig. 6.18), but a strong peak is evident when they are rearing young. Male Mississippi Sandhill Cranes and either sex of Whooping Cranes seldom perform this display.

Probably all species show some unique seasonal trends. For example, most cranes lay eggs in spring or summer, however, many wild Wattled Cranes lay in winter (Johnson and Barnes 1991) and their captive counterparts lay eggs from fall to spring (Beall 1985). Gray Crowned, Black Crowned, Blue, and Demoiselle Cranes may also lay eggs in indoor winter quarters (K. Kawata, Detroit Zoological Park, Royal Oak, Michigan; R. Lastavica, Omaha Zoo, Omaha, Nebraska; and P. Strasser, National Aviary, Pittsburgh, Pennsylvania, personal communications).

Some performance trends are probably common to most or all cranes. For example, we hypothesize that cranes (captive and wild) vocalize less once the eggs

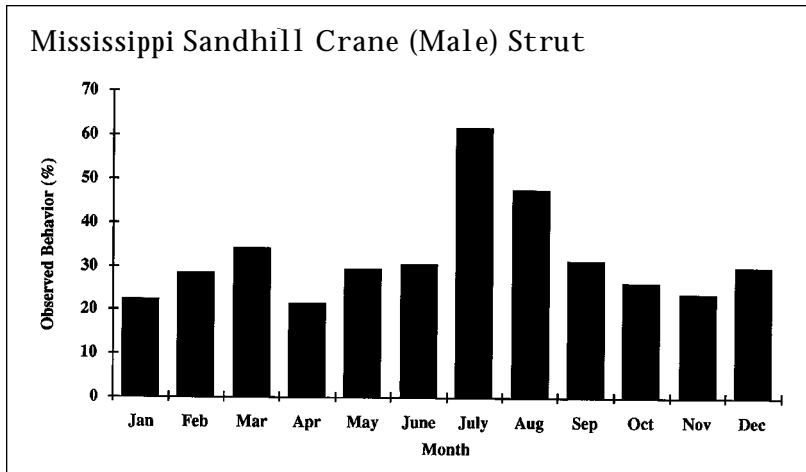


Fig. 6.17. Monthly performance levels in strut for male Mississippi Sandhill Cranes.

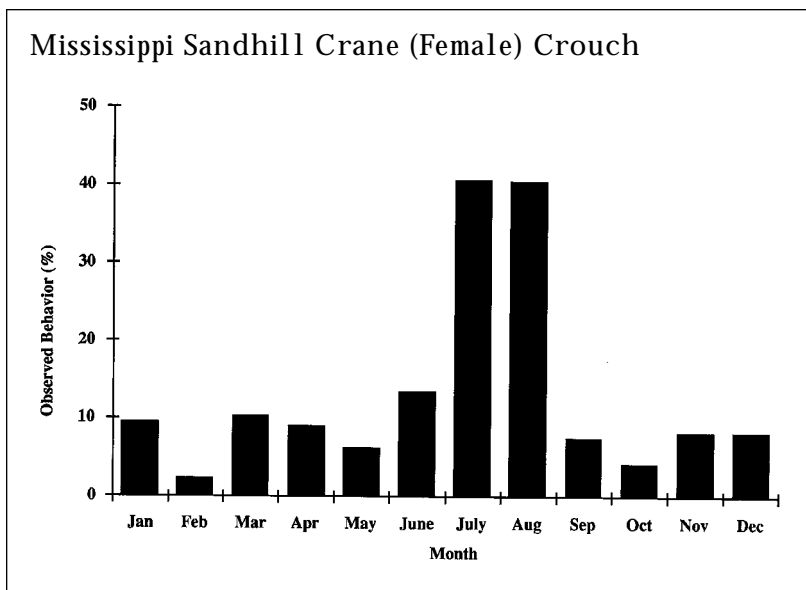


Fig. 6.18. The annual cycle in Crouch performance for female Mississippi Sandhill Cranes.

are laid. Katz (1979) found what appeared to be reduced calling in captive Hooded Cranes as the egg-laying season approached. Probably all species of cranes (captive and wild) begin copulating 2 to 5 weeks before egg laying, even those that are still migrating (Littlefield and Ryder 1968; Walkinshaw 1973a; Littlefield 1985). Captive pairs build nests a few days or weeks before egg laying. By watching for the onset of nest building, a colony manager may know when to begin artificial insemination (AI) to fertilize the first eggs.

Surprisingly, in our walk-through study, we failed to find strong seasonal trends for such activities as

Dancing and Pre-copulatory display for either species, and Strutting was seasonal only in male Mississippi Sandhill Cranes.

All species of cranes feed, brood, and defend their young, and some pairs of most or all species are extremely aggressive when raising chicks. All cranes become more sensitive to disturbance when incubating and rearing chicks. Most cranes show some behavioral changes associated with the molt. Most undergo an annual or biennial simultaneous molt of all flight feathers (Blauuw 1897; see Chapter 7 for physiology of molt and characteristics of juvenal molt). African Crowned Cranes and Brolgas, however, molt continuously; Demoiselle Cranes and most Sandhill Cranes molt sequentially. The molt is physiologically stressful and is accompanied by a decrease in performance values of social displays during the time that the bird is flightless. Such changes are probably adaptations for survival. Because cranes (even normally aggressive individuals) become very shy when molting, minimize human contact during this time. The female may even become the dominant member of the pair while the male is molting. Watch molting cranes carefully to forestall penmate aggression. In group pens, molt can destabilize the dominance hierarchy. Molt can also cause cranes to appear ill.

By knowing behavioral norms for each sex and species, it is possible to promote survival and productivity in captive colonies.

Pairing Cranes

Pairing cranes can begin in the birds' second year. Well-established pairs remain together for many years. When pairing subadult cranes (i.e., 3 years of age), be aware that new pairs are frequently ephemeral. Pairs should not be viewed as permanent until they remain stable for several months and/or reproduce. Wild, subadult Florida Sandhill Cranes usually associate with several potential mates before a firm bond is

established (Nesbitt and Wenner 1987). In choosing potential pairs, be mindful that birds of similar age pair more readily: young cranes are sometimes intimidated by older adults. Potential mates should not be genetically related nor should they have been reared together.

Potential mates should be placed in adjacent pens ideally with a common door to allow herding of a crane from one enclosure to the other without capture. Close contact can be encouraged by placing food and water near the fence dividing the two pens. Pairing pens should be arranged so that concealed caretakers can observe the birds and quickly enter the pens if necessary to separate the birds.

Pairing stages were summarized by Mirande and Archibald (1990) as follows. The first sign of pairing in the wild is one crane following another. In captivity, pairing is evidenced when the birds frequently stand side-by-side. As pairing continues, the behavior of the two birds becomes progressively synchronized. They feed and rest simultaneously. Synchronized displays, such as threats, Guard Calls, and Unison-calls may also indicate pairing; however, such behavior can also indicate intense intrapair aggression or aggression toward caretakers. A crane may interpret its intended mate's aggressive displays as sexual attraction, but when the birds are placed together, the aggressive crane may attack. To prevent injury, keep an over-aggressive male in the pen adjacent to the female; she may thereby be stimulated to lay eggs without risk. For AI pairs with aggressive males, this is sometimes the best strategy, even long term.

Although cranes (even chicks) sometimes **dance** solo or in larger groups, dancing is also associated with pairing and is believed to synchronize mates for successful copulation. Lack of dancing between two cranes can indicate that pairing is not occurring. As a pair bond strengthens, the male generally becomes more defensive of the enclosure than does the female. The ultimate indication of successful pairing is copulation. Pairs that are well bonded should at least attempt to copulate, although some will be unsuccessful because of wing injury, etc.

When it is time to **move the cranes into the same pen** for the first time, move the more dominant bird (usually the male) into the subordinate crane's enclosure. This provides the subordinate bird with a psychological advantage because the pen is its territory. Watch the birds constantly at first and separate them immediately if excessive aggression is observed. Dancing strengthens the developing

pair bonds, especially when the female continues to dance after the male begins to run and flap in mock Rushes. In unpaired birds or unstable pairs, dancing can intimidate the subordinate crane. If one crane keeps dancing while the second crane flees, the first crane may chase the fleeing crane and attack. Death or serious injury can result if the cranes are not immediately separated. A single attack can negate weeks of progress in the pairing process.

Cranes can be further manipulated to promote pairing by brailing one wing of the dominant bird before placing it in the enclosure of the submissive crane. The brail stresses the crane, reducing its aggression. Dominance in cranes is related to height; more dominant birds are generally taller than submissive ones. However, the dominance of a submissive crane can be increased by providing a **mound of earth** 0.3 to 0.5 m high near the fence separating the cranes. Displaying cranes will often stand on such mounds. By this simple manipulation, the dominance of a subordinate crane may be increased.

If cranes seem compatible, they can be left together during the day with hourly checks. When Unison-calls and dancing do not lead to aggression or intimidation, the pair is considered to be solidly mated and can be trusted to occupy the same enclosure at all times.

For cranes, some action patterns are "contagious." For example, if one bird yawns or flaps its wings there is a good chance that one or more penmates will do likewise. If one crane is extremely aggressive towards humans, its penmates will often become aggressive. By pairing cranes with different behavioral traits, characteristics of one crane can be encouraged in the other. Parent-reared cranes become much tamer and adapt to captivity better if penned or paired with hand-reared birds. Conversely, hand-reared birds can become less attached to humans if integrated with parent-reared mates.

Some paired cranes that have not bred can be induced to become more confident and better bonded if a crane chick is placed in an adjacent pen. Sometimes the pair attempts to adopt the chick (as evidenced by their passing food items through the wire to the chick, brood calling, and by their lack of threat displays); others will try to kill the chick or ignore it.

Stress and Disturbance

Crane pairs are healthier and breed better when disturbance is minimized (Mirande et al. 1988 unpubl.). Pairs are normally less stressed when: (1) their pens have visual barriers separating them from neighboring cranes; (2) routine tasks are done on a regular schedule; (3) the same people perform these tasks; and (4) the breeding area is closed to certain kinds of vehicles (aircraft and large trucks) and activities (pen repairs and construction) during, and three months prior to, the breeding season.

Some cranes breed well without **visual barriers** between neighboring pairs, but pairs should at least be separated by empty pens to prevent fighting through the fence. Sandhill, White-naped, Red-crowned, and some other species have bred well without visual barriers, but Whooping, Sarus, and Siberian Cranes need them.

Cranes breed best when they have a large, secure **territory**. Breeding pairs need at least 100 m², but 300 m² is preferable. They often benefit from a shelter in which to retire from view or gain protection during inclement weather. Trees or bushes in the pen may provide **natural cover**. Some pairs breed better if they have a secluded spot for nesting. Satisfy their need to build nests by providing dry twigs or coarse grasses (fine or moist vegetation will mold more rapidly).

If AI is intended, line **capture corners** with nonabrasive cloth such as tennis netting for 3-6 m in each direction from the corner. At ICF, discarded 2 m tall conifers (old Christmas trees) are used to line these corners.

Stress can be reduced by taming cranes. The process involves conditioning birds to human activity through providing treats (favorite foods), avoiding direct eye contact, announcing your approach by calling when still far away, and other techniques as discussed in Chapter 5.

Pair Bonds

Well paired cranes perform synchronous activities and stay near one another most of the time. If a male is excessively dominant or if the female is dominant over the male, the pair may never breed (Derrickson and Carpenter 1987). Several circumstances can result in weak pair bonds. If one member of a pair is excessively submissive to the other or if one mate prefers a neighboring crane, the pair bond can be weakened. An

unstable pair bond can result in one crane injuring or killing its mate.

Some pairs are compatible but never lay eggs; this occasionally occurs with birds that were paired when very young. Such birds seem to view each other as siblings. For these pairs, pair-related displays and territorial defense are less intense.

The "**Location-call test**" is a good means of testing the strength of a pair bond. This requires that the male be removed from the female by at least 100 m, but within earshot. If both cranes perform the loud, single-note Location-call, and promptly answer the calls of their mate from a distance, the pair bond is probably genuine. If either member of the pair fails to Location-call or fails to respond to the other crane's Location-calls, the pair bond is probably weak and the pair should be dissolved.

If a closely bonded pair is to be divided and new pairs formed, Location-calling can seriously delay or prevent the re-pairing process. It is wise to postpone introducing the new intended mate until a week or more following separation of the old pair. Former mates must sometimes be separated by a great distance (1 km or more) to facilitate pairing them with other cranes.

Sometimes **wild cranes harass captive pairs** which can result in captive males redirecting aggression toward their mates. In these situations, pairs may need to be separated until the wild cranes leave. The pair can generally be safely reunited after a few days.

Wild cranes occasionally **switch mates** even though neither member of the pair has died (Littlefield 1981; Nesbitt and Wenner 1987). Young cranes frequently form ephemeral pairs in the wild and may take years to form permanent pairs (Bishop 1984; Nesbitt and Wenner 1987). Pairs that produce offspring are much more likely to persist (Nesbitt and Wenner 1987). In wild Florida Sandhill Cranes, re-pairing efforts vary by sex: males quickly find new mates, while females may take several years to re-pair (Nesbitt 1989).

Hand-reared cranes that are overly attached to humans can often be made to breed if they are given a suitable mate, and if thereafter they have minimal contact with humans. Once the pair has eggs, its pair bond is often strengthened, and further bonds to humans are weakened by the pair's mutual defense of the eggs or chicks.

Before including birds in an AI program, allow young pairs to attempt copulation for one or two breeding seasons. Flightless males that are unable to fertilize eggs due to unilateral wing impairment may

be clipped on the whole wing to improve their wing symmetry and thus help them balance. Allowing the pair to raise a chick may also synchronize their reproductive cycles or strengthen their pair bond, thereby increasing the chance of fertility in the future.

Parent-reared cranes may be more likely to copulate than hand-reared ones, and cranes hand-reared in groups may be more likely to copulate than birds that were hand-reared alone (Derrickson and Carpenter 1987). However, most hand-reared cranes that are socialized with others as colts learn to copulate when they become adults.

If a pair does not produce fertile eggs after one or two years of management as described above, it may be necessary to re-pair them or initiate AI (Chapter 11A). In **using AI**, it is important to disturb the birds as little as possible. Some cranes will not lay eggs when they are regularly handled for AI. One strategy for such cranes is to wait until the female starts laying eggs, and then initiate AI. Normally, the female is not so stressed by this handling that she fails to lay more eggs. This strategy is less useful with Wattled Cranes, which frequently lay one-egg clutches. For those Wattled Cranes that are adverse to AI, one insemination 4-10 days before the next egg is expected can often produce a fertile egg (Monica Tuite, unpubl. data). The best AI schedule depends on the particular female's laying history (see Chapter 3). Inseminating a nervous female a few days before she is scheduled to lay her second and subsequent clutches, **but not repeatedly between each clutch**, can improve the chances of getting several fertile eggs while **minimizing disturbance**. Finally, be sure that egg searches and other visits to the pens of shy cranes are performed quickly and, if possible, use binoculars to scan the pen from a distance.

Behavioral Management of Cranes for Display

Cranes on public display normally receive more disturbance than other captive cranes. Because many cranes will not breed while on display, it is best to exhibit only those birds that are of low genetic value. Display cranes should tolerate human visitors but not be aggressive towards them. Extremely aggressive cranes are dangerous to caretakers and the public, and may damage themselves in their attempts to attack

people. To reduce the chance of injury to birds and visitors, design display pens so that the public cannot come closer than 1 m to the cranes.

Cranes on display adopt a daily schedule timed to periods of human visitation. They direct many of their social displays toward the public, and most remain within public view.

Several management practices can encourage exhibit cranes to breed. Caretaker entrances should allow servicing so that a portion of the pen is left undisturbed by keepers and the public. Performing some activities out of sight and with minimal contact encourages breeding. Cranes on display feel safer when they have a "sanctum" where they can go out of sight of humans. This hiding place can be an indoor shelter, a sheltered corner, or a patch of dense foliage ca 1.5 m tall.

Novel pen designs can also improve the display value of cranes. Elevated overlooks or moats allow people to view cranes unobstructed. These designs, however, require that the cranes be flightless.

Mixed species exhibits are attractive, but because cranes are solitary nesters, they are unlikely to breed when there are more than two cranes in one display. The dominant pair will too often defend most of the pen, driving the remaining cranes from its territory. Such pairs may even breed. However, dangerous encounters are likely whenever a breeding pair is penned with conspecifics. The only situation in which a breeding pair can coexist peacefully with other conspecifics is when the pen is very large. Patuxent maintains three pairs of Florida Sandhill Cranes in a 2 ha enclosure. The dominant pair normally defends more than half of the area while leaving enough room for the two subordinate pairs to escape and breed.

Behavioral Management of Cranes for Release

This section summarizes the management of cranes destined for release. For more details see Chapters 5 and 11D.

Parent-reared and hand-reared cranes for release are managed very differently. Parent-reared chicks develop normally and require no special training. Chicks reared by hand, in isolation from human contact, should be allowed to see and hear conspecific adults so that they learn to socialize and breed

with conspecifics. Normally, hand-reared chicks are handled by costumed human caretakers (costume-rearing).

Costume-reared chicks can be taken to the release site when they are as young as 10 weeks (Horwich 1986). Young cranes exhibit the behavioral plasticity important in rapidly learning new survival techniques. Horwich (1986, 1989) released his costume-reared cranes in early fall to coincide with the period when wild cranes are the most gregarious. Other studies also suggest that captive-reared cranes integrate better with wild cranes if released in early fall (Mitchell and Zwank 1987). Whatever the rearing method, cranes should be released gently over a period of weeks (**gentle-release**) to give them time to acclimate (Horwich 1986; Mitchell and Zwank 1987). If the chicks are costume-reared, this acclimation period can also be used to introduce them to natural foods.

Release pens should be large enough to allow cranes to move comfortably away from mammalian predators outside their enclosure. Pens used in successful releases of Mississippi Sandhill Cranes have been 0.5 to 2 ha (1-5 acres) in size.

Imprinting Attachment, and Behavioral Development in Cranes

contributed by Robert H. Horwich

There have been few studies of crane imprinting or early development (Voss 1976; Layne 1981). Most imprinting research in the 1960's focused on the short-term effects of imprinting on social preferences. Domestic fowl (*Gallus domesticus*) and domestic ducks (*Anas platyrhynchos*) rapidly restrict their filial attachment and following response to their parent, human caretaker, or to other stimuli encountered shortly after hatching. This learning phenomenon has been called **filial imprinting** (Bateson 1978). Many studies (Hess 1973; Hess and Petrovich 1977) show that there is a "critical" period when precocial birds imprint on a parental model.

An accumulating body of evidence indicates that relatively **early experiences** have profound effects on sexual choice later in life (Immelmann 1972; Bateson 1978). This evidence indicates that exposure to social

stimuli at certain age periods can reverse the preference of early filial imprinting (Gallagher 1976, 1977; Vidal 1976, 1980).

Filial Imprinting and Parental Care

Cranes exhibit imprinting patterns similar to domestic fowl. Imprinting probably begins in the egg about 2 days prior to hatching, when chicks begin answering the parents' brood calls. Chicks follow the adult on the first day and are often away from the nest by day 1 or 2 (Walkinshaw 1973b). Parental attachment is complete within the first 3 days and becomes stronger during the first 2 weeks.

Attachment is reinforced by a radical change in parental behavior at hatching. This includes increased brood calling, brooding, preening the chick, preening the adult's brood patch, and feeding the chick. This behavior encourages imprinting and the development of a following response by the chick during the first week, the initial sensitive period of development (Hartup and Horwich 1994).

Brooding of the chick occurred only during the first week in our study of Sandhill Cranes. Preening the chick, although rarely seen, was done by the female while brooding. Wild cranes may brood chicks that are up to six weeks old (G. W. Archibald, ICF, personal communication). The female invites the chick to brood by extending the wrist joint laterally while calling loudly and pointing her bill into or preening the opened cavity. The moving bill tip is very attractive to crane chicks, and probably induces the chick to accept brooding. Pecking the parent's bill, the chick's greeting, is a ritualized feeding behavior. It was elicited in puppet-reared chicks by extending the puppet's bill toward the chick (Fig. 6.19). A stereotyped bill presentation by White-naped Cranes also elicited the bill peck. A similar feeding posture occurs in wild Sandhill Cranes (Layne 1981).

Sexual Imprinting

Sexual imprinting is a form of learning which shares many characteristics with filial imprinting, but which also influences mate choice at sexual maturity. There are many instances of birds sexually imprinting on humans or other bird species (Immelmann 1972), but studies have shown that the process is reversible if cross-fostered or hand-reared birds are introduced to their own species during or before the end of the sensitive period.



Fig. 6.19. *Siberian Crane chick greeting puppet head.*

Photo David H. Ellis

Vidal (1976, 1980) neatly delineated two imprinting periods in chickens. He noted an early sensitive period for learning the following response and a second sexual imprinting period at 30–45 days. Cockerels exposed to a model at this later period became sexually imprinted on it despite their earlier training to follow a different model.

Proper sexual imprinting is critical in crane reintroduction programs. Although the rearing of Whooping Cranes by wild Sandhill Crane parents has produced a small wild population of Whooping Cranes in the Intermountain West, these cranes are not breeding (Ellis et al. 1992a). **Cross-fostering** is believed to have resulted in imprinting problems preventing the Whooping Cranes from breeding with their own species. The recent discovery of a Whooping-Sandhill Crane hybrid at Bosque del Apache National Wildlife Refuge (Pratt 1993) and unusual behavior by cross-fostered females (Mahan 1992; Swengel, personal observation) confirm this.

Imprinting Stimuli

Newly hatched precocial birds can be imprinted on a wide variety of objects in the absence of their natural parents, indicating that early parental recognition is largely acquired (Lorenz 1937, 1970; Ramsay 1951;

Spalding in Jaynes 1956). However, there are some innate preferences (Hinde 1961; Gaioni et al. 1978). Initially, vocal cues seem more important than visual ones (Ramsay 1951; Gottlieb 1971), and there may be other innate preferences for certain colors and forms (Jaynes 1956; Schaefer and Hess 1959; Salzen and Meyer 1968).

As part of our reintroduction study (Horwich 1986, 1989; Horwich et al. 1992), we reared crane chicks with a stuffed crane model that emitted brooding calls, fed chicks using a crane-head puppet (Fig. 11D.2), and led them while costumed and using the same puppet emitting the same calls (Fig. 11D.1). Although the main goal of costume-rearing (see Chapter 11D) was to imprint crane chicks on a crane-like substitute, we also hoped that use of the costume would allow us to control the birds after release while leaving them still fearful of uncostumed humans. The costume, although not overly crane-like, broke up the human gestalt by de-emphasizing the head, face, and hands while emphasizing the crane head and voice. Although the chicks reared with the costume did not show affinity to humans, they did not exhibit much fear either. Before release, an uncostumed person could approach within 3 m of the mildly wary chicks, but after associating with wild cranes for 2 weeks, the chicks' flight distance in response to human approach had increased to 100 m (Horwich et al. 1992). Fear of humans can, of course, be taught (see Human Avoidance Conditioning in Chapters 5 and 11D).

At 4–8 weeks of age, our chicks were given choices of stimuli in an attempt to assess the early effects of filial imprinting (Horwich and Owen unpubl.). For all chicks, the most important stimulus was the moving puppet head. They responded quickly by pecking the bill. It was clearly chosen over a mounted body or a vocalizing speaker.

The moving bill tip directs chicks of all ages in feeding (Hartup and Horwich 1994). When feeding a chick, White-naped Cranes sometimes drop and pick up an insect as many as 15 times before the chick will accept it. This motion was very attractive to chicks, who eventually picked up insects on their own. The parental bill attracts the chick, and the chick greets the parent by purring and pecking the extended bill. Later, juveniles watch the parents' bills probing the ground, and probe the same area. Bill movement is also very attractive in other precocial birds (Tinbergen and Perdeck 1950; Hailman 1967; Johnson and Horn 1988).

We tested the chicks' responses to various parts of the crane puppet head during their first few weeks. None of the main puppet features (red patch, head, or eye) was consistently important to the chicks. Chicks exposed to a mounted body for only a short period tended to choose the puppet, while those exposed to the body for a longer period of time tended to choose the body. This observation follows the general rule that the longer the exposure, the stronger the preference (Bateson 1978). Sound is another very important stimulus for precocial birds (Gottlieb 1971). Our results indicate that crane chicks are most responsive to brood calls during the first 3 weeks.

These experiments provided information for use in captive rearing. Although red is often used for feeding dishes or for rods dangling in the food bowl to induce feeding in young chicks (Kepler 1978), the red patch of the puppet head did not interest the chicks. The red patch is used in aggressive displays in Sandhill Cranes (Voss 1976). However, when combative Sandhill Crane chicks were separated by the puppet head, they redirected their attacks at the red patch of the puppet (Erickson et al. 1988).

By dangling a puppet-like head in the food dish (Fig. 11D.2), we taught chicks to feed themselves (Horwich 1986; Erickson et al. 1988). By pecking repeatedly at the moving beak tip, they eventually pecked the food below it. This gradually changed to ritualized pecking of the beak tip before feeding until, finally, they pecked only the food. At Patuxent, a taxidermic mount of a crane head (suspended from the ceiling with its bill contacting the food and manipulated from outside the pen; Fig. 5.10) proved effective in teaching Whooping and Sandhill Crane chicks to eat (Ellis et al. 1992b).

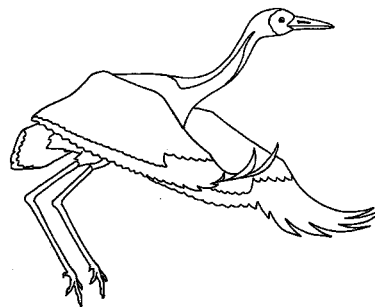
gradually entered a more independent foraging phase at 4-8 weeks of age. At fledging (11-14 weeks), they reattached to the costumed parent, stayed near it much of the time, and pecked its feathers. The intense sociality exhibited during this reattachment period induced the costume-reared chicks to rapidly join wild cranes following release (Horwich et al. 1992). This period seems equivalent to the sexual imprinting period (when the initial attachment can be reversed) in chickens, as identified by Vidal (1976) at 4-6 weeks when the adult plumage was largely complete.

Periodic regressions may be in synchrony with seasonal activities, as seen in mammals (Horwich et al. 1977; Horwich et al. 1982) and cranes (Horwich 1987; Horwich et al. 1992). The initial close bond of parent and chick during the first month protects the chick when it is most vulnerable and needs parental feeding. As the chick grows stronger and can feed itself, it begins a period of independent foraging, during which it follows its parents at a greater distance. The chicks regress by increasing contact with the parents at fledging time when they would otherwise be most likely to become lost if they fly far from their parents (Horwich 1987). This renewed bonding may also involve species and sexual identification. They exhibit a second reattachment period just before and during migration (Horwich 1987; Horwich et al. 1992). Many other bird species, both migratory and non-migratory, as well as mammals, show this same cyclic gregariousness (Nievergelt 1974; Guinness et al. 1979). Besides functioning to keep cranes on the correct migration route, this reattachment or gregariousness may allow orphaned chicks to learn the route from flock mates in the absence of their parents.

Behavioral Cycles and Reattachment Periods

Quantitative studies of bird and mammal behavior have shown that parent-young attachment and many other activities follow a cyclic pattern, with two or more cycles occurring in young animals before fledging or weaning (Horwich 1974, 1987; Ellis 1979). This has been termed a **regression** or **reattachment period** (Horwich 1974). This concept is fundamental to understanding ontogeny and sociality in mammals (Horwich et al. 1982) and birds (Ellis 1979).

After the initial attachment period, there follows a period of gradual independence from the parent. After spending 60% of their time next to a surrogate parent during the first 2 weeks, Sandhill Crane chicks



Literature Cited

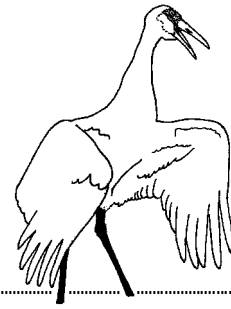
- Allen, R. P. 1952. The Whooping Crane. Research report No. 3. National Audubon Society, New York. 246 pp.
- Archibald, G. W. 1974a. Misty mornings in crane country. *Animal Kingdom* 77(2):19-24.
- Archibald, G. W. 1974b. Methods for breeding and rearing cranes in captivity. *International Zoo Yearbook* 14:147-155.
- Archibald, G. W. 1976a. Crane taxonomy as revealed by the unison call. Pages 225-251 in J. C. Lewis, editor. *Proceedings International Crane Workshop*. Oklahoma State University Printing Service, Stillwater.
- Archibald, G. W. 1976b. The unison call of cranes as a useful taxonomic tool. Ph.D. dissertation, Cornell University, Ithaca, New York. 167 pp.
- Archibald, G. W., and D. L. Viess. 1979. Crane propagation at the International Crane Foundation 1973-78. Pages 51-73 in J. C. Lewis, editor. *Proceedings 1978 Crane Workshop*. Colorado State University Printing Service, Fort Collins.
- Bateson, P. P. G. 1978. Early experience and sexual preferences. Pages 29-53 in J. B. Hutchison, editor. *Biological determinants of sexual behavior*. John Wiley & Sons, New York.
- Beall, F. 1985. *Regional (North American) Wattled Crane studbook, *Bugeranus carunculatus**. Baltimore Zoological Gardens, Md. 32 pp.
- Belتمان, R., and C. E. King. 1993. Captive management and husbandry of Red-crowned Cranes in Europe. *International Zoo News* 40(1):6-20.
- Bishop, M. A. 1984. Dynamics of subadult flocks of Whooping Cranes wintering in Texas, 1978-79 through 1982-83. M.S. thesis, Texas A & M University, College Station. 127 pp.
- Blauuw, F. E. 1897. A monograph of the cranes. E. F. Brill, Leiden, Netherlands. 64 pp.
- Derrickson, S. R., and J. W. Carpenter. 1980. Ethology and management of captive Sandhill Cranes. Pages 76-81 in *Annual Proceedings of the American Association of Zoo Veterinarians*.
- Derrickson, S. R., and J. W. Carpenter. 1987. Behavioral management of captive cranes—factors influencing propagation and reintroduction. Pages 493-511 in G. W. Archibald and R. F. Pasquier, editors. *Proceedings of the 1983 International Crane Workshop*. International Crane Foundation, Baraboo, Wis.
- Ellis, D. H. 1979. Development of behavior in the Golden Eagle. *Wildlife Monograph* No. 70. 94 pp.
- Ellis, D. H., G. W. Archibald, S. R. Swengel, and C. B. Kepler. 1991. Compendium of crane behavior. Part 1: individual (nonsocial). Pages 225-234 in J. T. Harris, editor. *Proceedings of the 1987 International Crane Workshop*. International Crane Foundation, Baraboo, Wis.
- Ellis, D. H., J. C. Lewis, G. F. Gee, and D. G. Smith. 1992a. Population recovery of the Whooping Crane with emphasis on reintroduction efforts: past and future. *Proceedings North American Crane Workshop* 6:142-150.
- Ellis, D. H., G. H. Olsen, G. F. Gee, J. M. Nicolich, K. E. O'Malley, M. Nagendran, S. G. Hereford, P. Range, W. T. Harper, R. P. Ingram, and D. G. Smith. 1992b. Techniques for rearing and releasing nonmigratory cranes: lessons from Mississippi Sandhill Crane program. *Proceedings North American Crane Workshop* 6:135-141.
- Ellis, D. H., S. R. Swengel, G. W. Archibald, and C. B. Kepler. *In prep.* A sociogram for the cranes of the world.
- Erickson, D., F. Boll, and R. Horwich. 1988. Raising crane. Videocassette. Ootek Production, Sauk City, Wis.
- Gaioni, C. C., H. S. Hoffman, and P. DePaulo. 1978. Imprinting in older ducklings: some tests of a reinforcement model. *Animal Learning and Behavior* 6:19-26.
- Gallagher, J. E. 1976. Sexual imprinting: the long term effects of various regimens of social experience on mate preference in Japanese Quail *Coturnix coturnix japonica*. *Behavior* 57:91-115.
- Gallagher, J. E. 1977. Sexual imprinting: a sensitive period in Japanese Quail (*Coturnix coturnix japonica*). *Journal of Comparative and Physiological Psychology* 91:72-78.
- Gottlieb, G. 1971. *Development of species identification in birds*. University of Chicago Press, Chicago, Ill. 176 pp.
- Guinness, F. E., M. J. Hall, and R. A. Cockerill. 1979. Mother-offspring association in red deer (*Cervus elaphus* L.) on Rhum. *Animal Behavior* 27:136-144.
- Hailman, J. P. 1967. The ontogeny of an instinct: the pecking response in chicks of the Laughing Gull (*Larus atricilla* L.). *Behavior Supplement* 15:1-159.
- Hartup, B. K., and R. H. Horwich. 1994. Early prenatal care and chick development in a cross-fostering trial with White-naped (*Grus vipio*) and Greater Sandhill (*Grus canadensis tabida*) Cranes. *Bird Behavior* 10:21-27.
- Hess, E. H. 1973. *Imprinting*. Van Nostrand Reinhold, New York. 472 pp.
- Hess, E. H., and S. B. Petrovich, editors. 1977. *Imprinting*. Dowden, Hutchinson and Ross, Stroudsburg, Pa.
- Hinde, R. A. 1961. The establishment of the parent-offspring relation in birds, with some mammalian analogies. Pages 175-19 in W. H. Thorpe and O. L. Zangwill, editors. *Current problems in animal behavior*. Cambridge University Press, Cambridge, England.
- Horwich, R. H. 1974. Regressive periods in primate behavioral development with reference to other mammals. *Primates* 15:141-149.
- Horwich, R. H. 1986. Reintroduction of Sandhill Cranes to the wild. *ICF Bugle* 12:1, 4-5.
- Horwich, R. H. 1987. Behavioral development in the Red-crowned Crane (*Grus japonensis*). *Zoo Biology* 6:379-389.
- Horwich, R. H. 1989. The use of surrogate parental models and age periods in a successful release of hand-reared Sandhill Cranes into the wild. *Zoo Biology* 8:379-390.
- Horwich, R. H., S. J. H. Cogswell, J. Burrows, and N. Mitchell. 1982. Seasonal variation in mother-daughter groupings in Siberian ibex (*Capra ibex siberica*). *Zoo Biology* 1:345-354.
- Horwich, R. H., and C. R. Owen. *Unpublished*. Imprinting stimuli for surrogate-reared Sandhill Crane chicks (*Grus canadensis*).

- Horwich, R. H., R. Van Dyke, and S. J. H. Cogswell. 1977. Regressive growth periods as a mechanism for herd formation in Siberian ibex (*Capra ibex*). *Zoologische Garten* 47:59-68.
- Horwich, R. H., J. Wood, and R. Anderson. 1992. Release of Sandhill Crane chicks hand-reared with artificial stimuli. Pages 255-261 in D. Wood, editor. Proceedings 1988 North American Crane Workshop. Florida Nongame Wildlife Program Technical Report #12.
- Immelmann, K. 1972. Sexual and other long-term aspects of imprinting in birds and other species. Pages 147-174 in D. S. Lehrman, R. A. Hinde, and E. Shaw, editors. *Advances in the study of behavior*. Vol. 4. Academic Press, New York.
- Jaynes, J. 1956. Imprinting: the interaction of learned and innate behavior. 1. Development and generalization. *Journal of Comparative and Physiological Psychology* 49(3):201-206.
- Johnson, D. N., and P. R. Barnes. 1991. The breeding biology of the Wattled Crane in Natal. Pages 377-386 in J. T. Harris, editor. Proceedings of the 1987 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Johnson, M. H., and G. Horn. Development of filial preferences in dark-reared chicks. *Animal Behavior* 36:675-683.
- Katz, B. 1979. Breeding ethology of the Hooded Crane. Pages 217-222 in J. C. Lewis, editor. Proceedings 1978 Crane Workshop. Colorado State University Printing Service, Fort Collins, Colo.
- Kepler, C. B. 1976. Dominance and dominance-related behavior in the Whooping Crane. Pages 177-196 in J. C. Lewis, editor. Proceedings International Crane Workshop. Oklahoma State University Publishing and Printing, Stillwater.
- Kepler, C. B. 1978. Captive propagation of Whooping Cranes: a behavioral approach. Pages 231-241 in S. A. Temple, editor. *Endangered birds: management techniques for preserving threatened species*. University of Wisconsin Press, Madison. 466 pp.
- Layne, J. N. 1981. Nesting, development of the young, and parental behavior of a pair of Florida Sandhill Cranes. *Florida Field Naturalist* 9:51-59.
- Littlefield, C. D. 1981. Mate-swapping of Sandhill Cranes. *Journal of Field Ornithology* 52:244-245.
- Littlefield, C. D. 1985. A record of preterritorial copulation by Sandhill Cranes. *Western Birds* 16:151.
- Littlefield, C. D., and R. A. Ryder. 1968. Breeding biology of the Greater Sandhill Crane on Malheur National Wildlife Refuge, Oregon. Pages 444-454 in Transactions of the 33rd North American Wildlife and natural resources conference. Wildlife Management Institute, Washington, D.C.
- Lorenz, K. Z. 1937. The companion in the bird's world. *Auk* 54:245-273.
- Lorenz, K. Z. 1970. *Studies in animal and human behavior*. Vol. 1. Translated by R. Martin. Harvard University Press, Cambridge, Mass. 403 pp.
- Mahan, T. A., and B. S. Simmers. 1992. Social preference of four cross-foster reared Sandhill Cranes. Proceedings North American Crane Workshop 6:114-119.
- Masatomi, H., and T. Kitagawa. 1975. Bionomics and sociology of Tancho or the Japanese Crane, *Grus japonensis*, II. Ethogram. *Journal of the Faculty of Science, Hokkaido University, Series VI, Zoology* 19(4):834-878.
- Mirande, C. M., and G. W. Archibald. 1990. Sexual maturity and pair formation of captive cranes at the International Crane Foundation. Pages 216-225 in Proceedings American Association of Zoological Parks and Aquariums 1990 Annual Conference.
- Mirande, C. M., J. W. Carpenter, and A. Burke. 1988 unpubl. The effect of disturbance on the reproduction and management of captive cranes. Paper presented at the 1988 North American Crane Workshop. 18 pp.
- Mitchell, L. C., and P. J. Zwank. 1987. Comparison of release methods for parent reared Mississippi Sandhill Cranes. Pages 399-409 in G. W. Archibald and R. F. Pasquier, editors. Proceedings of the 1983 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Nesbitt, S. A. 1989. The significance of mate loss in Florida Sandhill Cranes. *Wilson Bulletin* 101:648-651.
- Nesbitt, S. A., and G. W. Archibald. 1981. The agonistic repertoire of Sandhill Cranes. *Wilson Bulletin* 93:99-103.
- Nesbitt, S. A., and A. S. Wenner. 1987. Pair formation and mate fidelity in Sandhill Cranes. Pages 117-122 in J. C. Lewis, editor. Proceedings 1985 Crane Workshop. Platte River Whooping Crane Habitat Maintenance Trust and U.S. Fish and Wildlife Service, Grand Island, Nebr.
- Nievergelt, B. 1974. A comparison of rutting behavior and grouping in the Ethiopian and alpine ibex. Pages 324-340 in V. C. Geist and F. Walther, editors. *The behavior of ungulates and its relation to management*. International Union for the Conservation of Nature and Natural Resources, Morges, Switzerland.
- Poulsen, H. 1975. Agonistic behavior of two species of cranes. *Dansk Ornithologisk Forenings Tidsskrift* 69:119-122.
- Pratt, J. J. 1993. The Whooper-Sandhill Crane hybrid. *Grus Americana* 32(1):1.
- Ramsay, A. O. 1951. Familial recognition in domestic birds. *Auk* 68:1-15.
- Salzen, E. A., and C. C. Meyer. 1968. Reversibility of imprinting. *Journal of Comparative and Physiological Psychology* 66:269-275.
- Schaefer, H. H., and E. H. Hess. 1959. Color preferences in imprinting objects. *Zeitschrift fur Tierpsychologie* 16:161-172.
- Swengel, S. 1992. Sexual size dimorphism and size indices of six species of captive cranes at the International Crane Foundation. Proceedings North American Crane Workshop 6:151-158.
- Tacha, T. C. 1981. Behavior and taxonomy of Sandhill Cranes from mid-continental North America. Ph.D. dissertation, Oklahoma State University, Stillwater. 112 pp.
- Tinbergen, N., and A. C. Perdeck. 1950. On the stimulus situation releasing the begging response in the newly hatched Herring Gull chick (*Larus argentatus* Pont.). *Behavior* 3:1-39.
- Vidal, J. M. 1976. L'empreinte chez les animaux. *La Recherche* 63:24-35.

- Vidal, J. M. 1980. The relations between filial and sexual imprinting in the Domestic Fowl: effects of age and social experience. *Animal Behavior* 28:880-891.
- Voss, K. S. 1976. Ontogeny of behavior of the Greater Sandhill Crane. Pages 252-262 *in* J. C. Lewis, editor. *Proceedings International Crane Workshop*. Oklahoma State University Publishing and Printing, Stillwater.
- Voss, K. S. 1977. Agonistic behavior of the Greater Sandhill Crane. Pages 63-85 *in* R. D. Feldt, editor. *Eastern Greater Sandhill Crane Symposium*, October 24-26, 1977, Michigan City, Indiana.
- Walkinshaw, L. H. 1973a. *Cranes of the world*. Winchester Press, New York. 370 pp.
- Walkinshaw, L. H. 1973b. A history of Sandhill Cranes on the Haehnle Sanctuary, Michigan. *Jack-Pine Warbler* 51:54-74.



Behavior Management



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Management of crane behavior varies with use, age, and reproductive condition. We discuss five management classes: chicks, subadults, breeding birds, cranes for public display, and release birds. Cranes are individuals, and management practices that are beneficial to one crane may be harmful to another. Because management of these classes and individuals overlap, a combination of behavioral management techniques often works best for individual cranes.

This chapter emphasizes crane **social behavior** and how it affects captive management. Ellis et al. (1991) described the non-social (maintenance) behavior of all species. Because most body maintenance behavior patterns are superficially alike for both calm and highly stressed cranes, we discuss below only the few maintenance actions that suggest a crane is stressed. Studies of crane social behavior that include illustrations of displays are Allen (1952) on the Whooping Crane; Archibald (1974a) and Katz (1979) on the Hooded Crane; Masatomi and Kitagawa (1975) on the Red-crowned Crane; Poulsen (1975) on Siberian and Common Cranes; Voss (1976, 1977), Nesbitt and Archibald (1981), and Tacha (1981) on the Sandhill Crane; and Ellis et al. (*In prep.*) on all species. Previous papers describing behavioral management of captive cranes include Archibald (1974b), Kepler (1976, 1978), Archibald and Viess (1979), and Derrickson and Carpenter (1980, 1987).

Techniques in Behavior Observations

Observing cranes in the same context and at the same time each day reveals seasonal changes in social patterns and abnormalities suggesting health problems. Data collection can be as simple as merely noting abnormalities or social problems, or as complex as a bird-by-bird tabulation of behavior details on form

sheets. **Daily observations** are essential in monitoring the pairing and social interaction of cranes in the same pen or adjacent pens.

Blinds (Fig. 6.1) set up near cranes allow for longer-term observations of relatively undisturbed (by humans) behavior. Cranes behave more normally when people are not in view, so blinds are a valuable supplement to daily observations not just for research, but also in the management of pairs. Because some cranes remain disturbed by observers in nearby blinds, it is crucial to locate blinds with care and use one-way glass if necessary to enable the observer to be invisible to the crane.



Fig. 6.1. Brian Clauss enters an elevated observation blind.

Photo David H. Ellis

Remote monitoring (Fig. 6.2) via closed-circuit TV (CCTV) is valuable for observing pairing, breeding, or egg laying of cranes without risking them noticing the observer. Cranes seem to adjust quickly to cameras mounted high in a pen corner (although they notice cameras that move or make noise). CCTV allows for the most undisturbed watching of cranes and is ideal for making videotapes that can be replayed at high speed and searched for significant behavioral patterns, such as copulation and nest building. For cranes that break eggs, long-term egg vigils are made easier by CCTV. CCTV allows for a second look to determine if first impressions can be confirmed by review of videotapes.



Fig. 6.2. *Closed circuit television camera monitors Whooping Crane.*

Photo David H. Ellis

Aggressive Displays

Cranes that have red caps or bare skin on their heads expand the cap (**Crown-expansion**), extend their wattles (**Bare-skin-expansion**), or suffuse the bare skin with brighter color to supplement their displays (Fig. 6.3). Cranes with completely feathered heads elevate head plumage to increase the apparent size of their heads. These head displays are low-intensity displays when they appear alone, but they are also used as elements of many other aggressive displays.

Cranes have several low-intensity displays that are used to intimidate or repel intruders. Most of the displays described below apply to all cranes. The nomenclature for these activities follows Ellis et al. (*In prep.*). We will describe the displays in increasing order of intensity. In perhaps the lowest intensity display, a crane raises its head to full height and extends its neck upward and slightly forward in an

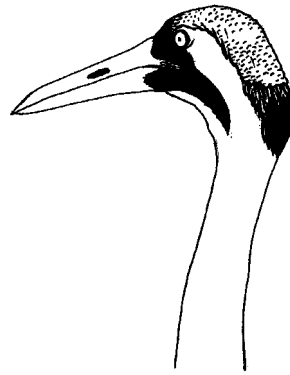


Fig. 6.3. *Crown expansion: Whooping Crane.*

Art Billi Wagner

Alert-posture. When in this posture, and during many of the social displays that follow, most cranes perform **Crown-expansion** or **Bare skin expansion**. The crane may then begin a ritualized display walk (**Horizontal-strut** or **Vertical-strut**) with the bill slowly bobbing up and down in time with exaggerated, rhythmic steps (Fig. 6.4). The toes

are rigidly fanned and extended during the **Vertical-strut**. The crane may either turn its bill away from the intruder (**Crown-present**) or direct its bill downward toward the intruder. In higher intensity strutting, the crane either increases the speed of the walk or lowers its head (sometimes nearly to the ground). Common, Hooded, Whooping, Black-necked, Sandhill, and Siberian Cranes (listed in descending order of feather elevation) raise their tertial feathers into a bustle during display walks (Fig. 6.4).



Fig. 6.4. *Strut (vertical): Eurasian Crane.*

Art Billi Wagner

Another low-intensity threat is the **Ruffle-bow**, in which the crane elevates its feathers and slowly, at first, ruffles its plumage until at length the whole body is rapidly shaking (Fig. 6.5). This display is much like the maintenance **Ruffle-shake**. All species perform the Ruffle-bow, and the Blue, Demoiselle, and Wattled Cranes use this as their principal display. Sandhill Cranes end the Ruffle-bow by throwing the head downward, then preening the breast or tibiotarsus. The Whooping Crane tucks the bill high at the end of its Ruffle-bow (Fig. 6.6). All cranes have a **Ritualized-preen** display in which they place the bill between the back and one wing (Fig. 6.7). Siberian Cranes lower the primaries of one wing and do not move the bill during the display. Other cranes do some rudimentary preening movements and periodically raise their heads to look around. Several species stamp their feet (**Stomp**) a few times in conjunction with the Ruffle-bow or Ritualized Preen. The Red-crowned Crane often Stomps, then arches its head back and raises its wings above the back until its head is nearly between the wings over the back (**Arch**). The Whooping Crane version of the Arch we call the Butterfly (Fig. 6.8). The Red-crowned Crane and its nearest relatives (see Chapter 1) sometimes raise their wings slightly during more intense versions of the Vertical-strut. The Brolga, Sarus, and White-naped Cranes have an exaggerated downward bow as part

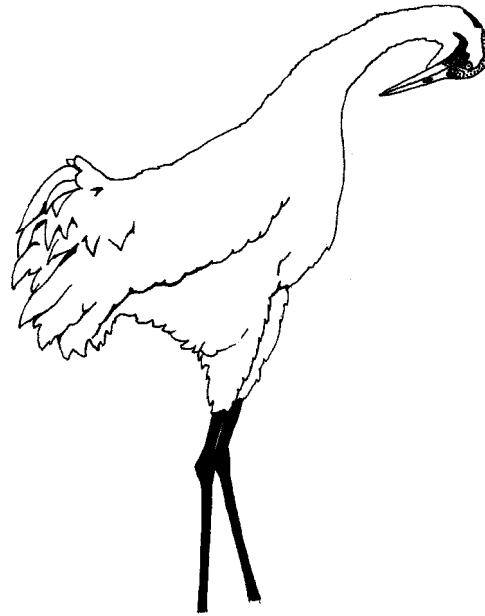


Fig. 6.6. *Ruffle-bow-up: Whooping Crane.* Art Billi Wagner

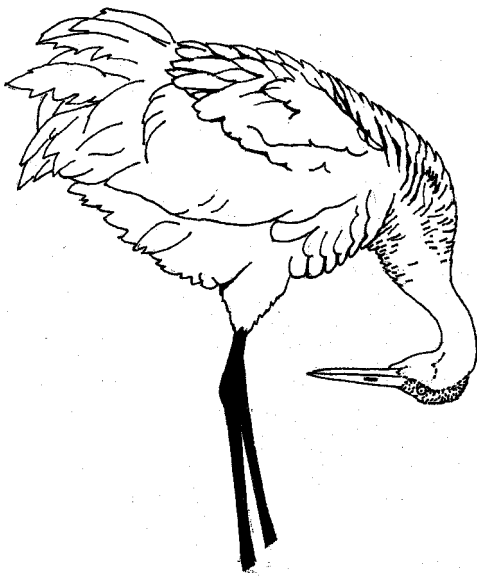


Fig. 6.5. *Ruffle-bow-down: Sandhill Crane.* Art Billi Wagner



Fig. 6.7. *Ritualized Preen: Whooping Crane.* Art Billi Wagner

of the Ruffle-bow. After the ruffle, they usually bow deeply forward, then do a Ritualized Thigh Preen or elevate the head far over the back.

We believe that the most aggressive display is the **Crouch**. In this display, the crane lowers to lying posture with its wings slightly to mostly spread and its bill in front, usually touching or probing at the ground. After performing the Crouch, cranes sometimes **Rush** an intruder by springing up suddenly and charging headlong, flapping their wings and gliding over the ground rapidly if the intruder fails to depart. The crane terminates the Rush with either a Stomp and Ruffle-bow, or by **Attacking** the intruder. Otherwise, cranes usually end the Crouch display with an Arch (Red-crowned) or Ritualized-preen (Siberian, Sandhill and others).

Methods of **Attacking** (Fig. 6.9) include spearing the opponent with the bill (**Bill-stab**), **Leaping** in the air and slashing the intruder with the inner toenails (**Jump-rake**), and thrashing with the wings (**Wing-thrash**). Cranes sometimes use stealth to approach an intruder: they circuitously walk closer and closer while feeding, then abruptly Rush the intruder (crane, human, or other animal). Rushes can also lead to aerial pursuit as a territory owner drives away an intruder, sometimes slashing at the intruder with its feet even during flight.

Aggressive cranes may also bill-spar with one another, spreading their wings and Bill-stabbing at their opponents while standing erect. This may

or may not lead to Jump-raking. Cranes also **Hiss** loudly while bill-sparring. A minor form of Attack is to peck at or grasp the wings or tail of a subordinate to displace it.

Vocalizations can indicate fear or aggression, and like the Unison-call, help drive away intruders and maintain pair bonds (Archibald 1976a, 1976b). Pairs **Unison-call** (Fig. 6.10) after repelling intruders. **Guard-calls**, which are short blasts separated by several seconds, also help to defend the territory (Archibald 1976b). See Archibald (1976a) and Chapter 11C, especially Fig. 11C.1, for more details of these vocal displays.

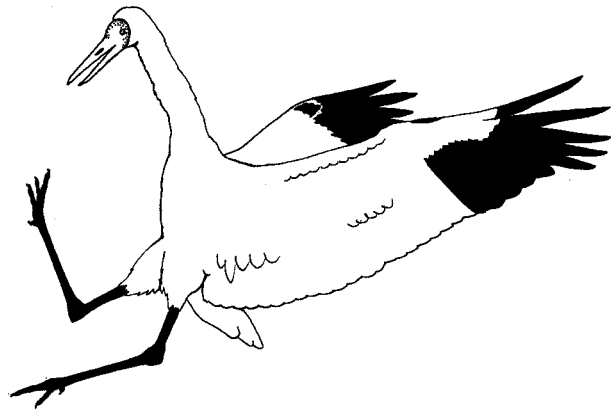


Fig. 6.9. *Jump-rake: Siberian Crane.*

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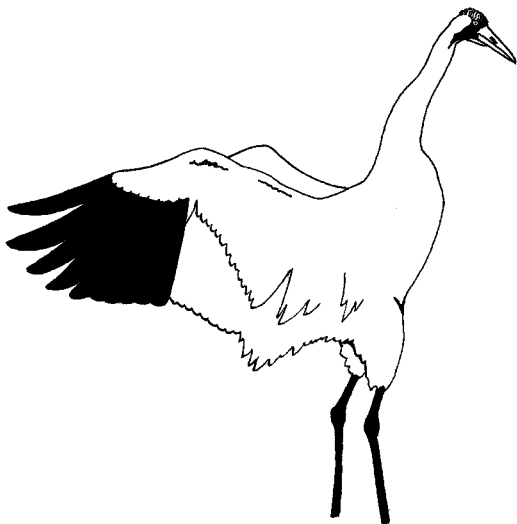


Fig. 6.8. *Butterfly: Whooping Crane.*

Art Billi Wagner



Fig. 6.10. *Unison-call: White-naped cranes.*

Art Billi Wagner

Dancing consists of bouts of Rushing, Flapping, Leaping, Tuck-bobbing (Fig. 6.11), gaping, and feather or stick tossing. Dances last from several seconds to a few minutes and are usually pair-related. However, young cranes often dance in apparent appeasement of dominant cranes. Sometimes Dancing includes sham Bill-stabs and Jump-rakes, but in stable pairs, physical contact is usually not present. Many other elements of aggressive displays (e.g., Crown-expansion) are evident in Dances.

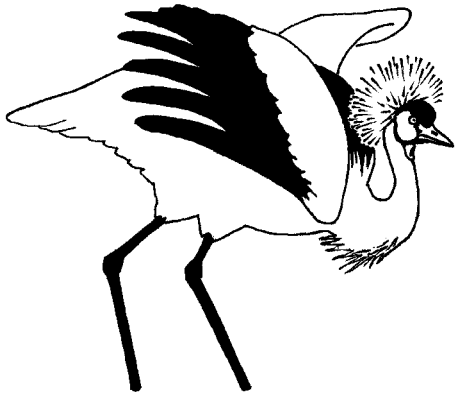


Fig. 6.11. *Tuck-bob: Gray-crowned Crane.* Art Billi Wagner

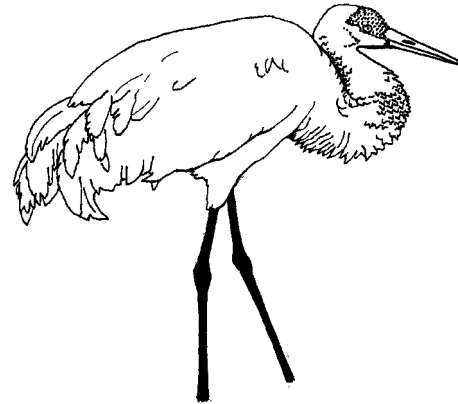


Fig. 6.12. *Cower: Sandhill Crane.* Art Billi Wagner

wings slightly (the elbow is lifted away from the flank), and give purring calls. Chicks often perform this display. When adult cranes Cower, they are probably reverting to chick behavior to placate a dominant crane or person. Often a submissive crane will spread its wings with the trailing edges drooped and turn its back to a dominant crane (or human) as in the adult female's **Pre-copulatory Display** (Fig. 6.13). This behavior is not limited to one sex and is often seen in chicks and subadults.

Submissive Behavior

Submissive displays and fleeing behavior are useful indicators that a crane is stressed by its environment. Some captive cranes run away when a person or dominant crane approaches and may push at the fence as if to walk through it. The crane may also nervously pace the fence, rake its feet along the fence in a climbing manner, and drag its neck and bill along the fence. These disturbance-related activities can cause physical damage, especially abraded wrists, and even more seriously, broken bills.

A submissive crane (Fig. 6.12) usually retracts its neck and adopts a hunched posture (**Cower**) with its head and neck feathers fluffed, and its crown, wattles, and/or bare skin patches contracted. Frequently, subordinate cranes lower their heads, spread their

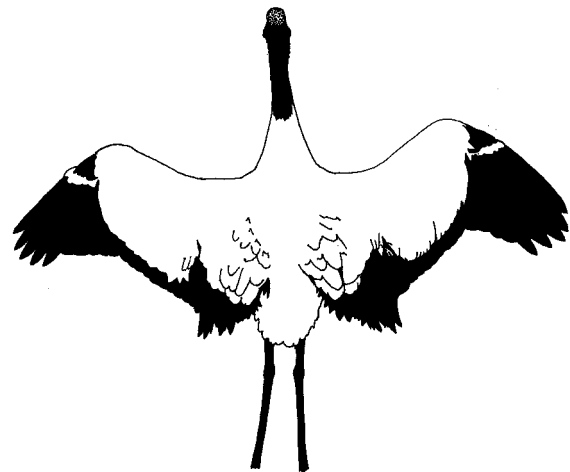


Fig. 6.13. *Pre-copulatory display: female Black-necked Crane.* Art Billi Wagner

Behavioral Management of Chicks and Subadult Cranes

This section provides a summary of the management of young chicks; other details are provided in Chapter 5. Chicks have strong social needs. Most species are gregarious, and nearly all cranes flock during the non-breeding season. Flocking probably results in increased survivorship and in foraging advantages. Cranes three months to three years old should be socialized with conspecifics so that they develop normal social behavior. Combine cranes in groups of two or more in pens that are at least 100 m² for two cranes and proportionately larger for larger groups. The group will establish a dominance hierarchy (Derrickson and Carpenter 1980) based on the size and sex of the cranes, with males and larger cranes typically being more dominant (Kepler 1976).

Parent-reared cranes will try to establish **vocal contact** with their parents after separation and will also try to reunite if allowed. Place such cranes at least 200 m away from and out of sight of their parents until they integrate into a social unit with other colts. Parent-reared crane chicks are typically fearful of people. As such, they should be habituated to humans if they are to remain in captivity (Archibald and Viess 1979). Frequent, nonstressful encounters with humans will help calm them down (e.g., provide a food treat when entering the pen). Placing wild colts with tame cranes will also help.

In grouping colts, avoid penning cranes together that are likely to be paired later. Cranes treat their penmates as siblings and may refuse to pair with them later. It is wise to keep intended mates separate until they are at least 18 months of age.

Cranes that are 6 months old or older often become aggressive towards their penmates, particularly those of the same sex. Groups should be reorganized when excessive aggression appears. However, it will help to place two food/water stations at opposite ends of the pen so submissive cranes can eat and drink. Even then, watch for cowering cranes that are afraid to go to the food. In large pens with 10-15 birds, a third station may be needed. Submissive cranes that are regularly attacked by penmates should be removed.

Chicks spend much time practicing flying when they are two or more months old and need an unobstructed area at least 15 m long for exercise.

They may be wing clipped after they fledge. Opinions vary on whether permanent flight restraint (see Chapter 11E for details) can prevent males from balancing properly during copulation and thus limit fertility. Many pinioned and tenotomized males fertilize their mate's eggs. However, in captive Red-crowned Cranes (the heaviest crane), full-winged males had higher rates of natural fertility than pinioned males (Belterman and King 1993).

Keep subadult cranes in genetically and behaviorally compatible groups (2 to 16 individuals) until they are paired. When pairs form in group pens, extreme aggression often appears, with the dominant pair often monopolizing food or water and occasionally killing pen mates. If the new pair in a group pen is desirable, move the pair to a breeding pen. If the pair is unwanted, remove one or both cranes to reduce aggression.

If sex is known, juveniles can be grouped by gender, however, same sex groups can also be aggressive, and homosexual pairs can form in such groups (Archibald 1974b; Kepler 1978; Derrickson and Carpenter 1987). Early sexing increases the chances of early reproduction through behavioral management.

Behavioral Management of Breeding Cranes

Cranes have pronounced species specific variation in nesting phenology (see Chapter 3). Their captive management should reflect species differences and allow for individual variation as well.

Annual Behavior Cycle of Breeding Cranes

Cranes exhibit seasonal cycles in social displays especially in activities related to pairing and those involved in rearing young. It would be helpful to have data on seasonal trends for all species of cranes in your colony, but such is available only for the Whooping Crane and Mississippi Sandhill Crane as described below. Also, Katz (1979) presented somewhat similar data for two pairs of Hooded Cranes from late February through early June.

For boreal species, display intensity ebbs in winter, but on warm days in late winter and early spring, display frequency and intensity increase. As the breeding season approaches, vocal and visual behavior

related to pair-bonding, territorial defense, and breeding is increasingly evident. There is also good evidence for decreases in performance of some displays during the molt and during the incubation and chick rearing period when the adults tend to be more secretive. The intensity and frequency of several vocal and visual displays again increases in fall, suggesting an autumnal recycling perhaps in response to a photoperiod mirroring that for spring. Thereafter, performance tendency wanes through the early months of winter.

In the following paragraphs, we will show some general trends in this annual cycle and compare behavior trends for the migratory Whooping Crane with those of the nonmigratory Mississippi Sandhill Crane. Male-female differences will also be emphasized. These generalizations are presented because of their usefulness in crane husbandry. Also there are seasonal cycles in non-social behavior such as the increase in fall food intake (or at least body weight) especially evident in northern latitude breeding cranes (Swengel 1992).

Our data on social displays derive from a 15-month period during which we conducted a standard morning "walk-through" in the crane colonies at Patuxent and recorded the responses to an approaching human. Because hand-reared cranes responded to the approaching human as though he was an intruding crane, for hand-reared pairs we were able to evaluate seasonal trends merely by recording all social displays as we walked through the colony. Our walk through the colony did not elicit social displays in cranes reared by crane foster parents (except for fleeing behavior) so parent-reared cranes were eliminated from the comparisons shown below. Data were used for 12 male and 12 female Whooping Cranes and 9 male and 7 female Mississippi Sandhill Cranes. All data were taken on form sheets as an observer (who was familiar to the cranes, but who avoided entering their pens) approached to within ca 2 m of the pen between 0.5 and 3.5 hours following sunrise.

As for all species with a red cap, Whooping Cranes, and to a lesser degree Mississippi Sandhill Cranes, expand the red, bare-skin areas and present this area toward approaching intruders (Fig. 6.3). For both species, Crown-expansion and presentation values are high throughout the year,

except during incubation or molt when a crane is trying to be inconspicuous. Crown contraction at other times signals fear or illness. The Whooping Crane exhibited two peaks. Females of both species cycle in concert with their mates, but they, on average, expand to a lesser degree than males.

The Unison-call (Fig. 6.10), a form of antiphonal duet given by both members of a pair, can be performed throughout the year and is given primarily in response to conspecific intruders (Walkinshaw 1973a; Archibald 1976b). Well-coordinated, frequent Unison-calling signals that a pair is properly bonded. Duetting is also believed to be important in the synchronization of the crane breeding cycle (Voss 1977). In the Whooping Crane pairs studied at Patuxent, the Unison-call was heard with some frequency all months of the year (Fig. 6.14), but was highest between October to April and lowest during the July molt. Unlike our Whooping Cranes which did not breed during the study year, our Mississippi Sandhill Cranes exhibited a strong peak from June through August when they did breed. This periodicity suggests the importance of the Unison call as a territorial display while breeding (Mississippi Sandhill Crane data) and also in reinforcing the bonding of a pair during migration (Whooping Crane data).

Contact- or Flight-calling was entirely absent in our pairs of the non-migratory Mississippi Sandhill Cranes, but our Whooping Cranes exhibited a strong peak in March and April and a minor peak in September (Fig. 6.15). The Contact-call communicates a bird's disposition to fly and is probably important in synchronizing long distance movements of the pair. Contact-calls may be given synchronously

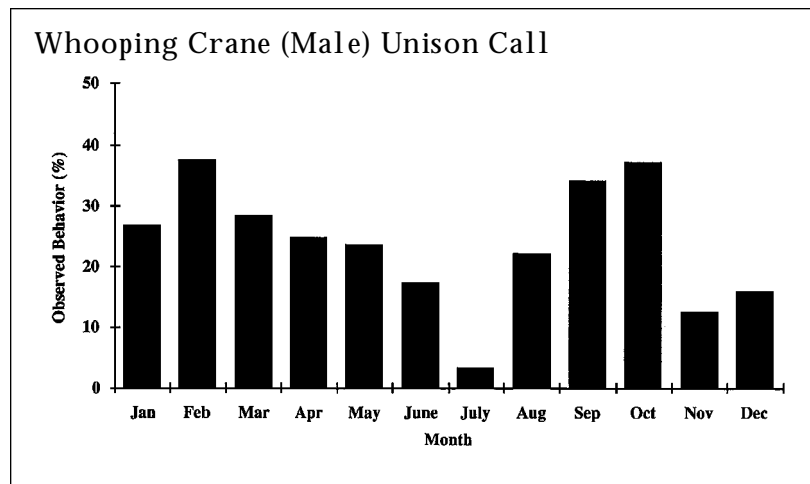


Fig. 6.14. Seasonal pattern in Unison-call performance for Whooping Cranes.

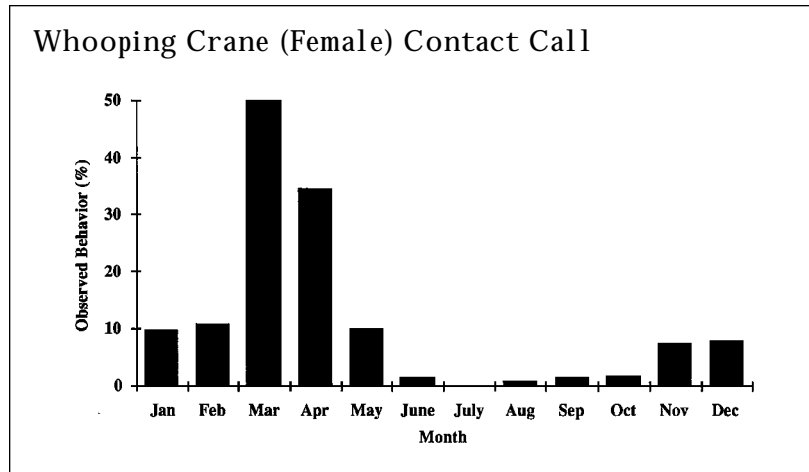


Fig. 6.15. Contact-call performance by month for female Whooping Cranes.

by males and females or singly by either sex. The male trend (not shown) closely paralleled the female trend (Fig. 6.15).

Contact-calling is often accompanied by a highly stereotyped Pre-flight posture (Fig. 6.16) wherein the neck is extended up and far forward. Both ethons had similar performance trends in both species.

We have observed, but not quantified, that the intensity of flight related behavior patterns is directly related to the length of the migration; being greatest in Siberian Cranes, followed by Whooping, Hooded, Red-crowned, White-naped, and Greater Sandhill Cranes. In captivity, the intensity of migratory restlessness seems also to decrease with age.

The Guard-call or Alarm-call is normally given in response to a distant disturbance such as an unfamiliar human or other alarming stimulus animal. In both Mississippi Sandhill and Whooping Crane pairs, the Guard-call is given less often than the Unison call, and seldom during the early part of the breeding season.

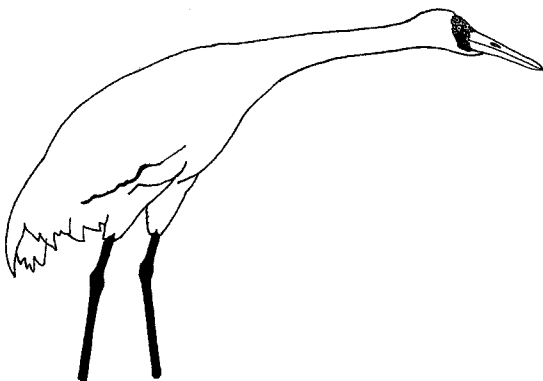


Fig. 6.16. Pre-flight Posture: Siberian Crane. Art Billi Wagner

The average distance between members of a pair is another indicator of the strength of the pair bond, especially during migration and nesting. During the incubation period, however, the non-incubating parent seems to avoid the vicinity of the nest causing a peak in the average distance between mates.

Both sexes of both species of cranes performed the exaggerated, rigid-toed, high-stepping that we call Strut. When Strutting, the body axis is either rotated down and forward (Horizontal Strut) or elevated anteriorly (Vertical Strut, Fig. 6.4). During a Strut performance,

Crown-expansion and presentation also occur. Although both species and both sexes Strut, in the Mississippi Sandhill Cranes, the males normally Horizontal Strut with their mates walking (Unison walk) in tow. Male Whooping Cranes typically Vertical Strut while their mates remain stationary and perform ritualized Preening or some other social display. For the male Mississippi Sandhill Crane, Strutting exhibited a single peak (Fig. 6.17) in midsummer. Strut was without peaks and much less frequently performed in the Whooping Crane.

Crouch is the display that shows the strongest male-female and species differences. For Crouch, a crane flops onto the ground and lies as if brooding young while aggressively billing the surrounding vegetation. Female Mississippi Sandhill Cranes may perform Crouch any time of the year (Fig. 6.18), but a strong peak is evident when they are rearing young. Male Mississippi Sandhill Cranes and either sex of Whooping Cranes seldom perform this display.

Probably all species show some unique seasonal trends. For example, most cranes lay eggs in spring or summer, however, many wild Wattled Cranes lay in winter (Johnson and Barnes 1991) and their captive counterparts lay eggs from fall to spring (Beall 1985). Gray Crowned, Black Crowned, Blue, and Demoiselle Cranes may also lay eggs in indoor winter quarters (K. Kawata, Detroit Zoological Park, Royal Oak, Michigan; R. Lastavica, Omaha Zoo, Omaha, Nebraska; and P. Strasser, National Aviary, Pittsburgh, Pennsylvania, personal communications).

Some performance trends are probably common to most or all cranes. For example, we hypothesize that cranes (captive and wild) vocalize less once the eggs

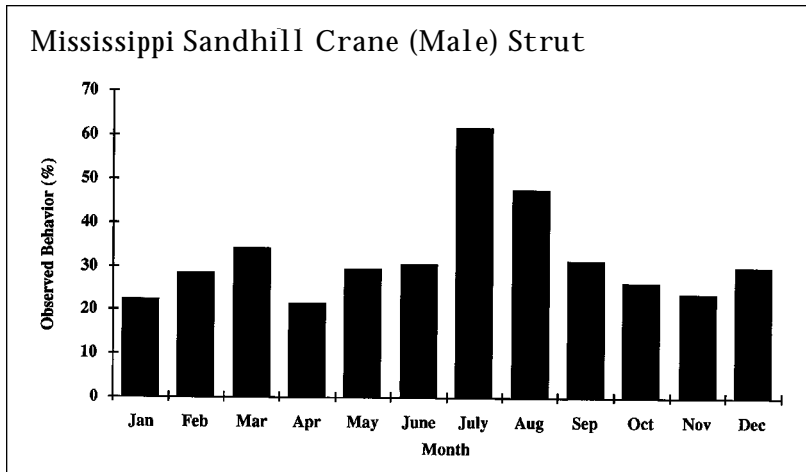


Fig. 6.17. Monthly performance levels in strut for male Mississippi Sandhill Cranes.

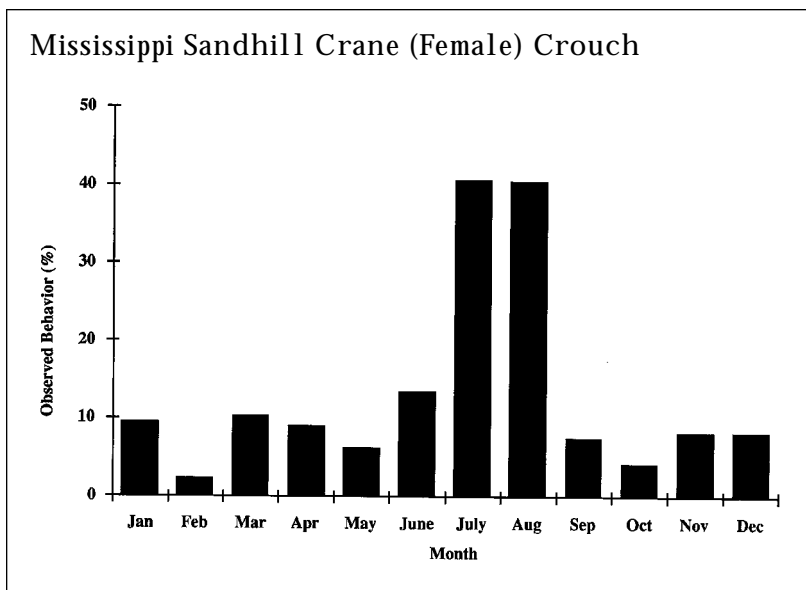


Fig. 6.18. The annual cycle in Crouch performance for female Mississippi Sandhill Cranes.

are laid. Katz (1979) found what appeared to be reduced calling in captive Hooded Cranes as the egg-laying season approached. Probably all species of cranes (captive and wild) begin copulating 2 to 5 weeks before egg laying, even those that are still migrating (Littlefield and Ryder 1968; Walkinshaw 1973a; Littlefield 1985). Captive pairs build nests a few days or weeks before egg laying. By watching for the onset of nest building, a colony manager may know when to begin artificial insemination (AI) to fertilize the first eggs.

Surprisingly, in our walk-through study, we failed to find strong seasonal trends for such activities as

Dancing and Pre-copulatory display for either species, and Strutting was seasonal only in male Mississippi Sandhill Cranes.

All species of cranes feed, brood, and defend their young, and some pairs of most or all species are extremely aggressive when raising chicks. All cranes become more sensitive to disturbance when incubating and rearing chicks. Most cranes show some behavioral changes associated with the molt. Most undergo an annual or biennial simultaneous molt of all flight feathers (Blauuw 1897; see Chapter 7 for physiology of molt and characteristics of juvenal molt). African Crowned Cranes and Brolgas, however, molt continuously; Demoiselle Cranes and most Sandhill Cranes molt sequentially. The molt is physiologically stressful and is accompanied by a decrease in performance values of social displays during the time that the bird is flightless. Such changes are probably adaptations for survival. Because cranes (even normally aggressive individuals) become very shy when molting, minimize human contact during this time. The female may even become the dominant member of the pair while the male is molting. Watch molting cranes carefully to forestall penmate aggression. In group pens, molt can destabilize the dominance hierarchy. Molt can also cause cranes to appear ill.

By knowing behavioral norms for each sex and species, it is possible to promote survival and productivity in captive colonies.

Pairing Cranes

Pairing cranes can begin in the birds' second year. Well-established pairs remain together for many years. When pairing subadult cranes (i.e., 3 years of age), be aware that new pairs are frequently ephemeral. Pairs should not be viewed as permanent until they remain stable for several months and/or reproduce. Wild, subadult Florida Sandhill Cranes usually associate with several potential mates before a firm bond is

established (Nesbitt and Wenner 1987). In choosing potential pairs, be mindful that birds of similar age pair more readily: young cranes are sometimes intimidated by older adults. Potential mates should not be genetically related nor should they have been reared together.

Potential mates should be placed in adjacent pens ideally with a common door to allow herding of a crane from one enclosure to the other without capture. Close contact can be encouraged by placing food and water near the fence dividing the two pens. Pairing pens should be arranged so that concealed caretakers can observe the birds and quickly enter the pens if necessary to separate the birds.

Pairing stages were summarized by Mirande and Archibald (1990) as follows. The first sign of pairing in the wild is one crane following another. In captivity, pairing is evidenced when the birds frequently stand side-by-side. As pairing continues, the behavior of the two birds becomes progressively synchronized. They feed and rest simultaneously. Synchronized displays, such as threats, Guard Calls, and Unison-calls may also indicate pairing; however, such behavior can also indicate intense intrapair aggression or aggression toward caretakers. A crane may interpret its intended mate's aggressive displays as sexual attraction, but when the birds are placed together, the aggressive crane may attack. To prevent injury, keep an over-aggressive male in the pen adjacent to the female; she may thereby be stimulated to lay eggs without risk. For AI pairs with aggressive males, this is sometimes the best strategy, even long term.

Although cranes (even chicks) sometimes **dance** solo or in larger groups, dancing is also associated with pairing and is believed to synchronize mates for successful copulation. Lack of dancing between two cranes can indicate that pairing is not occurring. As a pair bond strengthens, the male generally becomes more defensive of the enclosure than does the female. The ultimate indication of successful pairing is copulation. Pairs that are well bonded should at least attempt to copulate, although some will be unsuccessful because of wing injury, etc.

When it is time to **move the cranes into the same pen** for the first time, move the more dominant bird (usually the male) into the subordinate crane's enclosure. This provides the subordinate bird with a psychological advantage because the pen is its territory. Watch the birds constantly at first and separate them immediately if excessive aggression is observed. Dancing strengthens the developing

pair bonds, especially when the female continues to dance after the male begins to run and flap in mock Rushes. In unpaired birds or unstable pairs, dancing can intimidate the subordinate crane. If one crane keeps dancing while the second crane flees, the first crane may chase the fleeing crane and attack. Death or serious injury can result if the cranes are not immediately separated. A single attack can negate weeks of progress in the pairing process.

Cranes can be further manipulated to promote pairing by brailing one wing of the dominant bird before placing it in the enclosure of the submissive crane. The brail stresses the crane, reducing its aggression. Dominance in cranes is related to height; more dominant birds are generally taller than submissive ones. However, the dominance of a submissive crane can be increased by providing a **mound of earth** 0.3 to 0.5 m high near the fence separating the cranes. Displaying cranes will often stand on such mounds. By this simple manipulation, the dominance of a subordinate crane may be increased.

If cranes seem compatible, they can be left together during the day with hourly checks. When Unison-calls and dancing do not lead to aggression or intimidation, the pair is considered to be solidly mated and can be trusted to occupy the same enclosure at all times.

For cranes, some action patterns are "contagious." For example, if one bird yawns or flaps its wings there is a good chance that one or more penmates will do likewise. If one crane is extremely aggressive towards humans, its penmates will often become aggressive. By pairing cranes with different behavioral traits, characteristics of one crane can be encouraged in the other. Parent-reared cranes become much tamer and adapt to captivity better if penned or paired with hand-reared birds. Conversely, hand-reared birds can become less attached to humans if integrated with parent-reared mates.

Some paired cranes that have not bred can be induced to become more confident and better bonded if a crane chick is placed in an adjacent pen. Sometimes the pair attempts to adopt the chick (as evidenced by their passing food items through the wire to the chick, brood calling, and by their lack of threat displays); others will try to kill the chick or ignore it.

Stress and Disturbance

Crane pairs are healthier and breed better when disturbance is minimized (Mirande et al. 1988 unpubl.). Pairs are normally less stressed when: (1) their pens have visual barriers separating them from neighboring cranes; (2) routine tasks are done on a regular schedule; (3) the same people perform these tasks; and (4) the breeding area is closed to certain kinds of vehicles (aircraft and large trucks) and activities (pen repairs and construction) during, and three months prior to, the breeding season.

Some cranes breed well without **visual barriers** between neighboring pairs, but pairs should at least be separated by empty pens to prevent fighting through the fence. Sandhill, White-naped, Red-crowned, and some other species have bred well without visual barriers, but Whooping, Sarus, and Siberian Cranes need them.

Cranes breed best when they have a large, secure **territory**. Breeding pairs need at least 100 m², but 300 m² is preferable. They often benefit from a shelter in which to retire from view or gain protection during inclement weather. Trees or bushes in the pen may provide **natural cover**. Some pairs breed better if they have a secluded spot for nesting. Satisfy their need to build nests by providing dry twigs or coarse grasses (fine or moist vegetation will mold more rapidly).

If AI is intended, line **capture corners** with nonabrasive cloth such as tennis netting for 3-6 m in each direction from the corner. At ICF, discarded 2 m tall conifers (old Christmas trees) are used to line these corners.

Stress can be reduced by taming cranes. The process involves conditioning birds to human activity through providing treats (favorite foods), avoiding direct eye contact, announcing your approach by calling when still far away, and other techniques as discussed in Chapter 5.

Pair Bonds

Well paired cranes perform synchronous activities and stay near one another most of the time. If a male is excessively dominant or if the female is dominant over the male, the pair may never breed (Derrickson and Carpenter 1987). Several circumstances can result in weak pair bonds. If one member of a pair is excessively submissive to the other or if one mate prefers a neighboring crane, the pair bond can be weakened. An

unstable pair bond can result in one crane injuring or killing its mate.

Some pairs are compatible but never lay eggs; this occasionally occurs with birds that were paired when very young. Such birds seem to view each other as siblings. For these pairs, pair-related displays and territorial defense are less intense.

The "**Location-call test**" is a good means of testing the strength of a pair bond. This requires that the male be removed from the female by at least 100 m, but within earshot. If both cranes perform the loud, single-note Location-call, and promptly answer the calls of their mate from a distance, the pair bond is probably genuine. If either member of the pair fails to Location-call or fails to respond to the other crane's Location-calls, the pair bond is probably weak and the pair should be dissolved.

If a closely bonded pair is to be divided and new pairs formed, Location-calling can seriously delay or prevent the re-pairing process. It is wise to postpone introducing the new intended mate until a week or more following separation of the old pair. Former mates must sometimes be separated by a great distance (1 km or more) to facilitate pairing them with other cranes.

Sometimes **wild cranes harass captive pairs** which can result in captive males redirecting aggression toward their mates. In these situations, pairs may need to be separated until the wild cranes leave. The pair can generally be safely reunited after a few days.

Wild cranes occasionally **switch mates** even though neither member of the pair has died (Littlefield 1981; Nesbitt and Wenner 1987). Young cranes frequently form ephemeral pairs in the wild and may take years to form permanent pairs (Bishop 1984; Nesbitt and Wenner 1987). Pairs that produce offspring are much more likely to persist (Nesbitt and Wenner 1987). In wild Florida Sandhill Cranes, re-pairing efforts vary by sex: males quickly find new mates, while females may take several years to re-pair (Nesbitt 1989).

Hand-reared cranes that are overly attached to humans can often be made to breed if they are given a suitable mate, and if thereafter they have minimal contact with humans. Once the pair has eggs, its pair bond is often strengthened, and further bonds to humans are weakened by the pair's mutual defense of the eggs or chicks.

Before including birds in an AI program, allow young pairs to attempt copulation for one or two breeding seasons. Flightless males that are unable to fertilize eggs due to unilateral wing impairment may

be clipped on the whole wing to improve their wing symmetry and thus help them balance. Allowing the pair to raise a chick may also synchronize their reproductive cycles or strengthen their pair bond, thereby increasing the chance of fertility in the future.

Parent-reared cranes may be more likely to copulate than hand-reared ones, and cranes hand-reared in groups may be more likely to copulate than birds that were hand-reared alone (Derrickson and Carpenter 1987). However, most hand-reared cranes that are socialized with others as colts learn to copulate when they become adults.

If a pair does not produce fertile eggs after one or two years of management as described above, it may be necessary to re-pair them or initiate AI (Chapter 11A). In **using AI**, it is important to disturb the birds as little as possible. Some cranes will not lay eggs when they are regularly handled for AI. One strategy for such cranes is to wait until the female starts laying eggs, and then initiate AI. Normally, the female is not so stressed by this handling that she fails to lay more eggs. This strategy is less useful with Wattled Cranes, which frequently lay one-egg clutches. For those Wattled Cranes that are adverse to AI, one insemination 4-10 days before the next egg is expected can often produce a fertile egg (Monica Tuite, unpubl. data). The best AI schedule depends on the particular female's laying history (see Chapter 3). Inseminating a nervous female a few days before she is scheduled to lay her second and subsequent clutches, **but not repeatedly between each clutch**, can improve the chances of getting several fertile eggs while **minimizing disturbance**. Finally, be sure that egg searches and other visits to the pens of shy cranes are performed quickly and, if possible, use binoculars to scan the pen from a distance.

Behavioral Management of Cranes for Display

Cranes on public display normally receive more disturbance than other captive cranes. Because many cranes will not breed while on display, it is best to exhibit only those birds that are of low genetic value. Display cranes should tolerate human visitors but not be aggressive towards them. Extremely aggressive cranes are dangerous to caretakers and the public, and may damage themselves in their attempts to attack

people. To reduce the chance of injury to birds and visitors, design display pens so that the public cannot come closer than 1 m to the cranes.

Cranes on display adopt a daily schedule timed to periods of human visitation. They direct many of their social displays toward the public, and most remain within public view.

Several management practices can encourage exhibit cranes to breed. Caretaker entrances should allow servicing so that a portion of the pen is left undisturbed by keepers and the public. Performing some activities out of sight and with minimal contact encourages breeding. Cranes on display feel safer when they have a "sanctum" where they can go out of sight of humans. This hiding place can be an indoor shelter, a sheltered corner, or a patch of dense foliage ca 1.5 m tall.

Novel pen designs can also improve the display value of cranes. Elevated overlooks or moats allow people to view cranes unobstructed. These designs, however, require that the cranes be flightless.

Mixed species exhibits are attractive, but because cranes are solitary nesters, they are unlikely to breed when there are more than two cranes in one display. The dominant pair will too often defend most of the pen, driving the remaining cranes from its territory. Such pairs may even breed. However, dangerous encounters are likely whenever a breeding pair is penned with conspecifics. The only situation in which a breeding pair can coexist peacefully with other conspecifics is when the pen is very large. Patuxent maintains three pairs of Florida Sandhill Cranes in a 2 ha enclosure. The dominant pair normally defends more than half of the area while leaving enough room for the two subordinate pairs to escape and breed.

Behavioral Management of Cranes for Release

This section summarizes the management of cranes destined for release. For more details see Chapters 5 and 11D.

Parent-reared and hand-reared cranes for release are managed very differently. Parent-reared chicks develop normally and require no special training. Chicks reared by hand, in isolation from human contact, should be allowed to see and hear conspecific adults so that they learn to socialize and breed

with conspecifics. Normally, hand-reared chicks are handled by costumed human caretakers (costume-rearing).

Costume-reared chicks can be taken to the release site when they are as young as 10 weeks (Horwich 1986). Young cranes exhibit the behavioral plasticity important in rapidly learning new survival techniques. Horwich (1986, 1989) released his costume-reared cranes in early fall to coincide with the period when wild cranes are the most gregarious. Other studies also suggest that captive-reared cranes integrate better with wild cranes if released in early fall (Mitchell and Zwank 1987). Whatever the rearing method, cranes should be released gently over a period of weeks (**gentle-release**) to give them time to acclimate (Horwich 1986; Mitchell and Zwank 1987). If the chicks are costume-reared, this acclimation period can also be used to introduce them to natural foods.

Release pens should be large enough to allow cranes to move comfortably away from mammalian predators outside their enclosure. Pens used in successful releases of Mississippi Sandhill Cranes have been 0.5 to 2 ha (1-5 acres) in size.

Imprinting Attachment, and Behavioral Development in Cranes

contributed by Robert H. Horwich

There have been few studies of crane imprinting or early development (Voss 1976; Layne 1981). Most imprinting research in the 1960's focused on the short-term effects of imprinting on social preferences. Domestic fowl (*Gallus domesticus*) and domestic ducks (*Anas platyrhynchos*) rapidly restrict their filial attachment and following response to their parent, human caretaker, or to other stimuli encountered shortly after hatching. This learning phenomenon has been called **filial imprinting** (Bateson 1978). Many studies (Hess 1973; Hess and Petrovich 1977) show that there is a "critical" period when precocial birds imprint on a parental model.

An accumulating body of evidence indicates that relatively **early experiences** have profound effects on sexual choice later in life (Immelmann 1972; Bateson 1978). This evidence indicates that exposure to social

stimuli at certain age periods can reverse the preference of early filial imprinting (Gallagher 1976, 1977; Vidal 1976, 1980).

Filial Imprinting and Parental Care

Cranes exhibit imprinting patterns similar to domestic fowl. Imprinting probably begins in the egg about 2 days prior to hatching, when chicks begin answering the parents' brood calls. Chicks follow the adult on the first day and are often away from the nest by day 1 or 2 (Walkinshaw 1973b). Parental attachment is complete within the first 3 days and becomes stronger during the first 2 weeks.

Attachment is reinforced by a radical change in parental behavior at hatching. This includes increased brood calling, brooding, preening the chick, preening the adult's brood patch, and feeding the chick. This behavior encourages imprinting and the development of a following response by the chick during the first week, the initial sensitive period of development (Hartup and Horwich 1994).

Brooding of the chick occurred only during the first week in our study of Sandhill Cranes. Preening the chick, although rarely seen, was done by the female while brooding. Wild cranes may brood chicks that are up to six weeks old (G. W. Archibald, ICF, personal communication). The female invites the chick to brood by extending the wrist joint laterally while calling loudly and pointing her bill into or preening the opened cavity. The moving bill tip is very attractive to crane chicks, and probably induces the chick to accept brooding. Pecking the parent's bill, the chick's greeting, is a ritualized feeding behavior. It was elicited in puppet-reared chicks by extending the puppet's bill toward the chick (Fig. 6.19). A stereotyped bill presentation by White-naped Cranes also elicited the bill peck. A similar feeding posture occurs in wild Sandhill Cranes (Layne 1981).

Sexual Imprinting

Sexual imprinting is a form of learning which shares many characteristics with filial imprinting, but which also influences mate choice at sexual maturity. There are many instances of birds sexually imprinting on humans or other bird species (Immelmann 1972), but studies have shown that the process is reversible if cross-fostered or hand-reared birds are introduced to their own species during or before the end of the sensitive period.



Fig. 6.19. *Siberian Crane chick greeting puppet head.*

Photo David H. Ellis

Vidal (1976, 1980) neatly delineated two imprinting periods in chickens. He noted an early sensitive period for learning the following response and a second sexual imprinting period at 30–45 days. Cockerels exposed to a model at this later period became sexually imprinted on it despite their earlier training to follow a different model.

Proper sexual imprinting is critical in crane reintroduction programs. Although the rearing of Whooping Cranes by wild Sandhill Crane parents has produced a small wild population of Whooping Cranes in the Intermountain West, these cranes are not breeding (Ellis et al. 1992a). **Cross-fostering** is believed to have resulted in imprinting problems preventing the Whooping Cranes from breeding with their own species. The recent discovery of a Whooping-Sandhill Crane hybrid at Bosque del Apache National Wildlife Refuge (Pratt 1993) and unusual behavior by cross-fostered females (Mahan 1992; Swengel, personal observation) confirm this.

Imprinting Stimuli

Newly hatched precocial birds can be imprinted on a wide variety of objects in the absence of their natural parents, indicating that early parental recognition is largely acquired (Lorenz 1937, 1970; Ramsay 1951;

Spalding in Jaynes 1956). However, there are some innate preferences (Hinde 1961; Gaioni et al. 1978). Initially, vocal cues seem more important than visual ones (Ramsay 1951; Gottlieb 1971), and there may be other innate preferences for certain colors and forms (Jaynes 1956; Schaefer and Hess 1959; Salzen and Meyer 1968).

As part of our reintroduction study (Horwich 1986, 1989; Horwich et al. 1992), we reared crane chicks with a stuffed crane model that emitted brooding calls, fed chicks using a crane-head puppet (Fig. 11D.2), and led them while costumed and using the same puppet emitting the same calls (Fig. 11D.1). Although the main goal of costume-rearing (see Chapter 11D) was to imprint crane chicks on a crane-like substitute, we also hoped that use of the costume would allow us to control the birds after release while leaving them still fearful of uncostumed humans. The costume, although not overly crane-like, broke up the human gestalt by de-emphasizing the head, face, and hands while emphasizing the crane head and voice. Although the chicks reared with the costume did not show affinity to humans, they did not exhibit much fear either. Before release, an uncostumed person could approach within 3 m of the mildly wary chicks, but after associating with wild cranes for 2 weeks, the chicks' flight distance in response to human approach had increased to 100 m (Horwich et al. 1992). Fear of humans can, of course, be taught (see Human Avoidance Conditioning in Chapters 5 and 11D).

At 4–8 weeks of age, our chicks were given choices of stimuli in an attempt to assess the early effects of filial imprinting (Horwich and Owen unpubl.). For all chicks, the most important stimulus was the moving puppet head. They responded quickly by pecking the bill. It was clearly chosen over a mounted body or a vocalizing speaker.

The moving bill tip directs chicks of all ages in feeding (Hartup and Horwich 1994). When feeding a chick, White-naped Cranes sometimes drop and pick up an insect as many as 15 times before the chick will accept it. This motion was very attractive to chicks, who eventually picked up insects on their own. The parental bill attracts the chick, and the chick greets the parent by purring and pecking the extended bill. Later, juveniles watch the parents' bills probing the ground, and probe the same area. Bill movement is also very attractive in other precocial birds (Tinbergen and Perdeck 1950; Hailman 1967; Johnson and Horn 1988).

We tested the chicks' responses to various parts of the crane puppet head during their first few weeks. None of the main puppet features (red patch, head, or eye) was consistently important to the chicks. Chicks exposed to a mounted body for only a short period tended to choose the puppet, while those exposed to the body for a longer period of time tended to choose the body. This observation follows the general rule that the longer the exposure, the stronger the preference (Bateson 1978). Sound is another very important stimulus for other precocial birds (Gottlieb 1971). Our results indicate that crane chicks are most responsive to brood calls during the first 3 weeks.

These experiments provided information for use in captive rearing. Although red is often used for feeding dishes or for rods dangling in the food bowl to induce feeding in young chicks (Kepler 1978), the red patch of the puppet head did not interest the chicks. The red patch is used in aggressive displays in Sandhill Cranes (Voss 1976). However, when combative Sandhill Crane chicks were separated by the puppet head, they redirected their attacks at the red patch of the puppet (Erickson et al. 1988).

By dangling a puppet-like head in the food dish (Fig. 11D.2), we taught chicks to feed themselves (Horwich 1986; Erickson et al. 1988). By pecking repeatedly at the moving beak tip, they eventually pecked the food below it. This gradually changed to ritualized pecking of the beak tip before feeding until, finally, they pecked only the food. At Patuxent, a taxidermic mount of a crane head (suspended from the ceiling with its bill contacting the food and manipulated from outside the pen; Fig. 5.10) proved effective in teaching Whooping and Sandhill Crane chicks to eat (Ellis et al. 1992b).

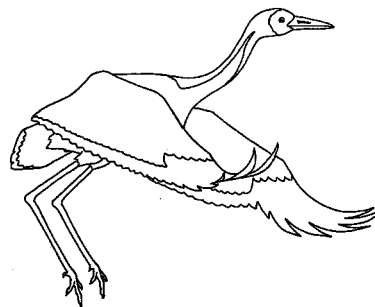
gradually entered a more independent foraging phase at 4-8 weeks of age. At fledging (11-14 weeks), they reattached to the costumed parent, stayed near it much of the time, and pecked its feathers. The intense sociality exhibited during this reattachment period induced the costume-reared chicks to rapidly join wild cranes following release (Horwich et al. 1992). This period seems equivalent to the sexual imprinting period (when the initial attachment can be reversed) in chickens, as identified by Vidal (1976) at 4-6 weeks when the adult plumage was largely complete.

Periodic regressions may be in synchrony with seasonal activities, as seen in mammals (Horwich et al. 1977; Horwich et al. 1982) and cranes (Horwich 1987; Horwich et al. 1992). The initial close bond of parent and chick during the first month protects the chick when it is most vulnerable and needs parental feeding. As the chick grows stronger and can feed itself, it begins a period of independent foraging, during which it follows its parents at a greater distance. The chicks regress by increasing contact with the parents at fledging time when they would otherwise be most likely to become lost if they fly far from their parents (Horwich 1987). This renewed bonding may also involve species and sexual identification. They exhibit a second reattachment period just before and during migration (Horwich 1987; Horwich et al. 1992). Many other bird species, both migratory and non-migratory, as well as mammals, show this same cyclic gregariousness (Nievergelt 1974; Guinness et al. 1979). Besides functioning to keep cranes on the correct migration route, this reattachment or gregariousness may allow orphaned chicks to learn the route from flock mates in the absence of their parents.

Behavioral Cycles and Reattachment Periods

Quantitative studies of bird and mammal behavior have shown that parent-young attachment and many other activities follow a cyclic pattern, with two or more cycles occurring in young animals before fledging or weaning (Horwich 1974, 1987; Ellis 1979). This has been termed a **regression** or **reattachment period** (Horwich 1974). This concept is fundamental to understanding ontogeny and sociality in mammals (Horwich et al. 1982) and birds (Ellis 1979).

After the initial attachment period, there follows a period of gradual independence from the parent. After spending 60% of their time next to a surrogate parent during the first 2 weeks, Sandhill Crane chicks



Literature Cited

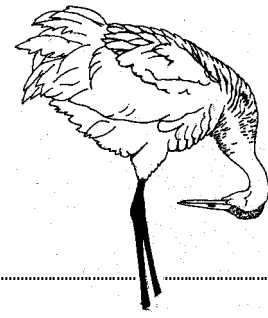
- Allen, R. P. 1952. The Whooping Crane. Research report No. 3. National Audubon Society, New York. 246 pp.
- Archibald, G. W. 1974a. Misty mornings in crane country. *Animal Kingdom* 77(2):19-24.
- Archibald, G. W. 1974b. Methods for breeding and rearing cranes in captivity. *International Zoo Yearbook* 14:147-155.
- Archibald, G. W. 1976a. Crane taxonomy as revealed by the unison call. Pages 225-251 in J. C. Lewis, editor. *Proceedings International Crane Workshop*. Oklahoma State University Printing Service, Stillwater.
- Archibald, G. W. 1976b. The unison call of cranes as a useful taxonomic tool. Ph.D. dissertation, Cornell University, Ithaca, New York. 167 pp.
- Archibald, G. W., and D. L. Viess. 1979. Crane propagation at the International Crane Foundation 1973-78. Pages 51-73 in J. C. Lewis, editor. *Proceedings 1978 Crane Workshop*. Colorado State University Printing Service, Fort Collins.
- Bateson, P. P. G. 1978. Early experience and sexual preferences. Pages 29-53 in J. B. Hutchison, editor. *Biological determinants of sexual behavior*. John Wiley & Sons, New York.
- Beall, F. 1985. *Regional (North American) Wattled Crane studbook, *Bugeranus carunculatus**. Baltimore Zoological Gardens, Md. 32 pp.
- Belتمان, R., and C. E. King. 1993. Captive management and husbandry of Red-crowned Cranes in Europe. *International Zoo News* 40(1):6-20.
- Bishop, M. A. 1984. Dynamics of subadult flocks of Whooping Cranes wintering in Texas, 1978-79 through 1982-83. M.S. thesis, Texas A & M University, College Station. 127 pp.
- Blauuw, F. E. 1897. A monograph of the cranes. E. F. Brill, Leiden, Netherlands. 64 pp.
- Derrickson, S. R., and J. W. Carpenter. 1980. Ethology and management of captive Sandhill Cranes. Pages 76-81 in *Annual Proceedings of the American Association of Zoo Veterinarians*.
- Derrickson, S. R., and J. W. Carpenter. 1987. Behavioral management of captive cranes—factors influencing propagation and reintroduction. Pages 493-511 in G. W. Archibald and R. F. Pasquier, editors. *Proceedings of the 1983 International Crane Workshop*. International Crane Foundation, Baraboo, Wis.
- Ellis, D. H. 1979. Development of behavior in the Golden Eagle. *Wildlife Monograph* No. 70. 94 pp.
- Ellis, D. H., G. W. Archibald, S. R. Swengel, and C. B. Kepler. 1991. Compendium of crane behavior. Part 1: individual (nonsocial). Pages 225-234 in J. T. Harris, editor. *Proceedings of the 1987 International Crane Workshop*. International Crane Foundation, Baraboo, Wis.
- Ellis, D. H., J. C. Lewis, G. F. Gee, and D. G. Smith. 1992a. Population recovery of the Whooping Crane with emphasis on reintroduction efforts: past and future. *Proceedings North American Crane Workshop* 6:142-150.
- Ellis, D. H., G. H. Olsen, G. F. Gee, J. M. Nicolich, K. E. O'Malley, M. Nagendran, S. G. Hereford, P. Range, W. T. Harper, R. P. Ingram, and D. G. Smith. 1992b. Techniques for rearing and releasing nonmigratory cranes: lessons from Mississippi Sandhill Crane program. *Proceedings North American Crane Workshop* 6:135-141.
- Ellis, D. H., S. R. Swengel, G. W. Archibald, and C. B. Kepler. *In prep.* A sociogram for the cranes of the world.
- Erickson, D., F. Boll, and R. Horwich. 1988. Raising crane. Videocassette. Ootek Production, Sauk City, Wis.
- Gaioni, C. C., H. S. Hoffman, and P. DePaulo. 1978. Imprinting in older ducklings: some tests of a reinforcement model. *Animal Learning and Behavior* 6:19-26.
- Gallagher, J. E. 1976. Sexual imprinting: the long term effects of various regimens of social experience on mate preference in Japanese Quail *Coturnix coturnix japonica*. *Behavior* 57:91-115.
- Gallagher, J. E. 1977. Sexual imprinting: a sensitive period in Japanese Quail (*Coturnix coturnix japonica*). *Journal of Comparative and Physiological Psychology* 91:72-78.
- Gottlieb, G. 1971. *Development of species identification in birds*. University of Chicago Press, Chicago, Ill. 176 pp.
- Guinness, F. E., M. J. Hall, and R. A. Cockerill. 1979. Mother-offspring association in red deer (*Cervus elaphus* L.) on Rhum. *Animal Behavior* 27:136-144.
- Hailman, J. P. 1967. The ontogeny of an instinct: the pecking response in chicks of the Laughing Gull (*Larus atricilla* L.). *Behavior Supplement* 15:1-159.
- Hartup, B. K., and R. H. Horwich. 1994. Early prenatal care and chick development in a cross-fostering trial with White-naped (*Grus vipio*) and Greater Sandhill (*Grus canadensis tabida*) Cranes. *Bird Behavior* 10:21-27.
- Hess, E. H. 1973. *Imprinting*. Van Nostrand Reinhold, New York. 472 pp.
- Hess, E. H., and S. B. Petrovich, editors. 1977. *Imprinting*. Dowden, Hutchinson and Ross, Stroudsburg, Pa.
- Hinde, R. A. 1961. The establishment of the parent-offspring relation in birds, with some mammalian analogies. Pages 175-19 in W. H. Thorpe and O. L. Zangwill, editors. *Current problems in animal behavior*. Cambridge University Press, Cambridge, England.
- Horwich, R. H. 1974. Regressive periods in primate behavioral development with reference to other mammals. *Primates* 15:141-149.
- Horwich, R. H. 1986. Reintroduction of Sandhill Cranes to the wild. *ICF Bugle* 12:1, 4-5.
- Horwich, R. H. 1987. Behavioral development in the Red-crowned Crane (*Grus japonensis*). *Zoo Biology* 6:379-389.
- Horwich, R. H. 1989. The use of surrogate parental models and age periods in a successful release of hand-reared Sandhill Cranes into the wild. *Zoo Biology* 8:379-390.
- Horwich, R. H., S. J. H. Cogswell, J. Burrows, and N. Mitchell. 1982. Seasonal variation in mother-daughter groupings in Siberian ibex (*Capra ibex siberica*). *Zoo Biology* 1:345-354.
- Horwich, R. H., and C. R. Owen. *Unpublished*. Imprinting stimuli for surrogate-reared Sandhill Crane chicks (*Grus canadensis*).

- Horwich, R. H., R. Van Dyke, and S. J. H. Cogswell. 1977. Regressive growth periods as a mechanism for herd formation in Siberian ibex (*Capra ibex*). *Zoologische Garten* 47:59-68.
- Horwich, R. H., J. Wood, and R. Anderson. 1992. Release of Sandhill Crane chicks hand-reared with artificial stimuli. Pages 255-261 in D. Wood, editor. Proceedings 1988 North American Crane Workshop. Florida Nongame Wildlife Program Technical Report #12.
- Immelmann, K. 1972. Sexual and other long-term aspects of imprinting in birds and other species. Pages 147-174 in D. S. Lehrman, R. A. Hinde, and E. Shaw, editors. *Advances in the study of behavior*. Vol. 4. Academic Press, New York.
- Jaynes, J. 1956. Imprinting: the interaction of learned and innate behavior. 1. Development and generalization. *Journal of Comparative and Physiological Psychology* 49(3):201-206.
- Johnson, D. N., and P. R. Barnes. 1991. The breeding biology of the Wattled Crane in Natal. Pages 377-386 in J. T. Harris, editor. Proceedings of the 1987 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Johnson, M. H., and G. Horn. Development of filial preferences in dark-reared chicks. *Animal Behavior* 36:675-683.
- Katz, B. 1979. Breeding ethology of the Hooded Crane. Pages 217-222 in J. C. Lewis, editor. Proceedings 1978 Crane Workshop. Colorado State University Printing Service, Fort Collins, Colo.
- Kepler, C. B. 1976. Dominance and dominance-related behavior in the Whooping Crane. Pages 177-196 in J. C. Lewis, editor. Proceedings International Crane Workshop. Oklahoma State University Publishing and Printing, Stillwater.
- Kepler, C. B. 1978. Captive propagation of Whooping Cranes: a behavioral approach. Pages 231-241 in S. A. Temple, editor. *Endangered birds: management techniques for preserving threatened species*. University of Wisconsin Press, Madison. 466 pp.
- Layne, J. N. 1981. Nesting, development of the young, and parental behavior of a pair of Florida Sandhill Cranes. *Florida Field Naturalist* 9:51-59.
- Littlefield, C. D. 1981. Mate-swapping of Sandhill Cranes. *Journal of Field Ornithology* 52:244-245.
- Littlefield, C. D. 1985. A record of preterritorial copulation by Sandhill Cranes. *Western Birds* 16:151.
- Littlefield, C. D., and R. A. Ryder. 1968. Breeding biology of the Greater Sandhill Crane on Malheur National Wildlife Refuge, Oregon. Pages 444-454 in Transactions of the 33rd North American Wildlife and natural resources conference. Wildlife Management Institute, Washington, D.C.
- Lorenz, K. Z. 1937. The companion in the bird's world. *Auk* 54:245-273.
- Lorenz, K. Z. 1970. *Studies in animal and human behavior*. Vol. 1. Translated by R. Martin. Harvard University Press, Cambridge, Mass. 403 pp.
- Mahan, T. A., and B. S. Simmers. 1992. Social preference of four cross-foster reared Sandhill Cranes. Proceedings North American Crane Workshop 6:114-119.
- Masatomi, H., and T. Kitagawa. 1975. Bionomics and sociology of Tancho or the Japanese Crane, *Grus japonensis*, II. Ethogram. *Journal of the Faculty of Science, Hokkaido University, Series VI, Zoology* 19(4):834-878.
- Mirande, C. M., and G. W. Archibald. 1990. Sexual maturity and pair formation of captive cranes at the International Crane Foundation. Pages 216-225 in Proceedings American Association of Zoological Parks and Aquariums 1990 Annual Conference.
- Mirande, C. M., J. W. Carpenter, and A. Burke. 1988 unpubl. The effect of disturbance on the reproduction and management of captive cranes. Paper presented at the 1988 North American Crane Workshop. 18 pp.
- Mitchell, L. C., and P. J. Zwank. 1987. Comparison of release methods for parent reared Mississippi Sandhill Cranes. Pages 399-409 in G. W. Archibald and R. F. Pasquier, editors. Proceedings of the 1983 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Nesbitt, S. A. 1989. The significance of mate loss in Florida Sandhill Cranes. *Wilson Bulletin* 101:648-651.
- Nesbitt, S. A., and G. W. Archibald. 1981. The agonistic repertoire of Sandhill Cranes. *Wilson Bulletin* 93:99-103.
- Nesbitt, S. A., and A. S. Wenner. 1987. Pair formation and mate fidelity in Sandhill Cranes. Pages 117-122 in J. C. Lewis, editor. Proceedings 1985 Crane Workshop. Platte River Whooping Crane Habitat Maintenance Trust and U.S. Fish and Wildlife Service, Grand Island, Nebr.
- Nievergelt, B. 1974. A comparison of rutting behavior and grouping in the Ethiopian and alpine ibex. Pages 324-340 in V. C. Geist and F. Walther, editors. *The behavior of ungulates and its relation to management*. International Union for the Conservation of Nature and Natural Resources, Morges, Switzerland.
- Poulsen, H. 1975. Agonistic behavior of two species of cranes. *Dansk Ornithologisk Forenings Tidsskrift* 69:119-122.
- Pratt, J. J. 1993. The Whooper-Sandhill Crane hybrid. *Grus Americana* 32(1):1.
- Ramsay, A. O. 1951. Familial recognition in domestic birds. *Auk* 68:1-15.
- Salzen, E. A., and C. C. Meyer. 1968. Reversibility of imprinting. *Journal of Comparative and Physiological Psychology* 66:269-275.
- Schaefer, H. H., and E. H. Hess. 1959. Color preferences in imprinting objects. *Zeitschrift fur Tierpsychologie* 16:161-172.
- Swengel, S. 1992. Sexual size dimorphism and size indices of six species of captive cranes at the International Crane Foundation. Proceedings North American Crane Workshop 6:151-158.
- Tacha, T. C. 1981. Behavior and taxonomy of Sandhill Cranes from mid-continental North America. Ph.D. dissertation, Oklahoma State University, Stillwater. 112 pp.
- Tinbergen, N., and A. C. Perdeck. 1950. On the stimulus situation releasing the begging response in the newly hatched Herring Gull chick (*Larus argentatus* Pont.). *Behavior* 3:1-39.
- Vidal, J. M. 1976. L'empreinte chez les animaux. *La Recherche* 63:24-35.

- Vidal, J. M. 1980. The relations between filial and sexual imprinting in the Domestic Fowl: effects of age and social experience. *Animal Behavior* 28:880-891.
- Voss, K. S. 1976. Ontogeny of behavior of the Greater Sandhill Crane. Pages 252-262 *in* J. C. Lewis, editor. *Proceedings International Crane Workshop*. Oklahoma State University Publishing and Printing, Stillwater.
- Voss, K. S. 1977. Agonistic behavior of the Greater Sandhill Crane. Pages 63-85 *in* R. D. Feldt, editor. *Eastern Greater Sandhill Crane Symposium*, October 24-26, 1977, Michigan City, Indiana.
- Walkinshaw, L. H. 1973a. *Cranes of the world*. Winchester Press, New York. 370 pp.
- Walkinshaw, L. H. 1973b. A history of Sandhill Cranes on the Haehnle Sanctuary, Michigan. *Jack-Pine Warbler* 51:54-74.



Reproductive Physiology



George F. Gee and Shirley E. Russman

Cranes differ physiologically from the more commonly studied avian species by exhibiting an **incomplete annual molt**, **low reproductive rate**, and **delayed sexual maturity**. Cranes appear to be especially **susceptible to stress** from physical and behavioral disturbance, unfamiliar territories, and disease. Although mature cranes normally reproduce annually, they may experience one or more years of reduced productivity apparently due to stress (Mirande et al. 1988 unpubl.). Unfortunately, little is known specifically about crane reproductive physiology; what little we do know indicates that cranes function like other birds.

In some ways most birds, including cranes, can tolerate environmental insults better than mammals. For example, some foods which are toxic to mammals provide good nutrition for birds. Breakdown products of food absorbed from the intestines are shunted through the liver and kidneys where **toxic products are removed** before reaching the general circulation (Sturkie 1954, 1986; Duke 1986). In another area, although cranes have long, exposed legs, they can withstand cold extremes by using **counter current circulation** in the tibiotarsus to conserve body heat and prevent hypothermia by warming blood returning from the feet (Whittow 1986). Unlike mammals, cranes (and all other birds) are better adapted to stave off dehydration because they excrete nitrogenous wastes as semi-solid **urates** instead of urea. This mechanism conserves body water that would be required to carry away the urea (Grimminger and Scanes 1986).

Cranes and swans possess a unique anatomical feature; the **trachea is coiled and posteriorly embedded** in the bony mass of the sternum (Pettingill 1970). The amount of tracheal coiling varies by species with the Gray Crowned Crane possessing the least coiled and the Japanese Crane the most highly coiled trachea. Some authors relate the coiled and elongated trachea to sound amplification, other authors believe it decreases skeletal mass, and still others relate it to thermoregulation (Prange et al. 1985; Gaunt et al. 1987).

In the following, we emphasize crane physiology. The references in Table 7.1 provide additional details on avian physiology.

Reproduction

Cranes generally mate for life (Walkinshaw 1973), but they do replace mates that die and some mate swapping has been reported (Littlefield 1981; E. Kuyt, Department of Environment, Edmonton, Canada, personal communication). Cranes begin laying when

TABLE 7.1

Avian physiology references.

Topic	Reference
Environmental, ecological	Farner et al. 1971
General, molt	Farner et al. 1972
Light, photorefractoriness	Farner et al. 1983
Incubation, domestic fowl	Landauer 1967
Molt	Lucas and Stettenheim 1972a, 1972b
Reproduction, behavior	Lofts and Murton 1973
General, light, reproduction	Marshall 1960
Season, reproduction, migration	Marshall 1961a, 1961b
Endangered species	Martin 1975
Reproductive cycles	Murton and Westwood 1977
Reproductive anatomy and physiology, domestic fowl	Nalbandov 1958
General, anatomy	Pettingill 1970
General, reproduction	Skutch 1976
Incubation	Stromberg 1975
Anatomy and physiology, domestic fowl	Sturkie 1986
Incubation, domestic fowl	Taylor 1949

3-5 years old (Walkinshaw 1973; S. A. Nesbitt, Florida Game and Fresh Water Fish Commission, Gainesville, Florida, personal communication), but there are exceptions in captivity and in the wild (see Chapter 3; Radke and Radke 1986).

Reproduction is controlled by an **integrated neuroendocrine system** (Kobayashi and Wada 1973; van Tienhoven 1980; El Halawani et al. 1982; Oksche 1983). To use an analogy, the system functions like a symphony orchestra. The brain and related neural systems conduct the orchestra and the endocrine organs, gonads, and accessory reproductive organs produce the music. Like the conductor's eyes and ears, the bird's visual, tactile, olfactory, auditory, and other neural sensory elements relay environmental and body conditions, so the brain, more specifically the hypothalamus and pituitary, can control reproductive functions.

Light entrains endogenous physiological rhythms, especially production and release of luteinizing hormone releasing hormone (LHRH), to time the reproductive effort (Wingfield et al. 1981; Wada 1984; Gwinner and Dittami 1985; Farner 1986; Hiatt et al. 1987). By their presence or absence, rainfall, humidity, and a variety of other **environmental and behavioral factors** can either induce or thwart reproduction (Marshall 1961a, 1961b; Ghosh and Banerjee 1983; Wingfield 1985; Vleck and Priedkalns 1985; Bluhm 1985a). Behavioral interactions (e.g., presence of mate or territorial encounters with conspecifics) lead to increased or decreased reproductive development (El Halawani et al. 1980, 1982; Ottinger 1983; Bluhm 1985b).

Feedback mechanisms on the central nervous system effecting control of reproduction include endocrine products from the **pituitary, target hormones** (from reproductive organs), and **non-target hormones**. **Other feedback factors** are less understood and include refractory periods, target tissue exhaustion, and entrainment of **endogenous rhythms**. The brain and pineal show obvious **endogenous rhythms** while helping to control the release of inhibiting or releasing factors in the hypothalamus (Kobayashi and Wada 1973; Gorbman et al. 1983; Ueck and Umar 1983). The hypothalamus, deep at the base of the brain, acts as the reproductive coordinator. The brain senses surrounding conditions, integrates information from the internal milieu, and uses neural and chemical pathways to signal the **hypothalamus** to turn on or off production of **releasing or inhibiting factors** (Wingfield 1980).

The hypothalamus is not only controlled by higher neural centers in the brain, but also through **feedback mechanisms** from within the rest of the body. The hypothalamus produces the following **releasing or inhibiting factors**: LHRH, thyrotropin-releasing hormone (TRH), prolactin-inhibiting factor (PIF), somatostatin, growth-releasing factor (GRF), and corticotropin-releasing factor (CRF) (Kobayashi and Wada 1973; El Halawani et al. 1982; Farner 1986). Table 7.2 provides the specific function, signs, and effects of the hypothalamic and pituitary hormones that control reproduction.

The **gonadotropins** are produced and released in response to LHRH (Bluhm et al. 1983; Bluhm 1985a) (Fig. 7.1, Cycle 1). The ratio of follicle stimulating hormone (FSH) to luteinizing hormone (LH) released from the pituitary in response to LHRH stimulation differs and is dependent on the circulating levels of gonadal steroids (androgens, estrogens, and progestins) (Cusick and Wilson 1972; Davies et al. 1976; Cheng and Balthazart 1982; Gorbman et al. 1983; Scanes 1986). In a **basic feedback mechanism**, **increased** plasma levels of **FSH and LH** lead to **increased gonadal steroid** levels, and eventually (Fig. 7.1, Cycle 3) the **elevated steroid levels decrease LHRH** release (Temple 1974; Pavgi and Chandola 1981; Lal and Thapliyal 1985; Groscolas et al. 1986; Sharp et al. 1986; Hiatt et al. 1987).

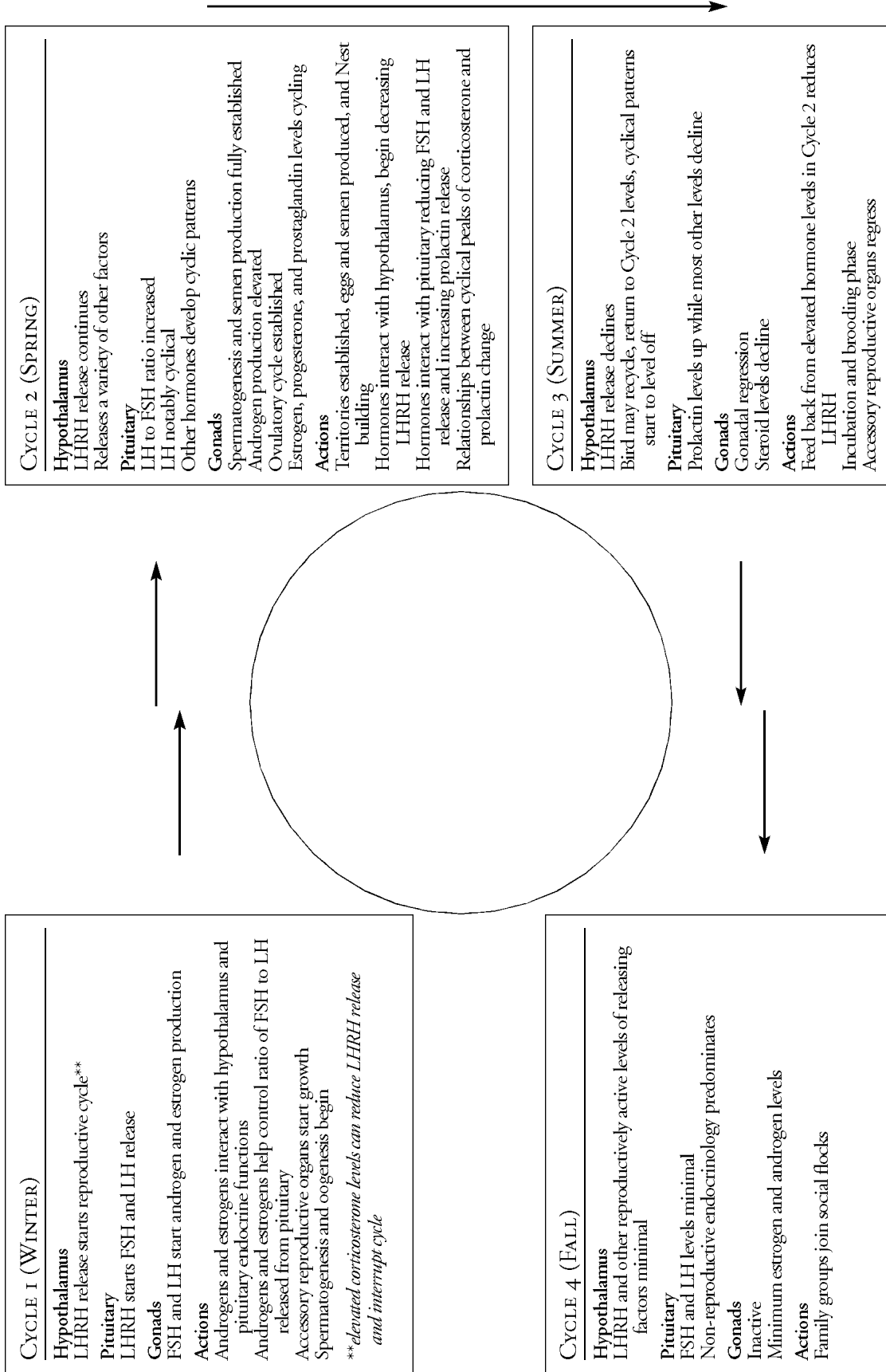
Somatostatin, which is secreted from the pancreas and small intestine, inhibits release of growth hormone and is an important component in carbohydrate metabolism. Arginine vasotocin and isoleucine oxytocin (mesotocin) are produced by **neurosecretory neurons** located in the hypothalamus. Vasotocin and oxytocin are stored in the pituitary (posterior lobe) before release (Kobayashi and Wada 1973; Gorbman et al. 1983).

Male

The reproductive system in both sexes of cranes **regresses in the summer and fall** and **develops** again the next **spring**. Generally, male cranes produce **semen** at least one year **earlier** than females produce eggs. For example, 80% of captive male Whooping Cranes (12 of 15) produced semen by three years of age, but only 13% of captive female Whooping Cranes (2 of 15) lay eggs by five years of age (Ellis et al. 1992). A healthy, well-fed crane may be **reproductively active** each breeding season for its **entire adult life** (60 or more years).

FIGURE 7.1

Hormonal Control of the Crane Reproductive Cycle*



* A simplified view, see standard avian reproductive physiology texts for more detail.

TABLE 7.2
Hypothalamic and pituitary control of reproduction.

HORMONE ¹	SOURCE	PRODUCTION EFFECTS	FUNCTION	PRODUCTION SIGNS
LHRH	hypothalamus	elevates LH, FSH and corticosterone; elevates gonadal steroids	adenohypophysis response dependent on circulating androgens, estrogens, and progestins. LHRH release dependent on temporal peaks of prolactin and corticosterone	control of seasonal reproduction gonad and accessory reproductive development
TRH	hypothalamus	stimulates thyrotropin, growth hormone, and prolactin	controls metabolic rate; promotes or inhibits reproductive development; stimulates growth hormone	thyroxin production can effect reproduction, effect in cranes unknown
PRF	hypothalamus	stimulates prolactin	release dependent on prolactin and serotonin levels	induces broodiness, stops egg production
PIF	hypothalamus	inhibits prolactin	role unclear in birds	—
Somatostatin	pancreas and small intestines	inhibits growth hormone	helps control carbohydrate metabolism	—
GRF	hypothalamus	stimulates growth hormone	controls growth	—
CRF	hypothalamus	stimulates corticosterone	seasonal cycling of diurnal release patterns	may help control onset and termination of breeding
Arginine vasotocin	hypothalamus neurosecretory neurons, stored neurohypophysis	—	interacts with prostaglandins; important in controlling water balance	—
isoleucine oxytocin (mesotocin)	hypothalamus neurosecretory neurons, stored neurohypophysis	—	same as above	oviposition
FSH	adeno-hypophysis	LHRH stimulates; response dependent on circulating levels of androgens, estrogens, and progestins	increase seasonal levels of androgens, estrogens, progestins, and prostaglandins	spermatogenesis or ovary growth, elevates gonadal steroids, initiates reproductive growth
LH	adeno-hypophysis	LHRH stimulates; response dependent on circulating levels of androgens, estrogens, and progestins	increased seasonal levels of androgens, estrogens, progestins, and prostaglandins	ovulation, gonadal steroid production (especially androgens and progestins), initiates reproduction
Thyrotropin	adeno-hypophysis	—	elevates growth hormone, elevates thyroxin	helps control metabolic rate, may affect crane reproduction

TABLE 7.2 CONTINUED

HORMONE ¹	SOURCE	PRODUCTION EFFECTS	FUNCTION	PRODUCTION SIGNS
prolactin	adeno-hypophysis	PRF elevates prostaglandin E, increases secretion rate; release stimulated by serotonin and decreased by prolactin levels	LHRH release dependent on temporal peaks of prolactin and corticosterone	induces broodiness, stops egg production
ACTH	adeno-hypophysis	elevated corticosterones response to stress, ovulation, mineral corticoids and glucocorticoids	—	integral part of seasonal reproduction, stunts growth
growth hormone	adeno-hypophysis	somatostatin, decreases GRF, increases serotonin	increases free fatty acids for energy, reduces glucose utilization	induces growth, inhibits reproduction
serotonin	adeno-hypophysis, GI tract, adrenal gland, thymus neurohormone of wide distribution	stress and digestion stimulate production	elevates prolactin (acting on pituitary), decreases growth hormone (acting on hypothalamus)	—
Thyroxin	thyroid	TRH elevates	inhibits or promotes gonadal development, may be species or sex dependent	reproduction effect in cranes unknown
Androgens	gonads	aggressive encounters elevate release; helps control ratio of FSH and LH released, LH stimulates release	aids estrogen effect in ovulation and reproductive cycling	determines female rank in dominance hierarchy; cyclical in egg formation
Estrogens	gonads, especially developing follicles	FSH and LH stimulates, especially LH	oviduct growth, maturation, and maintenance; helps control ratio of FSH and LH released; elevates progestins	deposition of body fat and medullary bones, secondary sexual characteristics, sexual behavior, ova developmental cycling
Progestins	growing follicles especially just before and after ovulation	stimulated by FSH and especially LH, stimulated by estrogen	growth of oviduct and albumin secretion	ova developmental cycling
Prosta-glandins	gonads, oviduct	vasotocin interaction unclear, elevated in follicle just before oviposition	induces muscle contractions, closely related to essential fatty acid metabolism	induces oviposition
Corti-costerone	adrenal	stress increases related	diurnal peaks part of ovulatory cycle, LHRH releases dependent on temporal peaks of prolactin and corticosterone	elevated levels interfere with reproduction

¹ LHRH = luteinizing hormone-releasing hormone, TRH = thyrotropin-releasing hormone, PRF = prolactin-releasing factor, PIF = prolactin inhibitory factor, GRF = growth hormone-releasing factor, CRF = corticotropin-releasing factor, FSH = follicle-stimulating hormone, LH = luteinizing hormone, ACTH = adrenocorticotropin.

FIGURE 7.2

Male reproductive tract.

Testes

Epididymis

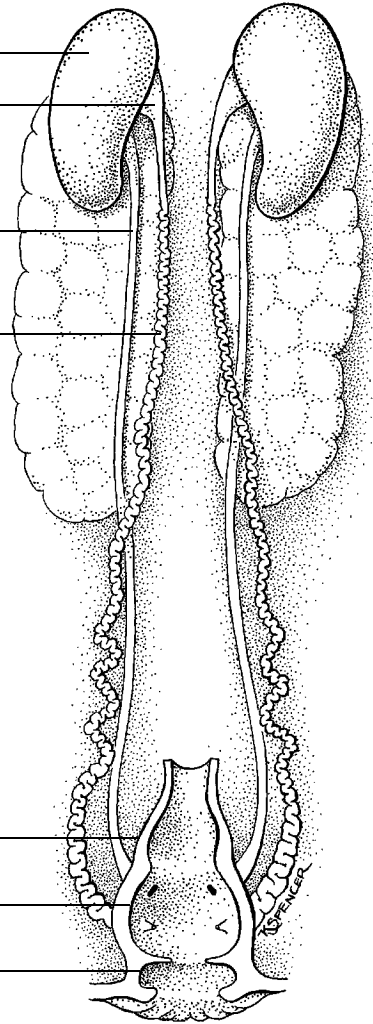
Ureter

Vas deferens

Coprodeum

Cloaca Urodeum

Proctodeum



Art Kate Spencer

The crane's reproductive tract (Fig. 7.2) resembles that of the domestic fowl but is larger (Johnson 1986a, 1986b). By locating the terminal papilla of the vas deferens in the urodeum (middle chamber of the cloaca), one can **sex** reproductively active cranes. Male cranes possess a rudimentary phallus and ejaculatory groove in the cloaca; therefore, **copulation** is completed by cloacal contact with the everted cloaca of the female (Gee 1983).

The crane stores **semen** in the distal vas deferens and releases it when stimulated. Components of the ejaculate (sperm, other cells, and lymph) come from the seminiferous tubules within the testes, the cells lining the vas deferens, and from the lymph folds in the cloaca. Cranes do not produce specialized **accessory seminal fluids** like some other birds (Quay 1967;

Lake 1981; King 1981). In the domestic chicken, spermatids may be released into the seminiferous tubule lumen early in its regeneration phase. After regeneration and early in the reproductive season, the seminiferous tubules in most birds are full of spermatids. Sperm cells mature from the spermatids and **accumulate** in the much convoluted **vas deferens** before ejaculation (Sturkie 1965; Lake 1981; Johnson 1986b). The entire process, from spermatid to sperm, takes 10 to 15 days in domestic fowl during the height of the reproductive season (Johnson 1986b). A spermatozoa takes one to four days to pass through the vas deferens (Munro 1938). In the Japanese Quail (*Coturnix coturnix japonica*), Jones and Jackson (1972) estimated that the entire process requires ca 25 days. For cranes, this duration is unknown.

FIGURE 7.3

Female reproductive anatomy (Florida Sandhill Crane)

Empty Follicle

Immature Yolk

Mature Yolk

Infundibulum (13cm)

Magnum (39cm)

Isthmus (18cm)

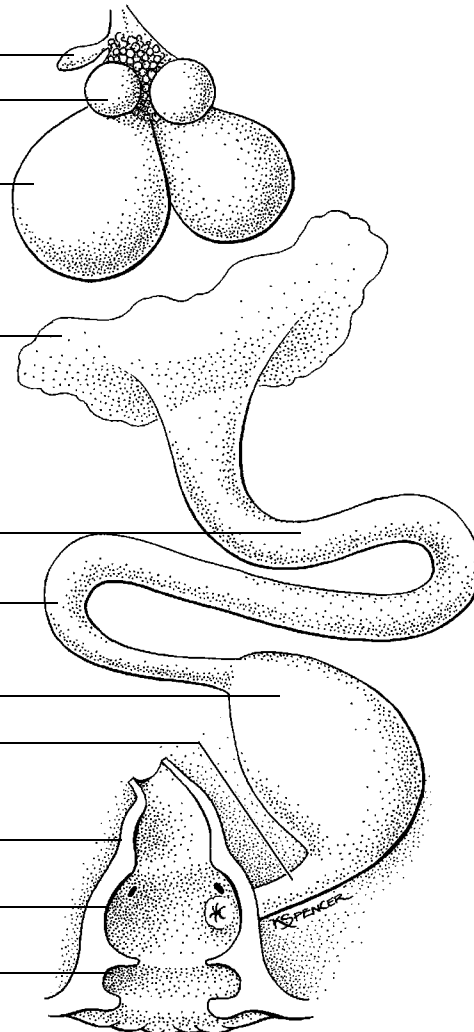
Uterus (Shell Gland) (17cm)

Vagina (5cm)

Coprodeum

Cloaca Urodeum

Proctodeum



Art Kate Spencer

Female

With increasing daylight in the spring, the **ovary** and **oviduct** develop on the left side of the peritoneal cavity (Fig. 7.3). The ovary contains thousands of oocytes and several developing ova. With the chicken, the mature oocyte, also known as the **yolk**, contains about 51% water, 33% lipid, and 16% protein (Johnson 1986a). As for all birds, crane ovaries contain more oocytes than will ever be needed. The developing **ovum** is highly vascularized with only the central stigma containing a lesser number of arteries and veins on the exterior surface. Just before ovulation, the stigma blanches (blood flow stops and the blood drains out of the area), and the stigma ruptures releasing the ovum. Only a few ova are developing

at any one time, and one is always larger than the rest (follicular hierarchy). Follicles that become yolk filled, but are not ovulated, are reabsorbed.

In chickens and cranes, the **egg production cycle** begins with the enlargement of several oocytes into ova (follicular maturation). In chickens, a descending size hierarchy is obvious in the largest three or four ova. The three largest follicles (F_1 - F_3) have different endocrinological responsibilities. The largest follicle (F_1) produces **progesterone** and **prostaglandins** before and immediately following ovulation (Hertel and yetal. 1974; Day and Nalbandov 1977; Poyser and Pharm 1981). The third largest follicle (F_3) produces the largest quantities of **estrogens** and releases the hormone in daily cycles to support the growth of the largest follicle (Huanget al. 1979; Kamiyohki and Tanaka 1983; Wang

and Bahr 1983; Johnson 1986a). The endocrinological functions of these second largest follicle (F_2) are intermediate between F_1 and F_3 . In cranes, which lay one to three egg clutches (Sijuade 1978; Wylie 1978; Blossom 1984; see Chapter 3 for details), these events may differ from poultry with large multi-egg clutches.

Each segment of the oviduct (see Johnson 1986a for details) serves a function in the process of fertilization and egg production. Sperm penetration of the ova occurs in the infundibulum and **fertilization** occurs in the magnum. The albumen is added while the egg passes through the **magnum**. The inner and outer shell membrane are added to the egg in the **isthmus** where (in the chicken) the embryo reaches the 4 to 8 cell stage (Patterson 1910; Olsen 1942; Eyal-Giladi and Kochav 1976; Kochav et al. 1980). The egg is completed in the **shell gland** or uterus where salts and water are added to expand the egg within the shell membranes. About three quarters of the time needed for egg formation is spent in the shell gland. In the chicken, the embryo has reached the blastocyst stage containing about 60,000 cells when the egg is laid.

Cranes take 3-4 days to **form eggs**. The period between ovulation and oviposition in Sandhill and Red Crowned Cranes is 55-60 **hours**. **Shell** formation takes about 45 **hours** and is equal to the rate of calcium deposition in chickens (about 250 mg per hour) (M. S. Putnam, University of Wisconsin, Madison, Wisconsin, personal communication). Cranes produce one to three eggs per clutch and may produce several clutches per season if the preceding clutches are lost. **The number** of eggs produced may be **increased** if eggs are **removed as laid**, but the clutch interval will be less predictable. Cranes will stop producing eggs if eggs are added to the nest (Lack 1933).

Endocrinology

A series of complex endocrinological and behavioral events **begins weeks before the production of eggs and semen** (Fig. 7.1, Cycle 1) (Haase et al. 1976; El Halawani et al. 1980, 1982). The sequence for cranes remains to be determined, but is probably similar to those in other birds. In reproductively active Sandhill Cranes, the gonad mass and size increase, and estrogen and testosterone levels rise in the spring. Gonad weight, testosterone and estrogen increase in nonproductive adults but less than in reproductively active cranes (Tacha et al. 1985). The sequence begins with

rising levels of LHRH (Fig. 7.1, Cycle 1) which stimulates the **release of gonadotropins** (FSH and LH) from the pituitary (Cheng and Balthazart 1982; Bluhm et al. 1983; Scanes et al. 1983; Silverin 1984; Bluhm 1985a; Scanes 1986). Thereafter, LHRH and other components of the reproductive cycle support reproductive development in both sexes (Fig. 7.1). LH controls gonadal secretion of estrogens and androgens (Nalbandov 1958; Murton and Westwood 1977; Maung and Follett 1978).

The actions of **estrogens and androgens** appear to be the result of interactions with environmental and social cues to induce proper sexual behavior (Cheng 1974; Hutchinson 1975; Slater 1978; Donham 1979; Dittami 1981; Akesson and Raveling 1983; Bluhm et al. 1983; Bluhm 1985a, 1985b). **Androgens**, produced primarily by the ovary, are important to the normal cyclic phenomenon **associated with egg production**. The **LH surge** associated with ovulation is always preceded by an androgen rise (van Tienhoven 1980; Kamiyoshi and Tanaka 1983; Johnson 1986a). Androgens may also be responsible for rank in a female dominance hierarchy (Hohn and Cheng 1967). Androgen levels in both sexes of some birds rise in response to aggressive and territorial encounters, while in other birds, it is the rising androgen levels which increase aggressive encounters (Adkins-Regan 1981; Akesson and Raveling 1983; Wada 1983; Wingfield 1984a, 1984b, 1985; Wingfield et al. 1987).

Progesterones are secreted by the follicles throughout the reproductive period. Secretions of progesterone by the preovulatory follicle increase from a low level two to three days before ovulation to a peak several hours after ovulation (Shahabi et al. 1975; van Tienhoven 1980; Bahr et al. 1983; Kamiyoshi and Tanaka 1983; Johnson 1986b). Progesterone is important to the **growth and function of the oviduct** and acts on the oviduct to **increase secretion of albumen**.

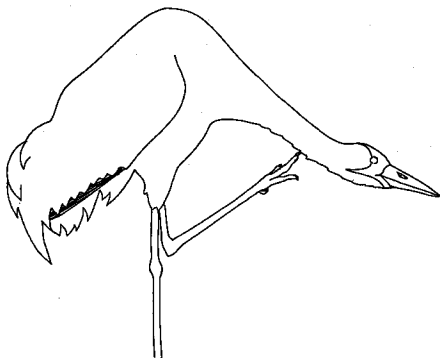
Plasma **prostaglandins and vasotocin** increase to their highest levels before **oviposition** (Day and Nalbandov 1977; Poyser and Pharm 1981; Johnson 1986a). Prostaglandins begin to rise in the preovulatory follicle about six hours before ovulation and continue to rise in the postovulatory follicle until after oviposition (van Tienhoven 1980; Poyser and Pharm 1981; Johnson 1986a). Exogenous doses of prostaglandins are potent stimulants of oviposition (Hertelendy et al. 1974; Hargrove and Ottinger 1991). Because many hormonal interactions in cranes are unknown, **hormone therapy is not recommended** without extensive laboratory support.

Incubation

Cranes need about four months to complete the nesting cycle. **Nest building** begins a few days before egg production in the female crane and may start even earlier in the male. The endocrine events leading to nest building include increasing levels of FSH and LH, estrogen, progesterone, and prolactin (Cheng 1974, 1979; Cheng and Balthazart 1982; Bluhm et al. 1983). Males and females may both build nests if in separate pens. Unmated wild birds may also build nests (Gee, personal observation). When given a choice of materials, male Sandhill Cranes at Patuxent build coarser, looser nests than females. Nest building often continues after egg laying if stimulated by rising water levels. Sometimes nests become extensive, floating platforms. However, if water levels rise too rapidly, Sandhill Cranes will abandon the submerged nest (P. W. Skyes, Jr., USFWS, Athens, Georgia, and S. A. Nesbitt, Florida Game and Fresh Water Fish Commission, Gainesville, Florida, personal communications).

Both sexes incubate the eggs (Walkinshaw 1965). In incubating birds **prolactin levels rise** and most of the other reproductive hormone levels decline. The metabolic clearance rate of some of the reproductive hormones may increase at the same time due to **increases in thyroid activity** (Jallageas and Assenmacher 1974; Kar and Chandola 1985). Although some feathers may be lost during the incubation period (see Molt section in this chapter), cranes do not develop a brood patch. Egg production normally stops after incubation begins, however, captive Sandhill Cranes that have been brooding chicks can resume egg laying if a chick is removed.

Crane reproductive physiology has received little study. Years of study are necessary to understand the unusual events that occur in captive crane colonies.



Molt

During **molt**, new feathers push out the old. First, **natal down** is replaced or covered by juvenal plumage. The **juvenal plumage** is replaced by **basic plumage** during the first fall and winter. Subsequent molts of the basic plumage follow each year throughout the bird's life. In any one year, feather molts can be complete (all feathers) or incomplete (affecting certain tracts or specific feathers) (Lucas and Stettenheim 1972a, 1972b; Noordhuis 1989).

In cranes, contour feathers emerge during the first two months. First, the primary remiges (large feathers of the hand) emerge, then the secondaries (large feathers on the ulna, elbow, and upper arm) and rectrices (tail), and within the next 2-3 weeks the body contour feathers grow, completing the juvenal plumage. The natal down clings to the tips of the emerging contour feathers for a few weeks before breaking off (Stephenson 1971). Young cranes can fly even before the remiges harden.

Some **immature** cranes need 2 or more **years to completely molt** all juvenal remiges (Lewis 1979; Gee 1981; Layne 1981; Nesbitt 1987; Kaschentseva 1988). However, the body contours and rectrices are probably replaced annually. In Greater Sandhill Cranes, all secondary remiges are replaced in 2.5-year-old birds and all primary remiges in 4.5-year-old birds (Lewis 1979; Gee 1981).

In breeding Whooping Cranes, molt of the basic remiges begins during the incubation period, and most are lost in one or a few days. Breeding Sandhill Cranes molt their remiges during incubation too, but molt may continue for two or more months. Some adult Sandhill Cranes, adult Whooping Cranes, and possibly other species may not complete their molt of remiges every year (Lewis 1979). Cranes do not respond to exogenous hormonal treatments (progesterone, thyroxine, and FSH) as do waterfowl, doves and domestic fowl (Payne 1972; Assenmacher and Jallageas 1980; van Tienhoven 1980; Dittami 1981; Gee 1981). However, there may be ways to induce molt using chemical agents or lights. There is some anecdotal evidence that a sudden reduction in artificial photoperiod can induce rapid and extensive molt (S. Haeffner, Denver Zoo, Denver, Colorado, personal communication). The induction of molt on demand would be a useful management tool, but will require more research.

External Factors Controlling Breeding

Crane reproduction can be influenced by a variety of factors such as stress, nutrition, disease, and photoperiod. Increasing or long photoperiod is probably the most influential factor and has been linked to avian reproductive periodicity since the late 1800's (Farner 1986). For reproductively active Sandhill Cranes, gonad weights and testosterone and estrogen levels show a positive correlation with photoperiod (Tacha et al. 1985). The natural photoperiod at temperate latitudes is sufficient to breed all species of cranes in captivity. However, cranes from northern latitudes held captive at temperate latitudes often breed more readily, earlier, and have greater fecundity if provided with an artificially lengthened photoperiod in the early spring (Gee and Pendleton 1992). By extending photoperiod in the spring in the mid-temperature latitude, we can bring together several environmental factors favoring reproduction (see Chapter 3).

A variety of **stressors**, such as disease, inclement weather, moves to new pens, intraspecific conflict, and human activities, can **interfere with the onset and maintenance of reproduction** (Mirande et al. 1988 unpubl.). In birds, disturbance can cause a variety of physiological and behavioral changes that result in stress (Marshall 1961a, 1961b; Ghosh and Banerjee 1983). The most conspicuous stress effect in birds is an elevation of plasma **corticosterone** levels (Siegel 1971; Dittami 1981; Deviche 1983; Dufty and Wingfield 1986) and a suppression of LHRH releases (Bluhm et al. 1983) which in turn disrupt the endocrine balance responsible for regeneration of the reproductive tract, ovulation and other reproductive functions (van Tienhoven 1980; Deviche 1983; Ghosh and Banerjee 1983). By contrast, cyclical variations of low levels of corticosterone are essential to the ovulatory cycle (Follett and Davies 1979; Peczely and Pethes 1980; van Tienhoven 1980; Peczely 1985). How stress affects the crane reproductive effort and how it can be accommodated needs more study.

Conclusions

Although the **general pattern** of avian physiology **applies** to cranes, we have identified many physiological mechanisms (e.g., effects of disturbance) that need further study. Studies with cranes are expensive compared to those done with domestic fowl because of the **crane's larger size**, **low reproductive rate**, and **delayed sexual maturity**.

To summarize, the crane reproductive system is composed of physiological and anatomical elements whose function is controlled by an **integrated neural-endocrine system**. Males generally produce semen at a younger age than when females lay eggs. Eggs are laid in clutches of two (1 to 3), and females will lay additional clutches if the preceding clutches are removed.

Both sexes build **nests and incubate** the eggs. **Molt** begins during incubation and body molt may be completed annually in breeding pairs. However, remiges are replaced sequentially over 2 to 3 years, or abruptly every 2 to 3 years in other species. Most immature birds replace their juvenal remiges over a 2 to 3 year period.

Stress interferes with reproduction in cranes by reducing egg production or terminating the reproductive effort. In other birds, stress elevates corticosterone levels and decreases LHRH release. We know little about the physiological response of cranes to stress.



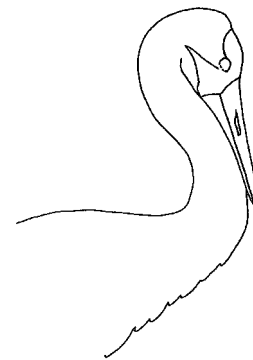
Literature Cited

- Adkins-Regan, E. 1981. Effect of sex steroids on the reproductive behavior of castrated male Ring Doves (*Streptopelia* sp.). *Physiological Behavior* 26:561-565.
- Akesson, T. R., and D. G. Raveling. 1983. Endocrine and behavioral correlates of nesting Canada Geese. *Canadian Journal of Zoology* 62:845-850.
- Assenmacher, I., and M. Jallageas. 1980. Adaptive aspects of endocrine regulations in birds. Pages 93-102 in S. Ishii et al., editors. *Hormones, adaptations, and evolution*. Japan Scientific Society Press, Tokyo/Springer-Verlag, Berlin.
- Bahr, J. M., S. C. Wang, M. Y. Huang, and F. O. Calvo. 1983. Steroid concentrations in isolated theca granulosa layers of preovulatory follicles during the ovulatory cycle of the domestic hen. *Biology of Reproduction* 29:326-334.
- Blossom, J. 1984. A breeding of the African Crowned Crane (*Balearica regulorum*). *Proceedings of Symposium of the Association of British Wild Animal Keepers* 9:11-19.
- Bluhm, C. K. 1985a. Seasonal variation in pituitary responsiveness to luteinizing hormone-releasing hormone of Mallards and Canvasbacks. *General and Comparative Endocrinology* 58:491-497.
- Bluhm, C. K. 1985b. Social factors regulating avian endocrinology and reproduction. Pages 247-264 in B. K. Follett, S. Ishii, and A. Chandola, editors. *The endocrine system and the environment*. Japan Scientific Society Press, Tokyo/Springer-Verlag, Berlin.
- Bluhm, C. K., R. E. Phillips, and W. H. Burke. 1983. Serum levels of luteinizing hormone, prolactin, estradiol, and progesterone in laying and nonlaying Mallards (*Anas platyrhynchos*). *Biology of Reproduction* 28:295-305.
- Cheng, M. F. 1974. Ovarian development in the female Ring Dove in response to stimulation by intact and castrated male Ring Doves. *Journal of Endocrinology* 63:43-53.
- Cheng, M. F. 1979. Progress and prospects in Ring Dove research: a personal view. Pages 97-129 in J. S. Rosenblatt, R. A. Hinde, C. Beer, M.-C. Busnel, editors. *Advances in the study of behavior*. Vol. 9. Academic Press, New York.
- Cheng, M. F., and J. Balthazart. 1982. The role of nest-building activity in gonadotrophin secretions and the reproductive success of Ring Doves (*Streptopelia risoria*). *Journal of Comparative Physiology* 96:307-324.
- Cusick, E. K., and F. E. Wilson. 1972. On control of spontaneous testicular regression in three sparrows (*Spezella arborea*). *General and Comparative Endocrinology* 19:441-456.
- Davies, D. J., Z. P. Goulden, B. K. Follett, and N. L. Brown. 1976. Testosterone feedback on luteinizing hormone (LH) secretion during a photoperiodically induced breeding cycle in Japanese Quail. *General and Comparative Endocrinology* 30:477-486.
- Day, S. L., and A. V. Nalbandov. 1977. Presence of prostaglandin F (PGF) in hen follicles and its physiological role in ovulation and oviposition. *Biology of Reproduction* 16:486-494.
- Deviche, P. 1983. Interactions between adrenal function and reproduction in male birds. Pages 243-254 in S. Mikami, K. Homma, and M. Wada, editors. *Avian endocrinology: environmental and ecological perspectives*. Japan Science Society Press, Tokyo/Springer-Verlag, Berlin.
- Dittami, J. P. 1981. Seasonal changes in the behavior and plasma titers of various hormones in Barheaded Geese, *Anserindicus*. *Zeitschrift fuer Tierpsychologie* 55:289-324.
- Donham, R. S. 1979. Annual cycle of plasma luteinizing hormone and sex hormones in male and female Mallards (*Anas platyrhynchos*). *Biology of Reproduction* 21:1273-1285.
- Dufty, A. M., Jr., and J. C. Wingfield. 1986. Temporal patterns of circulating LH and steroid hormones in a brood parasite, the Brown-headed Cowbird, *Nolothrus ater*. I. Males. *Journal of Zoology, London* 208:191-203.
- Duke, G. E. 1986. Alimentary canal: secretion and digestion, special digestive functions and absorption. Pages 289-302 in P. D. Sturkie, editor. *Avian physiology*. 4th ed. Springer-Verlag, New York.
- El Halawani, M. E., W. H. Burke, and L. A. Ogren. 1980. Involvement of catecholaminergic mechanisms in the photoperiodically induced rise in serum luteinizing hormone of Japanese Quail (*Coturnix coturnixjaponica*). *General and Comparative Endocrinology* 41:14-21.
- El Halawani, M. E., W. H. Burke, P. T. Dennison, and J. L. Silsby. 1982. Neuropharmacological aspects of neural regulation of avian endocrine function. Pages 33-40 in C. G. Scanes, M. A. Ottinger, A. D. Kennyh, O. Balthazart, J. Cronshaw, and I. C. Jones, editors. *Aspects of avian endocrinology: practical and theoretical implications*. Graduate Studies, Vol. 26, Texas Tech University, Lubbock.
- Ellis, D. H., J. C. Lewis, G. F. Gee, and D. G. Smith. 1992. Population recovery of the Whooping Crane with emphasis on reintroduction efforts: past and future. *Proceedings North American Crane Workshop* 6:142-150.
- Eyal-Giladi, H., and S. Kochav. 1976. From cleavage to primitive steak formation: a complementary table and a new look at the first stage of development of the chick. I. General morphology. *Developmental Biology* 49:321-337.
- Farner, D. S. 1986. Generation and regulation of annual cycle in migratory passerine birds. *American Zoologist* 26:493-501.
- Farner, D. S., J. R. King, and K. C. Parkes. 1971. *Avian biology*. Vol. I. Academic Press, New York. 586 pp.
- Farner, D. S., J. R. King, and K. C. Parkes. 1972. *Avian biology*. Vol. II. Academic Press, New York. 612 pp.
- Farner, D. S., R. S. Donham, K. S. Matt, P. W. Mattocks, Jr., M. C. Moore, and J. C. Wingfield. 1983. The nature of photorefractoriness. Pages 149-166 in S. Mikami, K. Homma, and M. Wada, editors. *Avian endocrinology: environmental and ecological perspectives*. Japan Scientific Society Press, Tokyo/Springer-Verlag, Berlin.
- Follett, B. K., and D. T. Davies. 1979. The endocrine control of ovulation in birds. Pages 323-344 in H. W. Hawk, C. A. Kiddy, and H. C. Cecil, editors. *Animal Reproduction, Beltsville Symposia in Agricultural Research*, 3. John Wiley & Sons, New York.

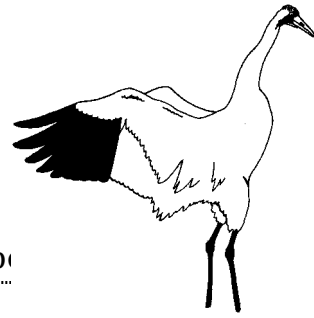
- Gaunt, A. S., S. L. L. Gaunt, H. D. Prange, and J. S. Wasser. 1987. The effects of tracheal coiling on the vocalizations of cranes (Aves: Gruidae). *Journal of Comparative Physiology A* 161:43-58.
- Gee, G. F. 1981. Hormonal treatment and flight feather molt in immature Sandhill Cranes. Pages 237-244 *in* R. Drewien and J. C. Lewis, editors. *Proceeding 1981 Crane Workshop*. National Audubon Society, Tavernier, Fla.
- Gee, G. F. 1983. Avian artificial insemination and semen preservation. Pages 375-398 *in* Jean Delacour/IFCB. *Symposium on breeding birds in captivity*, North Hollywood, Calif.
- Gee, G. F., and G. W. Pendleton. 1992. Effects of extended photoperiod on Sandhill Crane reproduction. Pages 149-159 *in* D. A. Wood, editor. *Proceedings 1988 North American Crane Workshop*. Florida Nongame Wildlife Program Technical Report #12.
- Ghosh, A., and J. Banerjee. 1983. Effect of population stress on the histophysiology of avian endocrine organs. *Journal Yamashima Institute Ornithology* 15:156-166.
- Gorbman, A., W. W. Dickhoff, S. R. Vigna, N. B. Clark, and C. L. Ralph. 1983. *Comparative endocrinology*. John Wiley & Sons, New York. 572 pp.
- Grimminger, P., and C. S. Scanes. 1986. Protein metabolism. Pages 326-344 *in* P. D. Sturkie, editor. *Avian physiology*. 4th ed. Springer-Verlag, New York.
- Groscolas, R., M. Jallageas, A. Goldsmith, and I. Assenmacher. 1986. The endocrine control of reproduction and molt in male and female Emperor (*Aptenodytes forsteri*) and Adelle (*Pygoscelis adeliae*) Penguins. 1. Annual changes in plasma levels of gonadal steroids and LH. *General and Comparative Endocrinology* 62:43-53.
- Gwinner, E., and J. Dittami. 1985. Photoperiodic responses in temperate zone and equatorial Stonechats: a contribution to the problem of photoperiodism in tropical organisms. Pages 279-294 *in* B. K. Follett, S. Ishii, and A. Chandola, editors. *The endocrine system and the environment*. Japan Scientific Society Press, Tokyo/Springer-Verlag, Berlin.
- Haase, E., E. Paulke, and P. J. Sharp. 1976. Effects of seasonal and social factors on testicular activity and hormone levels in domestic pigeons. *Journal of Experimental Zoology* 197:81-88.
- Hargrove, T. L., and M. A. Ottinger. 1991. Induced oviposition of precalcified eggs following prostaglandin administration. *Poultry Science* 71:548-552.
- Hertelendy, F., M. Yeh, and H. V. Biellier. 1974. Induction of oviposition in the domestic hen by prostaglandins. *General and Comparative Endocrinology* 22:529-531.
- Hiatt, E. S., A. R. Goldsmith, and D. S. Farner. 1987. Plasma levels of prolactin and gonadotrophins during the reproductive cycle of White-crowned Sparrows (*Zonotrichia leucophrys*). *Auk* 104:208-217.
- Hohn, E. O., and S. C. Cheng. 1967. Gonadal hormones in Wilson's Phalarope (*Steranapastricolor*) and other birds in relation to plumage and sex behavior. *General and Comparative Endocrinology* 8:1-11.
- Huang, E. S. R., K. J. Kao, and A. V. Nalbandov. 1979. Synthesis of sex steroids by cellular components of chicken follicles. *Biology of Reproduction* 20:454-460.
- Hutchinson, R. E. 1975. Influence of oestrogen on the initiation of nesting behavior in female Budgerigars. *Journal of Endocrinology* 64:417-428.
- Jallageas, M., and I. Assenmacher. 1974. Thyroid gonadal interactions in the male domestic duck in relationship with the sexual cycle. *General and Comparative Endocrinology* 22:13-20.
- Johnson, A. L. 1986a. Reproduction in the female. Pages 403-431 *in* P. D. Sturkie, editor. *Avian physiology*. 4th ed. Springer-Verlag, New York.
- Johnson, A. L. 1986b. Reproduction in the male. Pages 432-451 *in* P. D. Sturkie, editor. *Avian physiology*. 4th ed. Springer-Verlag, New York.
- Jones, P., and H. Jackson. 1972. Estimation of the duration of spermatogenesis in Japanese Quail, using antispermatogonial chemicals. *Journal Reproduction and Fertility* 31:319-322.
- Kamiyoshi, M., and K. Tanaka. 1983. Endocrine control of ovulatory sequence in domestic fowl. Pages 167-177 *in* S. Mikami, K. Homma, and M. Wada, editors. *Avian endocrinology: environmental and ecological perspectives*. Japan Scientific Society Press, Tokyo/Springer-Verlag, Berlin.
- Kar, A., and A. Chandola. 1985. Seasonality in birds and reptiles: the involvement of thyroxine and triiodothyroxine. Pages 117-126 *in* B. K. Follett, S. Ishii, and A. Chandola, editors. *The endocrine system and the environment*. Japan Scientific Society Press, Tokyo/Springer-Verlag, Berlin.
- Kaschentseva, T. A. 1988. Pterilography of cranes. Pages 120-131 *in* N. M. Litvinenko and I. A. Neufeldt, editors. *The Palearctic cranes*. USSR Academy of Sciences, Vladivostok. [In Russian].
- King, A. S. 1981. Cloaca. Pages 63-105 *in* A. S. King and J. McLelland, editors. *Form and function in birds*. Vol. 2. Academic Press, New York.
- Kobayashi, H., and M. Wada. 1973. Neuroendocrinology in birds. Pages 287-347 *in* D. S. Farner, J. R. King, and K. C. Parkes, editors. *Avian biology*. Vol. III. Academic Press, New York.
- Kochav, S., M. Ginsburg, and A. Eyal-Giladi. 1980. From cleavage to primitive streak formation: a complementary normal table and a new look at the first stages of development of the chick. II. Microscopic anatomy and cell population dynamics. *Developmental Biology* 79:296-308.
- Lack, D. 1933. Nesting conditions as a factor controlling breeding times in birds. *Proceedings of the Zoological Society of London* 1933:231-237.
- Lake, P. E. 1981. Male genital organs. Pages 1-61 *in* A. S. King, and J. McLelland, editors. *Form and function in birds*. Vol. 2. Academic Press, New York.
- Lal, P., and J. P. Thapliyal. 1985. Photorefractoriness in migratory Red-headed Bunting (*Emberiza buniceps*). Pages 137-148 *in* B. K. Follett, S. Ishii, and A. Chandola, editors. *The endocrine system and the environment*. Japan Scientific Society Press, Tokyo/Springer-Verlag, Berlin.
- Landauer, W. 1967. The hatchability of chicken eggs as influenced by environment and heredity. *Monograph 1 (Revised)*. Storrs Agricultural Experiment Station, University of Connecticut, Storrs. 15 pp.

- Layne, J. N. 1981. Observations on wing molt of Florida Sandhill Cranes. *Florida Field Naturalist* 9:60-61.
- Lewis, J. C. 1979. Molt of the remiges of *Grus canadensis*. Pages 255-259 *in* J. C. Lewis, editor. *Proceeding 1978 Crane Workshop*. Colorado State University Printing Service, Fort Collins.
- Littlefield, C. D. 1981. Mate-swapping of Sandhill Cranes. *Journal of Field Ornithology* 52:244-245.
- Lofts, B., and R. K. Murton. 1973. Reproduction in birds. Pages 1-107 *in* D. S. Farner, J. R. King, and K. C. Parkes, editors. *Avian biology*. Vol. III. Academic Press, New York.
- Lucas, A. M., and P. R. Stettenheim. 1972a. Molts and plumages of domestic chickens. Pages 197-233 *in* A. M. Lucas and P. R. Stettenheim, editors. *Avian anatomy, Part I. Agriculture Handbook 362*. Agricultural Research Service, U.S. Department of Agriculture, in cooperation with Michigan Agricultural Experiment Station, Michigan State University, East Lansing.
- Lucas, A. M., and P. R. Stettenheim, editors. 1972b. *Avian anatomy, Part II. Agriculture Handbook 362*. Agricultural Research Service, U.S. Department of Agriculture, in cooperation with Michigan Agricultural Experiment Station, Michigan State University, East Lansing. 750 pp.
- Marshall, A. J., editor. 1960. *Biology and comparative physiology of birds*. Vol. I. Academic Press, New York. 518 pp.
- Marshall, A. J. 1961a. Breeding seasons and migration. Pages 307-339 *in* A. J. Marshall, editor. *Biology and comparative physiology of birds*. Vol. II. Academic Press, New York.
- Marshall, A. J. 1961b. Reproduction. Pages 169-213 *in* A. J. Marshall, editor. *Biology and comparative physiology of birds*. Vol. II. Academic Press, New York.
- Martin, R. D., editor. 1975. *Breeding endangered species in captivity*. Academic Press, New York. 420 pp.
- Maung, S. L., and B. K. Follett. 1978. The endocrine control by luteinizing hormone of testosterone secretion from the testis of the Japanese Quail. *General and Comparative Endocrinology* 36:79-89.
- Mirande, C. M., J. W. Carpenter, and A. Burke. 1988 unpubl. The effect of disturbance on the reproduction and management of captive cranes. Paper presented at the 1988 North American Crane Workshop. 18 pp.
- Munro, S. S. 1938. Functional changes in fowl sperm during their passage through the excurrent ducts of the male. *Journal of Experimental Zoology* 79:71-92.
- Murton, R. K., and N. J. Westwood. 1977. *Avian breeding cycles*. Clarendon Press, Oxford. 594 pp.
- Nalbandov, A. V. 1958. *Reproductive physiology: comparative reproductive physiology of domestic animals, laboratory animals, and man*. W. H. Freeman, San Francisco. 271 pp.
- Nesbitt, S. A. 1987. A technique for aging Sandhill Cranes using wing molt—preliminary finding. Pages 224-229 *in* J. C. Lewis, editor. *Proceedings 1985 Crane Workshop*. Platte River Whooping Crane Habitat Maintenance Trust, Grand Island, Nebr.
- Noordhuis, R. 1989. Patterns of primary molt: ecophysiological adaptations. *Limosa* 62:35-45.
- Olsen, M. W. 1942. Maturation, fertilization and early cleavage in the hen's eggs. *Journal Morphology* 70:513-533.
- Oksche, A. 1983. Reflections on the structural basis of avian neuroendocrine systems. Pages 3-10 *in* S. Mikami, K. Homma, and M. Wada, editors. *Avian endocrinology: environmental and ecological perspectives*. Japan Scientific Society Press, Tokyo/Springer-Verlag, Berlin.
- Ottinger, M. A. 1983. Hormonal control of reproductive behavior in the avian male. *Poultry Science* 62:1690-1699.
- Patterson, J. T. 1910. Studies on the early development of the hen's egg. I. History of early cleavage and of the accessory cleavage. *Journal of Morphology* 21:101-134.
- Pavgi, S. S. N., and A. Chandola. 1981. Role of gonadal feedback in annual reproduction of the Weaver Bird: interaction with photoperiod. *General and Comparative Endocrinology* 45:521-526.
- Payne, R. B. 1972. Mechanisms and control of molt. Pages 103-155 *in* D. S. Farner, J. R. King, and K. C. Parks, editors. *Avian biology*. Vol. II. Academic Press, New York.
- Peczely, P. 1985. Adrenocortical adaptations in birds. Pages 213-220 *in* B. K. Follett, S. Ishii, and A. Chandola, editors. *The endocrine system and the environment*. Japan Scientific Society Press, Tokyo/Springer-Verlag, Berlin.
- Peczely, P., and G. Pethes. 1980. Plasma corticosterone, thyroxine and triiodothyroxine level in Collared Dove (*Streptopelia decaocto*) during the reproductive cycle. *ACTA Physiologica Academiae Scientiarum Hungaricae* 56:421-430.
- Pettingill, O. S., Jr. 1970. *Ornithology in laboratory and field*. 4th ed. Burgess Publishing, Minneapolis, Minn. 524 pp.
- Poyser, N. L., and B. Pharm. 1981. *Prostaglandins in reproduction*. Research Studies Press, New York. 260 pp.
- Prange, H. D., J. S. Wasser, A. S. Gaunt, and S. L. L. Gaunt. 1985. Respiratory responses to acute heat stress in cranes (Gruidae): the effects of tracheal coiling. *Respiration Physiology* 62:95-103.
- Quay, W. B. 1967. Comparative survey of the anal glands of birds. *Auk* 84:379-389.
- Radke, M. F., and W. R. Radke. 1986. Breeding by a two-year old Sandhill Crane. *Western Birds* 17:192-193.
- Scanes, C. G. 1986. Pituitary gland. Pages 383-402 *in* P. D. Sturkie, editor. *Avian physiology*. 4th ed. Springer-Verlag, New York.
- Scanes, C. G., T. J. Lauterio, and F. C. Buonomo. 1983. Annual, developmental, and diurnal cycles of pituitary hormone secretion. Pages 307-326 *in* S. Mikami, K. Homma, and M. Wada, editors. *Avian endocrinology: environmental and ecological perspectives*. Japan Scientific Society Press, Tokyo/Springer-Verlag, Berlin.
- Shahabi, N. A., H. W. Norton, and A. V. Nalbandov. 1975. Steroid levels in follicles and the plasma of hens during the ovulatory cycle. *Endocrinology* 96:962-968.
- Sharp, P. J., H. Klandorf, and A. S. McNeilly. 1986. Plasma prolactin, thyroxine, triiodothyroxine, testosterone, and luteinizing hormone during a photoinduced reproductive cycle in Mallard drakes. *Journal of Experimental Zoology* 238:409-413.
- Siegel, H. S. 1971. Adrenals, stress, and the environment. *World's Poultry Science Journal* 27:327-349.
- Sijuade, A. A. 1978. Breeding the West African Crowned Crane in captivity. *International Zoo Yearbook* 18:103-105.

- Silverin, B. 1984. Annual gonadotrophin and testosterone cycles in free-living male birds. *Journal of Experimental Zoology* 232:581-587.
- Skutch, A. 1976. *Birds and their young*. University of Texas Press, Austin. 503 pp.
- Slater, P. J. B. 1978. Sex hormones and behavior. Institute of Biology. Studies in Biology No. 103. E. Arnold Publisher, London. 58 pp.
- Stephenson, J. D. 1971. Plumage development and growth of young Whooping Cranes. M.S. thesis, Oregon State University, Corvallis. 56 pp.
- Stromberg, J. 1975. *A guide to better hatching*. Stromberg Publishing Co., Fort Dodge, Iowa. 100 pp.
- Sturkie, P. D. 1954. Alimentary canal: anatomy, prehension, deglutition, passage of ingesta, motility. Pages 151-163 *in* Avian physiology. Comstock/Cornell University Press, Ithaca, New York.
- Sturkie, P. D. 1965. *Avian physiology*, 2nd ed. Comstock/Cornell University Press, Ithaca, New York. 766 pp.
- Sturkie, P. D. 1986. *Avian physiology*, 4th ed. Springer-Verlag, New York. 516 pp.
- Tacha, T. C., P. A. Vohs, G. C. Iverson, and J. A. Bantle. 1985. Reproductive physiology of Sandhill Cranes during winter and spring. Pages 245-248 *in* J. C. Lewis, editor. Proceedings 1985 Crane Workshop. Platte River Whooping Crane Habitat Maintenance Trust and U. S. Fish and Wildlife Service, Grand Island, Nebr.
- Taylor, L. W., editor. 1949. *Fertility and hatchability of chicken and turkey eggs*. John Wiley & Sons, New York. 423 pp.
- Temple, S. A. 1974. Plasma testosterone during the annual reproductive cycle of Starlings (*Sturnus vulgaris*). *General and Comparative Endocrinology* 22:470-479.
- Ueck, M., and H. Umar. 1983. Environmental, neural, and endocrine influences on the parenchyma of the avian pineal organ and its various responses. Pages 201-215 *in* S. Mikami, K. Homma, and M. Wada, editors. Avian endocrinology: environmental and ecological perspectives. Japan Scientific Society Press, Tokyo/Springer-Verlag, Berlin.
- van Tienhoven, A. 1980. Neuroendocrinology of avian reproduction, with special emphasis on the reproductive cycle of the fowl (*Gallusdomesticus*). *World's Poultry Science Journal* 37:156-176.
- Vleck, C. M., and J. Priedkalns. 1985. Reproduction in Zebra Finches: hormone levels and effect of dehydration. *Condor* 87:37-46.
- Wada, M. 1983. Environmental cycles, circadian clock, and androgen-dependent behavior in birds. Pages 191-200 *in* S. Mikami, K. Homma, and M. Wada, editors. Avian endocrinology and ecological perspectives. Japan Scientific Society Press, Tokyo/Springer-Verlag, Berlin.
- Wada, M. 1984. Photoperiodism in avian reproduction. Pages 101-113 *in* K. Ochiai, Y. Arai, T. Shioda, and M. Takahashi, editors. Endocrine correlates of reproduction. Japan Scientific Society Press, Tokyo/Springer-Verlag, Berlin.
- Walkinshaw, L. H. 1965. Attentiveness of cranes at their nests. *Auk* 82:465-476.
- Walkinshaw, L. H. 1973. *Cranes of the world*. Winchester Press, New York. 370 pp.
- Wang, S. C., and J. M. Bahr. 1983. Estradiol secretion by theca cells of the domestic hen during the ovulatory cycle. *Biology of Reproduction* 26:618-621.
- Whittow, G. C. 1986. Regulation of body temperature. Pages 221-252 *in* P. D. Sturkie, editor. Avian physiology. 4th ed. Springer-Verlag, New York.
- Wingfield, J. C. 1980. Fine temporal adjustment of reproductive functions. Pages 367-389 *in* A. Eppl and M. H. Stetson, editors. Avian endocrinology. Academic Press, New York.
- Wingfield, J. C. 1984a. Androgens and mating systems: testosterone-induced polygamy in normal monogamous birds. *Auk* 101:665-671.
- Wingfield, J. C. 1984b. Environmental and endocrine control of reproduction in the Song Sparrow, *Melospiza melodia*. II. Agonistic interactions as environmental information stimulating secretion of testosterone. *General and Comparative Endocrinology* 56:417-424.
- Wingfield, J. C. 1985. Environmental and endocrine control of territorial behavior in birds. Pages 265-277 *in* B. K. Follett, S. Ishii, and A. Chandola, editors. The endocrine system and the environment. Japan Scientific Society Press, Tokyo/Springer-Verlag, Berlin.
- Wingfield, J. C., C. M. Vleck, and D. S. Farner. 1981. Effect of day length and reproductive state on diel rhythms of luteinizing hormone levels in the plasma of White-crowned Sparrows, *Zonotrichia leucophrys gambelii*. *Journal of Experimental Zoology* 217:261-264.
- Wingfield, J. C., G. F. Ball, A. M. Dufty, Jr., R. E. Hegner, and M. Ramenofsky. 1987. Testosterone and aggression in birds. *American Scientist* 75:602-608.
- Wylie, S. R. 1978. A review of techniques in the maintenance and propagation of cranes. Pages 15-25 *in* Proceedings First International Birds in Captivity Symposium, Seattle, Wash.



Medicine and Surgery



Glenn H. Olsen, James W. Carpenter, and Julia A. Langenb

Success in captive rearing and propagation of cranes is dependent on establishing appropriate health monitoring, disease prevention, and parasite control procedures. A knowledge of common diseases, surgical procedures, anesthesia, and clinical pathology is crucial.

Initial Examination

History

Before the crane is removed from its pen or enclosure, the veterinarian should obtain a complete history. Questions asked should range from general information such as age, sex, and length of time in the collection, to those more specific, including:

1. Current clinical signs of disease including unusual behavior.
2. Medical treatment given to date.
3. History of clinical signs in the crane flock.
4. Recent changes in management procedures or in the crane's environment.
5. Unusual behavior (see Chapter 6): listlessness, reluctance to fly or move wings, lack of balance, restlessness, lameness or limping, straining, ruffled feathers, head held low, shivering, fainting, eyes partially closed, regurgitation, reluctance to rise, etc.
6. Changes in food and water consumption.
7. Appearance and consistency of droppings.
8. Overt signs of trauma or swelling in extremities.
9. Bleeding.

Physical Examination

The physical examination should be thorough but brief (i.e., normally not exceeding 10 min). A checklist (Fig. 10.5) and an assistant help assure thoroughness. Even when an obvious injury is present, a complete examination should be performed to detect less

obvious problems or complications. A crane in critical condition should, of course, receive immediate treatment and be given a thorough examination later.

Head and Eyes. Restrain the crane (as described in Chapter 2). Examine the eye area for swollen lids, squinting, discharge, or a change in color of the globe. These changes can be due to injury, infection, foreign bodies under the lids, or swollen sinuses. Dilated pupils may indicate shock, concussion, or blindness. Hemorrhage in the anterior chamber or ear canal can be due to trauma to the head. A small light source is used to check pupillary response. In birds, pupils respond independently (i.e., no consensual reflex as in most mammals).

If abnormalities are noted or suspected during the initial superficial eye examination, the deeper structures of the eye should be examined using an ophthalmoscope. Because birds have striated rather than smooth muscle in the iris and ciliary body, atropine will not cause pupil dilation as in the mammalian eye. Iris color in Sandhill Cranes is blue in young chicks, changing to gray or gray-green, and finally yellow or orange as the bird matures. The retina is normally avascular. A dark, ribbon-like structure called the pecten is attached to the retina. The pecten is highly vascular and is considered to be the source of oxygen and nutrition for the retina.

Beak. The beak should be examined for bite and overgrowth. The sides of the beak should be checked for evenness of wear. Crane beaks grow several centimeters per year. As a result, some deformed beaks require frequent trimming (every 2-4 months). Beak trimming may also be required for non-deformed beaks during the winter when normal probing behavior is inhibited by frozen ground. When trauma is suspected, the beak should be palpated for fracture or other damage. The nares should be examined for any discharge or plugs.

Mouth. Often during the examination, the crane will open its mouth to vocalize, allowing a view of the structures inside. If not, the mouth can be opened by gently prying at the commissure with the index finger on one side and the thumb on the other side.

Normally, the mucous membranes are bright pink, but some cranes have gray or black pigmented tissues on the mucous membranes. Level of hydration can be estimated based on moistness of the mucous membranes. The tongue should be long and thin. There is a small, bright red structure at the tracheal opening (glottis) in Sandhill and Whooping Cranes.

Vitamin A deficiency can appear as proliferative, plaque-like lesions of the epithelium of the alimentary mucosa, conjunctiva, eyelids, ear canal, or skin. Protozoan (*Trichomonas*) and fungal (*Candida*) infections will occur as thick white, raised, plaque-like lesions covering the mucosa within the oral cavity and which may extend into the esophagus, proventriculus, etc. *Candida* is also seen as a lesion causing beak erosion. By contrast, scab-like lesions around the commissure of the mouth or on the eyelids are characteristic of the dry form of avian pox. "Wet" pox produces raised plaques in the oral cavity, esophagus, etc. and is considered a more severe, life threatening form of pox disease.

Auditory Canal. The auditory canals (Fig. 8.1) should be examined for exudate, infection, or blood. In cases of suspected trauma due to aggression, the canal is often swollen, partially closed, and filled with blood.

Neck. Carefully palpate the neck, trachea, and esophagus for the presence of solids, liquids, or gas (air). Because the lower part of the cervical esophagus of cranes, unlike many other bird groups, is not well developed as a storage area (crop), food and liquids generally pass quickly to the proventriculus, with the result that, on most examinations, the esophagus is empty. Gross distention can indicate blockage or impaction.



Fig. 8.1. Auricular feathers cover the auditory canal.

Photo David H. Ellis

Deformities of the vertebrae (Fig. 8.2) are unusual in cranes. Scoliosis and wryneck are seen occasionally in young chicks (Fig. 8.3). On palpation, the neck cannot be extended straight, or will not assume a normal curvature.

Thorax. The thoracic body cavity should be examined by palpation and by listening to the heart, lungs, and air sacs using a stethoscope. A bilateral enlargement, found on palpation of the thoracic inlet, is associated with enlarged thyroids (goiter) as seen

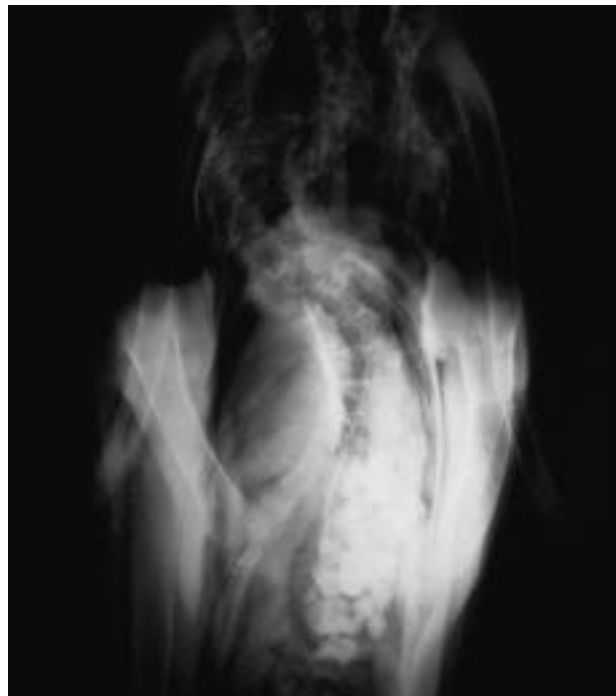


Fig. 8.2. Vertebral deformities (radiograph).

Photo Glenn H. Olsen



Fig. 8.3. Wryneck in a Sandhill Crane chick.

Photo Glenn H. Olsen

in some bird groups, but not yet reported in cranes. Some cranes occasionally develop subcutaneous emphysema. In this condition, air-filled pockets under the skin occur along the thorax or over the thighs, abdomen, neck, and even the head (Fig. 8.4). Generally, trauma, with rupture of an air sac and leakage of air under the skin, is suspected as the cause, but often no wound can be found.



Fig. 8.4. *Subcutaneous emphysema over the head of a male Greater Sandhill Crane.* Photo David H. Ellis

Body Condition Index and Weight. By palpating the breast (pectoral) muscles and sternum (keel), the degree of development or atrophy of skeletal muscles can be estimated and is reported as the bird's body condition index (BCI) (Fig. 8.5). A BCI of 4 or 5 (on a scale of 1-5) is indicative of a well-muscled or plump bird and the pectoral muscles will be rounded convexly from the keel. A bird with a BCI of 3 will have a rather flat profile to the pectoral muscles. A BCI of 2 is a bird with a concave shape to the pectoral musculature, and a BCI of 1 indicates severe muscle atrophy and emaciation. Differences exist seasonally in individuals and from bird to bird: healthy wild cranes and captive cranes that fly free usually have more devel-

oped pectoral muscles than birds with flight impairment. Generally, birds with amputations of a wing have a loss of pectoral musculature, especially on the side of the amputation. The greatest value in using BCI is not the comparison between birds, but rather a comparison with previous BCI readings for the individual. Caretakers should be trained to palpate and record BCI any time a crane is handled.

A drop in BCI normally indicates weight loss and a possible medical problem. Weighing is the best method for evaluating body condition and monitoring a bird's overall nutritional status. Direct weights can be taken as described in Chapter 2 (see Fig. 2.9). Weight loss signals the need for a complete examination by the veterinarian.

Abdomen. Gently palpate the abdomen for internal masses, fluid (ascites), or ovulated eggs. Generally the liver is not palpated unless enlarged. The gizzard and intestines are easily palpated, and any crepitus (gas), excess fluid, thickening, or masses in the intestines are possible to detect. The vent area should be examined for lesions, growths, protrusions, and for feces or urates accumulating on the feathers. A soiled vent in young chicks is frequently a sign of diarrhea, often caused by *Escherichia coli* infections (see Chapter 5, Veterinary Techniques section). The uropygial or preen gland above the base of the tail can be palpated for enlargement which can be caused by neoplasia, impaction, or infection.

Skin and Plumage. Examine the general condition of the skin and plumage. Look at the general level of hydration as indicated by the elasticity of the skin. Look for mites, lice, skin swellings (emphysema, abscesses), and missing or damaged feathers. Dull, split, or frayed feathers and stress bars across the feathers can indicate nutritional deficiencies, hormonal imbalances, or stress. Feather cysts and abnormally developed feathers are sometimes seen, especially on the wings. Skin irritation and broken feathers or an area of missing feathers on the thighs are seen in

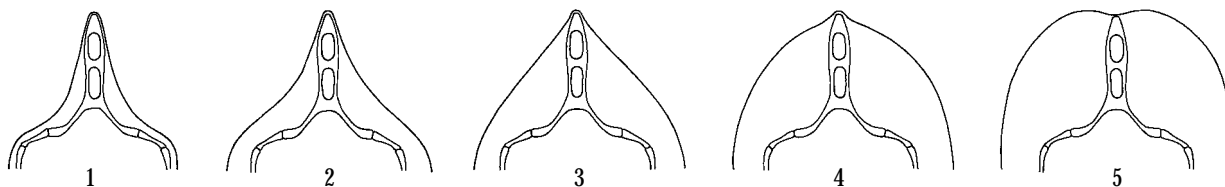


Fig. 8.5. *Body Condition Index (BCI) is an indicator of nutrition: cross section through mid sternum of Sandhill Crane (cavities are tracheal passages).*

Art. Kate Spencer

self-mutilation. A similar condition is often associated with reproductive activity in male cranes in the spring especially after being handled for artificial insemination (AI) for an extended period.

Wings. Examine each wing, palpating all bones and joints while also assessing muscle tone and extension. Be careful to maintain the wing in a natural position to avoid injury. Check the wings for swelling, bruising, or abrasions which are particularly common on the point of the carpus (wrist area). Subcutaneous hemorrhage in cranes will develop a green discoloration due to biliverdin (green pigment) released as red blood cells (RBCs) are destroyed. Matted feathers on a wing or over the body often indicate a wound.

Legs. Palpate each leg bone and joint while assessing muscle tone. Check the toes for normal extension, for broken nails, and for swollen areas on the toes or plantar foot. Toe swellings occur with fractures, luxations, and bumblefoot (pododermatitis), an infection frequently caused by *Staphylococcus aureus*. Cranes with a leg injury that forces them to stand on one foot, often develop pressure necrosis of this foot with resulting open wounds that develop into bumblefoot.

Auscultation. Auscultation of the chest should be included in every examination. Using a stethoscope, it is possible to determine heart rate, rhythm, and location of sound as well as to detect cardiac murmurs in chicks as young as 3 days. Some of these murmurs resolve within the first few days after hatching. Others are due to the most common congenital anatomic anomalies (i.e., atrial or ventricular septal defects). Other murmurs have been detected in severely anemic or dehydrated cranes and in cranes with pericarditis due to infection or visceral gout. Arrhythmia and cardiac tamponade have also been detected in cranes. Auscultation of the thorax is also useful for assessing the respiratory system. Unlike smaller birds, crane respiration produces distinct sounds associated with air movement. These sounds are normally louder on inspiration than expiration.

Clicks, wheezes, fluid-type sounds, or total absence of air movement sounds in one or more location can indicate a respiratory problem. Occasionally, unusual respiratory sounds are found on one examination and not on the next. The cause of these transitory sounds are unknown, but apparently are not related to major disease problems. Unilateral, dull respiratory sounds can indicate blockage of the major bronchi, or consolidation of lungs and air sacs on one side, and are often heard in advanced cases of aspergillosis.

Temperature. Unlike smaller birds, cranes appear to have a relatively constant body temperature (40.7°C , $\pm 0.4^{\circ}$ [105.3°F , $\pm 0.8^{\circ}$] in Whooping Cranes and 40.5°C , $\pm 0.6^{\circ}$ [104.7°F , $\pm 1.2^{\circ}$] in Sandhill Cranes). Although elevated temperatures have been observed in cranes suffering from bacterial disease, exertion, and stress, cloacal temperature monitoring is not commonly part of the physical examination.

Initial Care of a Sick Crane

After a sick or injured crane is found, it may be some time before a clear diagnosis is made. Causes for not having an immediate definitive diagnosis can be: (1) the crane is too weak to undergo extensive testing, (2) tests being performed for the suspected condition take time (sometimes days or weeks) for results to be received, (3) the crane's condition is attributed to multiple etiologies, not all of which have been properly identified, and (4) the condition is difficult to diagnose because the initial cause is no longer present. Having collected the required samples to confirm the clinical diagnosis, the practitioner must begin therapy until the results from the tests arrive.

Even prior to diagnosis, it is often important to begin some form of therapy. Of all the initial therapies, fluid therapy is the most important except in cases of severe anemia or hypoproteinemia. For cranes in shock from disease or trauma, bolus intravenous therapy has been useful (Redig 1984). We recommend using lactated Ringer's solution because of its similarity to avian plasma. Normal saline is also a good choice. Estimate the dehydration level (10% dehydration is a suitable estimate if dehydration is detected). The maintenance fluid requirement for a crane is about 44 mL/kg/day. Approximately 50% of the deficit should be given in the first 12 hours. The remainder of the deficit and the calculated maintenance dose should be given over the next two days. The fluids should be kept warm (35.5°C , 96°F) in an incubator or warm water bath.

Fluids can be delivered by one of several routes. The initial calculated dose of fluid can be administered as a bolus intravenously. The jugular or the medial tarsal vein are the easiest veins to access in the crane patient, although the ulnar or brachial vein can be used. Care needs to be taken to ensure that there is no further blood loss from hematoma formation, especially when using the jugular or brachial vein for

this procedure. Apply pressure over the venipuncture site for 1-2 min immediately after withdrawal of the needle. Using a small-gauge needle (25-gauge [5/8-in] or 26-gauge [3/4-in]) or butterfly needle may also help. For repeated intravenous therapy, a catheter (20-23-gauge in adults) can be placed in the medial tarsal, jugular, or brachial vein (under anesthesia).

Another equivalent route for frequent fluid administration is the intraosseous route (Ritchie et al. 1991). For this technique, feathers are removed from the carpal joint area, and the site is scrubbed with a disinfectant soak, rinsed, and sprayed with alcohol or iodine. A long needle (16-20-gauge, spinal needle) is used as a drill to pierce the distal cortex of the ulna and enter the marrow cavity. You should then be able to aspirate marrow. Flush the catheter with saline or heparin saline, then place an injection cap over the end of the needle. Suture or glue the needle to the skin of the wing. Finally, the wing is bandaged in a Figure-8 pattern to restrict movement and to cover the exposed needle hub and cap, protecting it from the probing beak of the crane.

In non-critical cases, fluids can be administered by the subcutaneous route. The advantage of this route is that it is the least traumatic for the crane and can easily be used for repeated dosages. The disadvantage is that absorption rates are slower, often as long as 30 min in a healthy crane and longer in severely depressed cranes. The best areas for subcutaneous fluid administration in cranes are the flanks just in front of the legs, the base of the neck, or intrascapular area.

With dehydration and capture myopathy, there may be an accompanying alteration in the acid-base balance resulting in acidosis. Ideally, the level of acidosis should be determined through laboratory testing, but immediate results are not always available. Therefore, if acidosis is suspected, a crane can be given 1 mEq/kg of sodium bicarbonate subcutaneously with fluids every 30 min to a maximum of 4 mEq/kg. Fluids containing dextrose are considered acidifying agents and should be used carefully in the dehydrated or acidotic crane. It is rare for a sick adult crane to be severely hypoglycemic. However, for young chicks, giving glucose or dextrose orally or by injection may be critical to survival.

For sick birds in general, a heated environment (30-32°C, 85-90°F) is helpful. This is especially true in very young crane chicks. Often sick older cranes are brought into a room heated to 21°C (70°F) and equipped with a heat lamp to raise the temperature in one area up to about 30°C (85°F).

A recumbent crane should be placed under the heat lamp, but continuously monitored for signs of overheating. When the crane becomes more active, it will move in or out from under the lamp as it requires heat supplementation.

Several medications may be indicated when the sick or injured crane is first presented. Corticosteroids such as dexamethasone or methyl prednisolone (see Table 8.1 for dosages) are indicated in cranes suffering from acute trauma or shock. Antibiotics are required in some traumatized and sick cranes. Ideally, the antibiotic chosen is based on cultures of the disease site and sensitivity testing, but results from such testing can take 4-7 days. Therefore, the clinician must decide on initial antibiotic therapy based on previous experience with the disease and the expected types of bacteria to be isolated by culturing.

Enrofloxacin, ampicillin, an aminoglycoside-penicillin combination such as amikacin and piperacillin sodium (with supplemental fluid therapy), and trimethoprim-sulfa combinations are each used on certain cases (see Table 8.1 for dosages). Enrofloxacin is a useful broad spectrum antibiotic, however, in young mammals, this drug has been shown to interfere with joint cartilage development. Although ontogenic problems have not been reported in any bird, use this drug with caution. Ampicillin is used with the injured, dehydrated crane because of this drug's safety, low renal toxicity, and bacteriocidal properties. Aminoglycoside-penicillin combinations are useful in sick cranes, especially for respiratory diseases, but not in cases involving impaired kidney function because aminoglycosides can be toxic to the kidneys.

Vitamin injections can be given as part of the supportive program (see dosages in Table 8.1). Vitamin A is important in cases where a deficiency is suspected (see initial examination) or in bumblefoot. Iron dextran injections (see Table 8.1 for dosage) are given in cases of anemia as indicated by a low hematocrit and pale mucous membranes. The use of iron dextran has been shown useful in raptors (Redig 1984), but no similar tests or dramatic results have been seen in cranes.

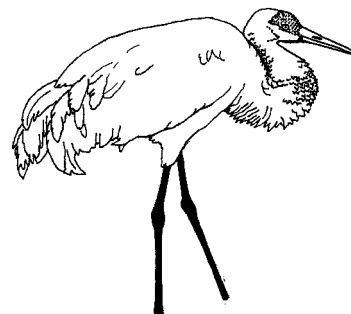


TABLE 8.1

Medications commonly used for cranes.¹

Drug	Indications	Route of Administration ²	Dosage	Treatment Schedule
Antibiotics				
Amikacin	Broad-spectrum, less nephrotoxic than gentamicin; often used in conjunction with piperacillin sodium; ensure adequate hydration	IM	10 mg/kg	2/day
Ampicillin	Broad-spectrum antibacterial drug for gram-negative and gram-positive bacteria, useful for several pathogenic enteric organisms	IM	100 mg/kg	2/day
Carbenicillin	Good only for 3 days after mixing; synergistic with aminoglycosides	IM, IV	100 mg/kg	2-3/day
Cefotaxime sodium	Broad-spectrum; sometimes used in conjunction with aminoglycosides	IM	50-100 mg/kg	3/day
Cephalexin	Broad-spectrum; effective against most gram-positive organisms and some gram-negative organisms, including various enteric organisms	oral	35-50 mg/kg	4/day
Cephalothin	Same as cephalexin	IM	100 mg/kg	4/day
Chloramphenicol	Broad-spectrum activity against both gram-positive and gram-negative bacteria, rickettsia, and chlamydia	SQ	100 mg/kg	3/day
Enrofloxacin	Broad-spectrum antibiotic	IM, oral	8-15 mg/kg	2/day
Gentamicin	Broad-spectrum; used therapeutically to treat bacterial infections in cranes and prophylactically against bacterial infections in newly hatched chicks; ensure adequate hydration	IM	5 mg/kg	2-3/day (1 in newly hatched chicks)
Piperacillin sodium	Used with amikacin	IM	100 mg/kg	2/day
Trimethoprim sulfa	Respiratory and enteric infections, also used as anticoccidial; regurgitation common orally	oral IM	16-24 mg/kg ³ 8 mg/kg ⁴	2-3/day 2/day
Tylosin	Effective against gram-positive and some gram-negative bacteria mycoplasma, and chlamydia; useful for respiratory infections	SQ	15 mg/kg	3/day
Corticosteroids				
Dexamethasone	Shock, trauma, endotoxemia capture myopathy, etc.	IM, IV, SQ	2-8 mg/kg (reduce doses for long-term therapy)	1-2/day
Prednisolone	Shock, trauma, chronic lameness	IM, IV	2 mg/kg	1-2/day
Prednisolone sodium succinate	Shock	IM, IV	10-20 mg/kg	as needed

TABLE 8.1 CONTINUED

Medications commonly used for cranes.

Drug	Indications	Route of Administration ²	Dosage	Treatment Schedule
Vitamins				
Vitamin A (Aquasol A) 100,000 units/mL	Hypovitaminosis A, sinusitis, ophthalmic diseases, avian pox	IM	1.0 mL/kg	twice weekly
Vitamins A, D ₃ , E (Injacom-100) 100,000 units A, 100,000 units D ₃ /mL	Hypovitaminosis A, ophthalmic diseases, fractures, egg binding, soft shelled eggs, and respiratory infections (especially sinusitis)	IM	1.0 mL/kg	once weekly
Vitamin B complex	CNS signs, trauma, muscular weakness, anemia, debilitation, and anorexia	IM	1-3 mg/kg	1/day
Vitamin E and selenium (50 mg Vitamin E, 1 mg Se/mL)	Muscular weakness, capture myopathy, leg dysfunctions, prior to or at times of capture or stressful event	IM	0.05-0.10 mg/kg	once every 14 days
Injectable Tranquilizers and Anesthetics				
Ketamine HCl	Disassociative anesthetic	IM	10-22 mg/kg	once, lasts 10-30 min
Xylazine	Tranquilizer, given with ketamine	IM	1.0-2.2 mg/kg	once
Diazepam	Tranquilizer, use alone or with ketamine	IM	0.5-1.0 mg/kg	once, lasts 2-6 hr
Midazolam	Tranquilizer	IM	15 mg/kg	once
Yohimbine	To reverse xylazine	IV	0.1 mg/kg	repeat in 10 min as needed
Tolazoline	To reverse xylazine	IV	15 mg/kg	once
Flumazenil	To reverse midazolam	IM	0.1 mg/kg	once
Other Medications				
Doxapram	Respiratory stimulant	IM, IV	5-10 mg/kg	1/day
Iron dextran	Following hemorrhage or for iron deficiency anemia	IM	10 mg/kg	once every 7-10 days if hematocrit is still low
Phenylbutazone	Non-steroid anti-inflammatory, and anti-pyretic	Oral	3.5-7 mg/kg	2-3 times per day
Methocarbamol	Muscle relaxant	Oral IV	50 mg/kg 32.5 mg/kg	once

¹ From Custer et al. 1979; Bush et al. 1979, 1981a, 1981b; Lock et al. 1982; Carpenter 1986, 1993; Klein et al. 1994; Olsen and Carpenter 1996.² IM = intramuscular; SQ = subcutaneous; IV = intravenous.³ Dose based on trimethoprim suspension (8 mg trimethoprim and 40 mg sulfamethazole/mL).⁴ Dose based on trimethoprim 24% for injection (4 mg trimethoprim and 200 mg sulfadiazine/mL).

Nutritional Support of a Sick Crane

If a crane is not eating, supplemental feeding (i.e., tube feeding or gavage) may be required. Insufficient intake of calories can lead to emaciation quickly in a sick crane. The bird will first mobilize body fat and then will catabolize muscle. Severe nutritional depletion is indicated by a drop in blood glucose; most normal adult cranes will maintain a blood glucose over 180 mg/100 mL (Table 8.2A). Restoring and maintaining a positive energy balance and adequate protein, vitamin, and mineral intake is critical in the treatment of sick cranes.

Tube feeding of cranes is easily accomplished with one person holding the crane in the normal fashion (Fig. 5.16), while a second person grasps the head and, using the thumb and fore-finger, gently forces the beak open. With the other hand, a flexible tube (we use red rubber urinary catheters, size 10-16 French) is inserted into the mouth, over the tongue, and down the esophagus to the level of the thoracic inlet or proventriculus. Care must be taken at this point to determine that the tube is in the esophagus and not the trachea. This is done by inspecting the mouth and seeing that the tube passes over the glottis. When the tube is in place properly, two cylindrical structures can be palpated, the trachea and the tube in the esophagus. If the tube is in the trachea, then only one cylindrical structure will be felt, and the tube should be removed immediately and reinserted properly before administering the food.

A 30-140-cc syringe containing the chosen tube feeding formula (see below) is attached to the tube, and the food is gently forced down the tube while watching the mouth for regurgitation. If the bird starts to regurgitate, stop tube feeding. First **remove the tube**, then quickly clean out the mouth especially around the airway, and then gently stroke the neck in a downward motion to encourage swallowing. Decrease the amount of formula fed on subsequent tubings to prevent further regurgitation. Because tube feeding also contributes to fluid balance, adjust total fluid therapy accordingly.

In the severely emaciated crane, an initial tube feeding or two should be of a high carbohydrate, low protein diet. Products useful in such situations are Emeraid I and some human enteral diets. Emeraid I is usually mixed 1:1 with water, though more dilute

preparations can be used. After several successful feedings and normal fecal production, a more complex diet providing protein, fat, and fiber may be tube fed long term. Several different formulas have been used successfully in cranes. One common approach is to make a gruel from the crane's pelleted diet then supplementing with several additional nutrients (e.g., Table 5.3). Other choices are Emeraid II (mixed 1:1 with warm water) and liquid human enteral products.

Most of these formulations have a caloric density of 0.5-1.5 kcal/mL. To determine the amount of tube feeding formula that the crane needs each day, calculate the adult crane's daily energy requirement (see also the Nutritional Support section of Chapter 5) using this formula:

$$\text{Daily Energy Requirement (Kcal)} = 1.5 \times \text{Basic Metabolic Rate (BMR)}$$

$$\text{BMR} = 78 \times \text{Weight}_{\text{kg}}^{0.75} \text{ (Quesenberry et al. 1989)}$$

The daily energy requirement divided by Kcal/mL of the formula equals mL of formula needed per day. The total volume should be divided into 2-4 meal. Do not feed more than 150 mL at one meal. It is best to start with meals of 60-80 mL.

Once a crane is eating normally again, tube feeding can be discontinued. The crane needs to eat approximately 100-200 g of pelleted food daily to maintain its body weight. It is often better to allow a crane some weight loss as it feeds itself rather than to continue the stressful practice of tube feeding.

Clinical Pathology

Hematology

Blood parameters are invaluable in diagnosing diseases and monitoring health. It is advisable to establish baseline blood profiles for your collection. Standard avian hematology techniques are appropriate for cranes. Excellent hematology references, complete with color plates of avian blood cells, are Dein (1984), Campbell (1988, 1994), and Hawkey and Dennet (1989).

Equipment for Blood Collection. Blood can be collected using a 1-cc, 3-cc, 10-cc, or 60-cc syringe. The only use for the 60-cc syringe is collecting blood for transfusions. A 25-gauge needle is adequate for collecting small amounts of blood, but larger needles (20-23-gauge) or a catheter are preferred for collecting larger samples.

TABLE 8.2A

Normal hematologic and serum chemistry values for captive cranes¹

	Sandhill Crane		Whooping Crane		Siberian Crane		Red-crowned Crane		Wattled Crane	
	Mean	Range	Mean	Range	Mean	Range	Mean	Range	Mean	Range
Hematocrit (%)	43.0	37-49	42.0	38-48	45.0	40-50	39.0	33-45	44.8	40-50
Hemoglobin (g/100mL)	13.5	10.5-18.7	14.4	13.0-16.7	—	—	—	—	—	—
Red Blood Cell (10 ⁶ /mm ³)	2.5	1.9-3.3	2.2	1.8-2.6	—	—	—	—	—	—
White Blood Cell (10 ³ /mm ³)	13.0	6.2-22.6	18.2	12.2-25.1	10.8	6.5-15.0	14.9	6.3-23.5	12.7	3.2-22.2
Total Protein (g/100mL)	3.9	2.9-7.9	3.8	3.1-4.4	3.6	3.1-4.1	3.3	2.9-3.6	3.1	2.8-3.4
Albumin (g/100mL)	1.5	1.0-2.5	1.5	1.2-1.7	1.4	1.2-1.5	1.2	1.1-1.3	1.1	1.0-1.3
Globulin (g/100mL)	2.3	1.8-3.4	2.3	1.8-2.8	2.3	1.9-2.7	2.1	1.9-2.3	2.0	1.8-2.1
Albumin/Globulin	0.6	0.4-1.3	0.7	0.6-0.8	0.6	0.5-0.7	0.6	0.5-0.7	0.6	0.4-0.6
Alkaline Phosphatase (IU/L)	164.0	34-423	46.0	28-72	45.2	28-68	226.4	128-409	37.2	13-61
Lactic Dehydrogenase (IU/L)	278.0	108-488	440.0	178-975	202.3	100-323	287.5	161-372	137.0	55-249
Aspartate Aminotransferase (IU/L)	181.0	16-260	261.0	133-612	181.6	117-254	208	108-456	188.5	148-230
Alanine Aminotransferase (IU/L)	50.0	19-162	53.0	42-71	16.1	6-25	29.6	18-67	10.5	10-11
Glucose (mg/100mL)	247.0	87-323	232.0	210-267	266.4	209-314	267.4	239-328	266.2	246-293
Uric Acid (mg/100mL)	9.7	4.1-24.6	8.1	6.5-10.2	9.0	5.5-12.6	7.8	4.2-11.3	7.7	5.4-11.1
Creatinine (mg/100mL)	0.7	0.4-1.2	0.6	0.4-0.8	0.3	0.3-0.4	0.3	0.2-0.4	0.4	0.3-0.5
Cholesterol (mg/100mL)	128.0	87-187	148.0	96-200	212.3	148-286	170.2	140-217	147.0	120-188
Creatine kinase (U/L)	—	—	—	—	106.1	48-205	114.3	53-333	76.7	19-139
Triglyceride (mg/dL)	—	—	—	—	142.4	128-555	265.4	108-520	110.3	58-169
Iron (mg/dL)	—	—	—	—	106.3	61-155	100.5	61-150	161.3	109-238
Calcium (mg/100mL)	9.7	8.8-10.9	9.1	8.3-9.7	10.5	9.5-11.2	10.8	10.4-11.2	10.8	10.3-11.6
Phosphorus (mg/100mL)	3.6	1.7-5.4	2.8	2.0-4.1	3.8	1.9-5.8	3.5	2.0-4.4	2.7	1.6-5.7
Sodium (mEq/L)	148.0	142-160	147.0	140-152	148.5	146-151	147.5	143-150	146.3	144-153
Chloride (mEq/L)	108.0	101-115	107.0	102-113	109.1	106-113	107.0	104-110	108.3	105-111
Potassium (mEq/L)	3.4	2.2-4.8	3.4	2.6-4.2	2.9	1.6-4.0	2.8	1.6-4.0	3.2	2.0-3.9

¹ Based on Gee et al. 1981; Carpenter 1986; and unpublished work at Patuxent and ICF.

Blood Collection. Sample requirements vary according to laboratory and test. The maximum blood volume that can be safely collected is 1% of the crane's body weight (1 cc/100 g). Some laboratories require blood to be collected into heparinized capillary tubes (see Appendix), others require heparin or ethylenediaminetetraacetic acid (EDTA) to be used in the test tube to prevent coagulation of the blood. Blood from African Crowned Cranes will sometimes "sludge" or begin to clot even when EDTA is used as an anticoagulant; therefore, heparin is preferred for these species.

There are **three major venipuncture sites** for cranes. The preferred site, especially for large samples, is the **right jugular vein**. The crane is held in a normal carrying position (Figs. 2.6 and 5.4) while a second person, standing to the left of the head, holds the head and neck outstretched and rotated so the right side of the neck is up. With the second hand, this person applies firm pressure at the base of the neck, causing the right jugular vein to fill with blood. Wetting the feathers with alcohol makes the vein more visible. The person doing the venipuncture then moves in from the right and collects the required blood sample by inserting the needle through the skin into the jugular vein (Fig. 8.6). Pressure should be applied to the jugular vein at the puncture site for a minimum of 1 min after the needle is withdrawn to prevent the development of a hematoma. If the crane struggles or moves during the venipuncture, the needle may lacerate the wall of the vein and produce a hematoma. On rare occasions, this has resulted in the death of a crane. If a hematoma results, continue to apply pressure over the jugular vein at the hematoma site until the hematoma is no longer enlarging. Observe the crane for the next 30 min after release for signs such as ruffled neck feathers, enlarged neck, lethargy, or collapse.



Fig. 8.6. *Venipuncture of jugular vein.*

Photo Glenn H. Olsen

The **medial metatarsal vein** is another preferred site for venipuncture. One person holds the bird in the normal carrying position and applies pressure to the extended leg above the hock joint. This causes the medial metatarsal vein to be prominent along the inside of the hock and tarsometatarsus. A second person cleans a small area with alcohol, grasps the leg below the venipuncture site with one hand, and with the other hand, draws the sample with the syringe. This technique works well in large cranes, but in smaller cranes, the vein is smaller and frequently collapses as blood is withdrawn. Only small amounts of blood (up to 5 cc) can be withdrawn from this vein, and the vein will collapse if blood is drawn quickly. Other disadvantages of this site are the difficulties in restraining the leg and the risk of injuring the leg if the bird struggles. Advantages are that hematomas are rare because minimal subcutaneous space is available for blood to pool between the bone, tendons, and scales. Also, if bleeding does occur, a pressure bandage can be easily and safely applied.

The third site commonly used for venipuncture is the **brachial vein** (cutaneous ulna vein) on the underside of the wing in the area of the elbow. This requires that the bird be held supine (breast up) on a flat surface (ground, table, etc.) with one wing extended. A second person applies pressure over the humerus to help fill the brachial vein. Clean the site with alcohol to separate the feathers and expose the vein. A third person moves in from the caudal aspect of the wing to obtain the sample. Up to 10 cc can be collected from this site. A disadvantage, though, is the difficulty of safely restraining the bird in this position and the increased risk of injury. Small hematomas often occur at this venipuncture site. This technique has been used to obtain blood samples from anesthetized cranes where manipulation of the neck may interfere with delivery of the gaseous anesthetic agent.

Making Blood Slides. The conventional method of using two slides to make a smear will damage avian cells unless a beveled edge slide is used to make the smear. Another technique is to dilute the blood 1:4 with 30% bovine albumin solution (Olsen and Gaunt 1985). A second method of making blood smears is to place a drop of blood on a glass coverslip, then place a second glass coverslip (or a slide) on the first, allowing the blood drop to spread between the coverslips. Thereafter, quickly slide the coverslips apart, leaving a smear of blood on each slip.

Staining Smears. Smears can be stained with a Wright or Wright-Giemsa stain, using Cameo Quick

Stain II or Diff-Quick (see Appendix). The three solutions used in staining slides (Wright-Giemsa technique) are methanol, phosphate buffered eosin, and phosphate buffered thiazine. Slides are first fixed in methanol for 10-15 sec, then stained for 10-15 sec in each of the other two solutions, and finally rinsed in distilled water.

Evaluating Blood Smears. When reading blood smears made by the two-slide method it is important to read all areas of the slide (both the edge and center of the smear) to get an accurate differential cell count. When reading slides made by the cover glass method read only the central portion of the smear.

Crane **red blood cells** (RBCs) are oval or elliptical. Each cell has a central nucleus, shaped similar to the cell and consisting of dark purple clumps of chromatin. Cytoplasm is slightly orange-yellow to pink tinged. Immature RBCs have blue-tinged cytoplasm, a lighter nucleus (less dense chromatin), and may be more prevalent in cases of anemia.

Thrombocytes are similar in function to mammalian platelets. Their cytoplasm is pale blue and may contain 1-3 small magenta granules which help to differentiate thrombocytes from immature RBCs or lymphocytes. The thrombocyte nucleus is dense and dark purple when stained. Thrombocytes are often found in clumps.

Heterophils are round with clear to pink cytoplasm containing usually elongated, pink, red, or purple, granules. The nucleus is blue to purple and can have two or more lobes. The cytoplasm of heterophils is normally clear, but toxic heterophils have deep blue vacuolated cytoplasm.

Eosinophils are round with pale blue cytoplasm and bright red, round to oval, granules. The nucleus is blue to purple with two or more lobes. The primary distinguishing characteristic between heterophils and eosinophils is the shape of the granules (elongated in the heterophil, round in the eosinophil).

Basophils are also round but have clear cytoplasm with deep purple granules and a blue or purple nucleus. Basophils are extremely rare cells, about 1-2 cells per 500 white blood cells (WBCs) counted.

Monocytes are large and irregular in shape. They have a light blue cytoplasm that may be vacuolated. The nucleus is often eccentrically located, round or elongated, and bilobate.

There are two types of **lymphocytes** seen in avian blood. Type I lymphocytes are similar in shape and appearance to thrombocytes. They have a small amount of deep blue cytoplasm. The nucleus is large compared

to cell size, blue to purple colored, round, and eccentrically located. Type II lymphocytes are larger than type I lymphocytes and can be confused with monocytes. They have pale blue cytoplasm, a blue irregular shaped nucleus and higher cytoplasm-nucleus ratio.

Hematocrit. The hematocrit (HCT) or packed-cell-volume (PCV) is measured by filling a microhematocrit tube and spinning the tube in a high speed centrifuge to obtain a centrifugal force of 2260G (ca 3800 rpm for 10 min in a No. 2 International Centrifuge [see Appendix] [Campbell 1988:7]). Read the results by measuring the height of the RBC column over the height of the total column (cells and serum) and express as a percentage. Light yellow coloring of the serum or plasma can be due to carotenoid pigments (Dein 1984) and does not necessarily indicate icterus (abnormal yellowing of serum and some tissues). Lipemia (elevated fat) may be present as a milky-white serum in obese birds, post-feeding birds, and laying females.

Hemoglobin. Hemoglobin (Hb) measurements are important because they relate to the ability of the RBCs to transport oxygen. Hemoglobin measurements can be made using standard methods developed for mammals.

One of the oldest methods uses a hemoglobin scale. A drop of whole blood is placed on white filter paper, and the resulting red dot is matched for intensity with the best choice on a red color chart. This method is rapid, simple, inexpensive, and a good technique under field conditions, but the error is $\pm 10-40\%$ (Schalm et al. 1975:52-55).

Another simple method, but one requiring the purchase of equipment, is the oxyhemoglobin method. In this procedure, a drop of blood is placed on a glass plate, and the cells are destroyed using a hemolytic agent (usually saponin) dried on the end of a special applicator stick. Then a second glass plate is placed over the first, and the two are pressed together. The glass plates are then placed in the Spencer hemoglobinometer (see Appendix). Finally, the green color of the sample is matched against the standards read through the hemoglobinometer. Green is the color used for matching because the maximum absorption of hemoglobin under visible light occurs in the green band, and for the human eye, green is an easy shade to match accurately (Coles 1980:79-83).

An extremely accurate technique for hemoglobin is the cyanmethemoglobin method. The first step is to prepare a 1:200 dilution of blood in 0.04% ammonium hydroxide. Then the solution is placed in a

spectrophotometer or a filter photometer and read at 540 m μ . The percent transmission at 540 m μ is compared to a standard solution of cyanmethemoglobin, converting percent transmission to grams of hemoglobin per dL of blood (Schalm et al. 1975:52-55; Coles 1980:79-83).

Red Blood Cell Count. The total RBC count can be obtained using a Coulter counter or by the Unopette system (see Appendix) using standard techniques as in mammals (Dein 1984).

Mean corpuscular volume (MCV):

$$\text{MCV} = (\text{HCT}/\text{RBC}) \times 100$$

Mean corpuscular hemoglobin concentration (MCHC):

$$\text{MCHC} = (\text{Hb}/\text{HCT}) \times 100$$

Mean corpuscular hemoglobin (MCH):

$$\text{MCH} = (\text{Hb}/\text{RBC}) \times 100$$

The above calculations are also used for other species of birds and mammals, but the reference values for normal individuals of most species of cranes need to be established. Table 8.2A contains some normal values for Whooping, Sandhill, Siberian, Red-crowned, and Wattled Cranes.

White Blood Cell (WBC) Count. Because avian RBCs are nucleated, the techniques used to separate red and white blood cells in mammals will not work. Therefore, automated counters such as the Coulter unit cannot be used. Similarly, hand-counting systems such as the Unopette WBC count will not work with crane blood. The Eosinophil Unopette method is the most useful technique for estimating WBCs in cranes. However, the Eosinophil-Unopette method counts only heterophils and eosinophils. Mononuclear cells (monocytes and lymphocytes) are not counted, but the count is corrected to include these cells based on the differential count (ratio of various WBCs seen on the blood smear). The procedures to follow are:

1. Use the protective cap over the Unopette pipette tip to puncture the diaphragm on the small reservoir containing the stain.
2. Fill the pipette until blood reaches the neck.
3. Carefully wipe the tip of the pipette.
4. Gently squeeze the reservoir containing stain as the pipette is inserted securely into the reservoir.
5. Mix the blood and fluid in the reservoir by inverting 10-15 times.
6. Wait 5-10 min for staining.
7. Switch the pipette direction so it can now be used to fill a hemacytometer. Gently invert 3-4 times to uniformly resuspend cells.

8. Discard the first 10 drops and then fill both sides of the hemacytometer.
9. Place the hemacytometer in a covered petri dish containing a small amount of moistened filter paper (to prevent loss of fluid due to evaporation from the hemacytometer). Leave the hemacytometer undisturbed for 5 min to allow the cells to settle. Read the sample within 20 min of beginning the test, because RBCs will also eventually absorb the stain.
10. Count purple-stained cells in all 9 bold squares of both sides of the hemacytometer (Fig. 8.7). Find the total heterophil-eosinophil count (H/E) by multiplying the total number of cells counted by 17.6. An alternative method is to count 5 bold squares on both sides of the hemacytometer (Fig. 8.7) and multiply by 32.
11. Evaluate the WBCs on a stained blood smear for a differential count (Table 8.2B: usually based on counting 100-200 WBC).

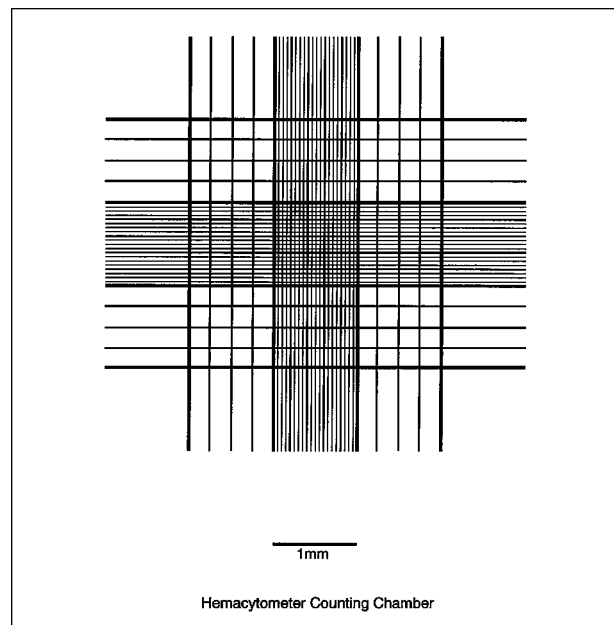


Fig. 8.7. An Improved Neubauer Hemacytometer grid. Each hemacytometer has two grids. Sperm counts are determined by counting 5 of the 25 squares on one grid within the 1-mm square. A normal counting pattern is to count the contents of the following squares: upper left, upper right, lower left, lower right, and center. For the white blood cell count, the same pattern can be used, but count 5 of the 9 squares within the 3-mm square and total the number of cells on this grid and the second grid. Red blood cell counts are made by counting all cells within the 1-mm central square. To avoid overcounting cells that contact grid lines, include only cells that fall on the upper and left boundary lines of the square.

TABLE 8.2B

White blood cell hematological values (average, standard deviation, and range) for healthy cranes of six species.¹

	WHOOPING CRANE N=17	SANDHILL CRANE N=5	RED CROWNED CRANE N=11	SARUS CRANE N=12	SIBERIAN CRANE N=14	WATTLED CRANE N=6
White Blood Cell (N/ μ L) ²	16999	13430	14933	14446	10770	12672
SD	6671	4626	4303	2687	2132	4761
Range	9752-29726	8392-20900	6327-23539	9072-19820	6506-15034	3150-22194
Heterophil (%)	56	53	41	59	53	48
SD	10	2	10	7	10	15
Range	38-74	50-56	22-62	45-73	33-73	21-81
Lymphocyte (%)	41	40	48	32	39	39
SD	11	3	9	8	10	20
Range	21-60	38-43	29-65	16-48	18-58	0-75
Monocyte (%)	2	6	6	4	3	5
SD	2	4	2	4	1	3
Range	0-6	3-13	2-10	0-12	2-6	0-11
Eosinophil (%)	1	2	5	5	5	8
SD	2	1	1	1	2	4
Range	0-5	0-3	3-7	3-7	1-9	1-17
Heterophil (N/ μ L)	9597	7078	6120	8486	5678	6108
SD	4616	2385	1859	1755	1030	1701
Range	4702-21997	4448-10868	2402-9838	4976-11996	3618-7738	2706-9510
Lymphocyte (N/ μ L)	6836	5353	7213	4571	4183	4890
SD	3064	2136	3051	1633	1814	4067
Range	3129-13506	3609-8987	1111-13315	1305-7837	555-7811	0-13024
Monocyte (N/ μ L)	390	802	882	606	370	604
SD	322	515	348	535	117	413
Range	0-976	334-1544	186-1578	0-1676	136-604	0-1430
Eosinophil (N/ μ L)	138	210	700	783	546	1070
SD	230	204	314	290	198	258
Range	0-688	0-418	72-1328	203-1363	150-942	554-1586

¹ Basophils were absent in samples.

² As stated in the text, the White Blood Cell count is not derived from, and does not exactly equal, the totals for all WBC groups at the bottom of the table.

12. The total WBC is determined as:

$$\text{Total WBC/mm}^3 = (\text{total heterophils} + \text{total eosinophils}) / (\% \text{ heterophils} + \% \text{ eosinophils}).$$

Interpretation of the Hemogram. Tables 8.2A and B have blood parameter reference ranges for five species of cranes. Similar values for other crane species are found in Connetta et al. (1974), Hawkey et al. (1983), Cook et al. (1989), and Puerta et al. (1990), and in the databases available through ISIS/MedARKS (see Chapter 10). Some additional parameters are not in the tables. Mean corpuscular volume is $1.12\text{--}2.58 \times 10^{-3}\text{mm}^3$ for Sandhill Cranes and $1.46\text{--}4.38 \times 10^{-3}$

mm^3 for Whooping Cranes. Mean corpuscular hemoglobin is $3.2\text{--}9.8 \mu\text{g}$ for Sandhill Cranes and $5.0\text{--}9.3 \mu\text{g}$ for Whooping Cranes. The mean corpuscular hemoglobin concentration is $21.4\text{--}50.5\%$ for Sandhill Cranes and $27.1\text{--}43.9\%$ for Whooping Cranes. Table 8.3 provides hematologic changes seen in developing Sandhill Crane chicks. Reference values for these chicks are not the same as for adults.

A crane's blood cells generally respond to disease as do blood cells of other birds (Hawkey et al. 1983). For example, just as for other avian groups, anemia in cranes is associated with bleeding wounds, gastrointestinal foreign bodies, lead poisoning, blood

TABLE 8.3

Pediatric hematologic and serum chemistry values (mean; range) for captive Sandhill Crane chicks.¹

Age in days	0-2	6-8	13-15	20-25	27-29	34-36
Hematocrit (%)	33; 27-37	28; 23-30	28; 25-30	29; 25-33	28; 24-35	28; 25-32
Red Blood Cell Count ($10^6/\text{mm}^3$)	1.47; 1.22-1.71	1.31; 1.12-1.52	1.30; 1.09-1.48	1.32; 1.14-1.59	1.37; 1.18-1.65	1.32; 1.17-1.56
Mean corpuscular volume (MCV) (fL)	217; 206-231	216; 187-235	206; 190-235	212; 195-235	208; 185-223	215; 192-235
White Blood Cell Count ($10^3/\text{mm}^3$)	20.5; 13.6-33.5	12.0; 7.0-48.3	16.0; 9.7-28.5	13.2; 7.8-19.4	12.9; 7.9-16.4	18.3; 8.3-29.1
Heterophil (%)	52; 38-77	52; 36-66	53; 36-75	53; 40-68	45; 24-61	43; 24-61
Lymphocyte (%)	40; 15-50	42; 30-59	45; 24-64	43; 28-49	50; 33-69	54; 35-76
Monocyte (%)	4; 1-8	5; 2-13	2; 0-6	4; 0-12	4; 1-11	3; 0-9
Eosinophil (%)	2; 0-4	2; 0-4	0; 0-1	1; 0-1	1; 0-2	0; 0-2
Basophil (%)	0; 0-1	1; 0-3	0; 0-1	1; 0-2	0; 0-1	0; 0
Total Protein (g/100mL)	3.4; 3.1-4.2	3.4; 2.9-4.5	3.5; 2.3-5.4	3.3; 3.0-3.6	3.4; 3.2-3.8	3.5; 3.0-4.0
Albumin (g/100mL)	<0.5	<0.5-0.5	<0.5	<0.5	<0.5-0.8	<0.5-0.5
Alkaline Phosphatase (IU/L)	179; 23-224	274; 186-372	331; 203-524	369; 230-538	391; 283-553	282; 327-510
Lactic Dehydrogenase (IU/L)	—	—	—	241; 204-288	271; 192-384	308; 293-322
Aspartate Amino-transferase (IU/L)	99; 88-110	262; 221-322	191; 143-263	144; 123-161	146; 118-160	151; 111-195
Glucose (mg/100mL)	—	—	228; 205-240	212; 178-251	241; 229-261	221; 204-237
Uric Acid (mg/100mL)	4.7; 3.7-6.9	5.5; 4.4-7.2	6.8; 6.4-8.3	7.6; 6.0-9.9	6.2; 4.8-9.3	5.4; 4.6-6.4
Gamma Glutamyl Transferase (IU/L)	—	—	—	2; 2-3	3; 2-3	3; 2-6
Creatinine (IU/L)	—	—	—	128; 62-252	144; 40-247	228; 140-290
Calcium (mg/100mL)	8.1; 6.0-10.6	6.9; 5.5-7.9	7.9; 6.8-8.8	8.0; 6.7-10.2	7.7; 5.7-9.9	8.9; 7.1-9.6
Phosphorus (mg/100mL)	—	—	5.8; 5.1-7.2	5.9; 4.5-8.1	6.1; 4.9-7.0	6.3; 5.7-7.0

destruction, or decreased blood cell production. An increase of basophilic appearing RBCs above 5% or a reticulocyte (a type of immature RBC) count above 10-15% is indicative of a regenerative anemia. Regenerative anemia is usually associated with blood loss or destruction from such causes as hemorrhage from wounds, blood parasites (causing RBC loss), gastrointestinal parasites, certain bacterial infections, lead, and other toxins. Non-regenerative anemia is associated with decreased RBC production from such causes as pesticide toxicity, lead toxicity, chloramphenicol toxicity, or certain viral infections.

High total WBC counts have been associated with chronic bacterial infections, avian tuberculosis, fungal infections, and stress. Low WBC counts are rarely seen, but have been documented in inclusion body disease of cranes, in overwhelming bacterial infections, and in immunologically depressed cranes.

Serum Chemistry

In addition to the information gained from examining the blood cells, there are many substances in the serum that can be analyzed to help assess the health of the crane patient. Serum chemistry reference

TABLE 8.3 CONTINUED

Pediatric hematologic and serum chemistry values (mean; range) for captive Sandhill Crane chicks.

Age in Days	41-43	48-50	55-57	62-64	69-76
Hematocrit (%)	29; 28-32	29; 28-31	28; 26-34	30; 28-34	34; 30-39
Red Blood Cell Count (10 ⁶ /mm ³)	1.30; 1.17-1.65	1.38; 1.19-1.62	1.34; 1.17-1.55	1.44; 1.29-1.73	1.57; 1.40-1.75
Mean corpuscular volume (MCV) (fL)	211; 189-235	209; 191-223	210; 200-222	206; 196-223	214; 206-225
White Blood Cell Count (10 ³ /mm ³)	24.1; 14.5-32.8	23.6; 10.8-39.8	16.4; 10.8-23.9	14.7; 6.7-24.1	15.8; 5.6-20.4
Percent Heterophil	35; 20-61	42; 20-68	41; 21-64	40; 31-60	39; 18-49
Lymphocyte (%)	61; 38-77	56; 21-75	57; 31-73	60; 51-68	60; 49-83
Monocyte (%)	3; 2-5	4; 1-7	2; 1-6	3; 1-7	2; 1-3
Eosinophil (%)	0; 0-2	0; 0-1	0; 0	0; 0	0; 0
Basophil (%)	0; 0	0; 0	0; 0	0; 0	0; 0
Total Protein (g/100 mL)	3.0; 3.4-4.3	3.6; 3.3-3.8	3.7; 3.4-4.0	3.7; 3.1-4.1	3.8; 3.3-4.2
Albumin (g/100mL)	<0.5-0.6	<0.5-0.6	<0.5-0.5	<0.5-0.8	<0.5-0.8
Alkaline Phosphatase (IU/L)	449; 232-691	388; 286-529	399; 281-529	386; 271-616	288; 225-354
Lactic Dehydrogenase (IU/L)	239; 222-262	249; 194-292	285; 240-431	294; 192-499	324; 176-472
Aspartate Amino-transferase (IU/L)	159; 151-161	159; 110-184	152; 133-177	152; 119-182	173; 152-199
Glucose (mg/100mL)	228; 219-259	219; 214-233	214; 196-228	223; 197-254	212; 202-222
Uric Acid (mg/100mL)	6.1; 5.9-6.4	5.4; 4.7-8.1	5.2; 5.0-5.3	5.6; 4.8-8.4	6.4; 4.7-8.0
Gamma Glutamyl Transferase (IU/L)	3; 3	3; 2-5	2; 2-4	2; 2-4	2; 2-3
Creatinine (IU/L)	159; 128-191	168; 152-184	127; 92-169	172; 81-306	169; 71-337
Calcium (mg/100mL)	8.9; 6.1-9.3	8.0; 6.9-9.2	9.3; 8.2-10.2	8.9; 6.4-11.0	9.2; 7.1-10.9
Phosphorus (mg/100mL)	6.9; 5.9-7.9	6.5; 5.4-7.1	6.8; 6.0-7.7	6.8; 6.0-7.8	6.9; 6.0-7.8

¹ Based on unpublished work from ICF

ranges for cranes are given in Tables 8.2 and 8.3, and in the same sources cited earlier for blood parameters plus Chappell and Brannian (1984). The serum chemistry tests described below can be performed on the automated analyzer used for mammalian and human serum chemistry analysis in a medical laboratory. Alternately, many veterinary facilities have analyzers. The actual test procedures will vary with the analyzer used and should be handled by the technical staff familiar with the operations of the machine.

Useful crane serum parameters include total protein, albumin, calcium, glucose, lactic dehydrogenase, alkaline phosphatase, aspartate aminotransferase (glutamic-oxaloacetic transaminase), uric acid, creatinine kinase, creatinine, bile acids, sodium, potassium, calcium, and phosphorus. Other serum chemistries, important in human medicine, such as blood urea nitrogen and alanine aminotransferase (glutamic-pyruvic transaminase) are not as useful for diagnosis of crane diseases. The alterations in serum chemistry seen in sick cranes are not well documented in the literature, but are believed to be similar to what has been recorded for other avian species (Hochleithner 1994).

Total protein (normal for all chemistries are listed in Table 8.2A) values can be elevated with dehydration, lipemia, or hyperglobulinemia secondary to chronic diseases (such as aspergillosis, avian tuberculosis, and chronic bacterial infections such as *Staphylococcus*). Total protein values can decrease in malnutrition, acute infections, chronic liver disease, or malabsorption of nutrients caused by severe intestinal parasitism.

Calcium values increase with egg laying and can be as high as 3 times normal. Decreased calcium values are seen with egg binding or nutritional imbalances. Low calcium levels have been documented as leading to seizures in some avian species, however, this has not been reported in cranes. The calcium/phosphorus ratio is important, especially in growing birds, and should be 2:1.

Glucose increases with stress or diabetes mellitus (a disease reported in other avian species but not in cranes to date). Decreases in serum glucose are seen in starvation, septicemia, severe liver disease, endocrine disorders, or with improperly processed samples. If the RBCs remain with the serum over 60 min at 22°C (70°F) or higher, the cells continue to use glucose from the serum, lowering the glucose reading.

Lactic dehydrogenase (LDH) levels in birds can be increased by liver disease, aminoglycoside therapy, intramuscular injections, cardiac or skeletal muscle catabolism, or *Chlamydia* infections. Decreases do not suggest any specific condition. In addition, increased LDH values can result as an artifact if the serum sample is hemolyzed. High levels of this enzyme are present in the liver, skeletal muscle, and heart, while moderate amounts are present in kidney tissue and small amounts in intestine based on studies in other avian species. Any process or disease affecting these tissues may result in serum elevations of the enzyme.

Aspartate aminotransferase (AST) can be increased in *Chlamydia* infections, liver disease, bacterial septicemia, soft tissue trauma, starvation, toxicity, neoplasia, aminoglycoside therapy, or intramuscular injections. Decreases have no clinical significance. The enzyme is found in the liver, heart, brain, lung, bone, and muscle. Any process affecting one or more of these tissues may cause the enzyme to be elevated. In cranes, elevations are common due to mild muscle damage associated with normal handling. If the creatinine kinase (CK) is also mildly to moderately elevated, then mild, reversible, muscle damage is probably the cause. Severe elevations of both (i.e., >2 times normal value) indicate a serious muscle disease such as exertional (capture) myopathy. If the AST is elevated and the CK is not, then liver disease is a possibility.

Creatinine kinase will increase in muscle trauma (including intramuscular injections), central nervous system disorders, cardiac disease, or lead poisoning. Decreases are not significant. Because CK increases in heart and muscle disease, but not in liver disease, it is important to test with AST and LDH. Elevations in AST and LDH, but not CK, suggest liver disease or *Chlamydia* infections, whereas elevations in all three generally indicate heart or muscle disease.

Uric acid is the primary excretion product of the kidneys from nitrogen (protein) break down in cranes. Increases are characteristic of renal disease (including gout and damage from aminoglycoside medications), but may also occur in dehydration, starvation, trauma, or neoplasia. Decreases are not considered significant.

Creatinine may increase with renal disease, septicemia, egg peritonitis, or high protein diets (the latter occurs in some avian species, but has not been documented in cranes). Decreases are not considered

significant. The test is not very sensitive and some authors consider it of limited diagnostic value, but elevations have been seen in cranes with severe renal disease.

Bile acids are only elevated by liver disease and are considered the best serum indicator of a liver disorder. Lower than normal levels of bile acids are not considered significant. Bile acid levels tested in cranes at ICF have generally been lower than $80 \mu\text{mol/L}$.

Alterations in the electrolytes sodium (Na), potassium (K), and chloride (Cl) indicate a serious change in the bird's acid/base balance and metabolic state. In a crane, this is most likely due to renal disease (increased K and decreased Na), diarrhea (decreased K and Cl), or shock acidosis (increased K and decreased Na). An elevated K level by itself is often due to hemolysis of the serum sample.

Parasitology

Intestinal parasites can be diagnosed at necropsy and by sampling fresh fecal samples in living cranes. Three types of fecal examination (direct smear, flotation, and sedimentation) are routinely performed.

Flotation is a practical technique because it helps to concentrate the eggs of the parasites and removes other material in the sample. The eggs of nematodes, cestodes, and acanthocephalans float as do oocysts of coccidia, the cysts of *Giardia*, and other protozoa. Examine an adequate amount (ca 1-2 g) of fresh feces. Nematode eggs will larvate and *Giardia* may perish in samples that have lain on the ground more than 15 min. Feces lying on the ground can also be invaded by free-living, non-parasitic, nematodes which can confuse results.

The diagnostic techniques are most valuable if standardized procedures (described in Greiner and Ritchie 1994 and various parasitology texts) are used. The standard flotation medium is saturated sodium nitrate (568 g/L water). However, Sheather's sugar solution (500 g table sugar, 320 mL water, 6.5 g phenol crystals) works best to detect coccidia oocysts. Saturated zinc sulfate (336 g/L water) works well for *Giardia* concentration and detection (Greiner and Ritchie 1994). The fecal specimen is mixed with at least 10 times the volume of flotation medium. The mixture can then be strained and placed in a small cylindrical container. The flotation medium should fill the container. A microscope coverslip is placed on top of the container and

should make contact with the flotation medium. The mixture should be allowed to stand 45 min before the coverslip is placed on a microscope slide and read. To speed up the process, the strained fecal/flotation medium mixture can be placed in a 15 mL centrifuge tube and spun at 1200 rpm for 10 min. Then the top layer of medium is collected and the drop placed on a microscope slide, covered with a coverslip, and read. Parasites commonly diagnosed from eggs seen on flotation include *Capillaria* sp., *Eucoleus* sp., ascarids, acanthocephalans (*Macracanthorhynchus* sp.), and gapeworms (*Syngamus* sp., *Cyathostoma* sp.). Oocysts of *Eimeria* are also seen.

Direct smears are useful for detecting *Giardia*, coccidia, and other protozoan parasites such as *Hexamita*. Samples must be fresh. A small amount of feces is mixed (1:2) with normal saline or lactated Ringer's solution on a microscope slide. A coverslip is placed on top, and the slide is read under the microscope.

Sedimentation is the only technique that will detect fluke eggs, but it can also detect nematode eggs. A sample of feces is mixed with 1% liquid soap in water. Remove the supernatant after 5 min, refill the tube with soap and water, mix, and let stand another 5 min. Again, remove the supernatant, then spread the sediment on a microscope slide, place a coverslip on top, and read.

Generally, a low power objective (10x) is used for scanning the microscope slide. The higher power objective (40x) can be used to examine individual eggs or oocysts. The entire area of the sample on the microscope slide should be scanned for each sample, because some parasites only produce small numbers of eggs, and the presence of even one egg is diagnostic. There is no direct relationship between egg counts and number of adult parasites present. However, if samples are taken before and after treatment, egg counts are useful in providing information on the effectiveness of antiparasitic medications. The significance of intestinal parasitism for cranes is discussed under Parasitic Diseases later in this chapter.

Although hematozoa (blood parasites) are not common in captive cranes in North America, all blood smears should be scanned for these protozoa. Thick smears are better than thin smears for this purpose. Routine hematology stains are sufficient for screening, but more specific techniques (Olsen and Gaunt 1985) are recommended.

Radiology

The radiographic examination is an additional diagnostic aid. All veterinarians working with cranes should have access to radiology equipment. Radiography techniques used for other avian species or for extremities of mammals can be adapted for cranes. Techniques are available in veterinary and radiology texts and are reviewed in McMillan 1994. Radiographic manifestations, even under normal conditions, vary with species of crane, age, sex, time in captivity, and even method of flight restraint (i.e., non-flighted birds normally have atrophied pectoral and wing muscles). Traumatic injuries to the long bones and skull are the most frequent injuries seen in crane radiographs. Radiographs are also important for diagnosis of soft tissue diseases and ingested foreign bodies.

Radiology Protocol

Each institution will need to develop its own exposure chart based on its particular machine, screen, and film. Start with a general bird or cat technique chart, and use the appropriate body measurements taken from the crane being radiographed. From a log book that records patient identification, species, date, body part, positioning, measurement, exposure technique used, and comments on the technical quality of the film, you will be able to develop an exposure technique chart for cranes.

It is often preferable to use general anesthesia (optimally isoflurane gas) for radiography of cranes. Having the crane anesthetized will decrease the stress for the bird, decrease movement, and improve positioning resulting in a better quality radiograph. If anesthesia is not available or indicated, hooding the crane may be helpful. Physical restraint (without anesthesia) is routinely used for radiographs of some body areas such as legs or feet, or for quickly scanning the gastrointestinal tract (e.g., looking for ingested metal).

For an unanesthetized crane, use the shortest exposure time to minimize any problems with motion. Therefore, a high kilovolt potential (kVP) and the highest milliamperage (mA) setting is used to compensate for a short exposure time. When radiographing an anesthetized crane, there is more leeway in adjusting settings to obtain the best quality radiograph for diag-

nostic purposes. Higher kVP settings provide for a longer scale of contrast and more exposure latitude. With lower kVP settings, more subtle changes in subject density can be seen on the radiograph.

Whenever possible, take two or more views (Fig. 8.8). Normally these are dorso-ventral and lateral radiographs, but oblique images are sometimes helpful in enhancing the diagnostic value of the radiographs. However, with some body parts such as the wing of a crane, positioning can be very difficult, and the information gained from the second view may not always be worth the additional time and handling.

All but essential personnel should leave the room. Anyone present in the room should wear a lead apron, neck shield and gloves, and have an exposure monitoring badge.

To obtain a good diagnostic radiograph, **develop** it properly. If an automatic film processor is used, feed the film in the darkroom and wait for the finished film. To manually process a negative:

1. Extinguish all lights except a safe light (red) when handling unprocessed film.
2. Touch the film only by the corners as it is taken from the cassette and attached to the appropriate size hanger.

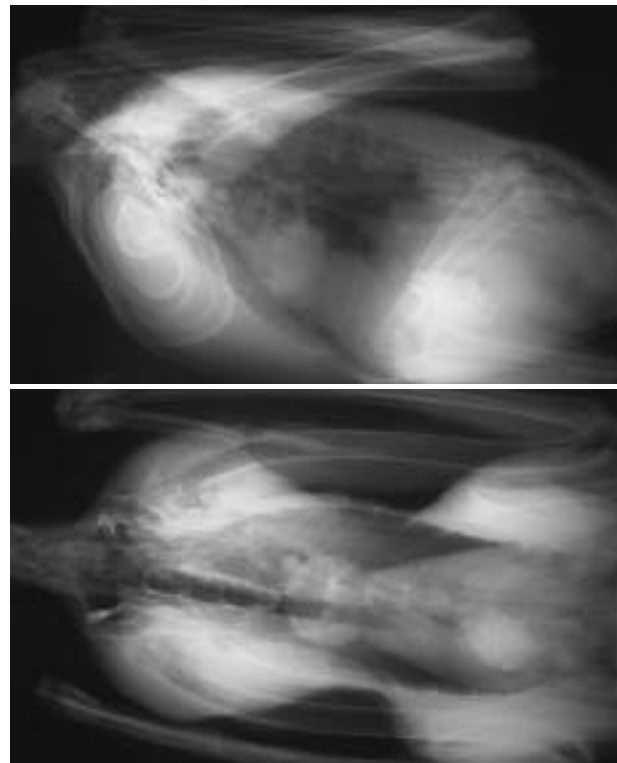


Fig. 8.8. *Lateral and dorsal-ventral view (radiograph) of normal Whooping Crane.*

Photo Glenn H. Olsen

3. Refill the radiograph cassette with the appropriate size of unexposed film. Make sure the unused film is returned to the stock box and resealed against light.
4. Take the hanger with the exposed film and place it in the developing solution. Turn on the pre-set timer. Periodically agitate the hanger gently during developing.
5. When the time has expired for developing, remove the hanger and film, and dip it several times in the wash solution (usually water). Then allow most of the wash to drain.
6. Place the hanger in the fixer solution for twice the length of time used for developing. Occasionally agitate.
7. If it is necessary to view film before drying, remove film from the fixer after the first minute and examine.
8. Remove the hanger and film from the fixer when time has expired, and place it in the wash (water) for twice as long as the time in the fixer. Then allow the film to dry on the hanger.

When storing radiographs, remove the corners with scissors so that the small puncture holes created by the hanger do not scratch other film in the envelope.

Radiographic Interpretation

Accurate interpretation of radiographs depends upon: (1) the case history of the crane, (2) findings from the physical examination, (3) radiograph quality, (4) density of the tissue or object being studied, and (5) proper positioning of the patient (take at least two views). Special techniques, such as the use of contrast media, can also help increase the diagnostic value of radiographs. Knowledge of normal anatomy is important in diagnosing abnormalities; it is helpful to keep a reference collection of radiographs of normal cranes of different species and ages for comparison. ICF has produced radiographic series on the developing legs of three species of crane chicks (Siberian, Whooping, and Florida Sandhill Cranes). Copies of these films are available at cost from ICF.

Radiographs can even be used for non-dense **carcinomas**. Osteosarcoma occurs rarely in cranes. Typical radiographic evidence includes osteosclerotic areas, osteolysis, or periosteal bone formation with some bone forming within the soft tissue. Chondroma-like lesions have been seen in wild Florida Sandhill Cranes and one Whooping Crane (M.G. Spalding, University of Florida, Gainesville, Florida, personal communication).

Infectious Diseases

Bacterial Diseases

Diseases of bacterial origin are commonly encountered in cranes. Stress can contribute to the outbreak and spread of bacterial diseases (Carpenter 1986). *Salmonella* spp., including a variety of serotypes, one being *S. typhimurium*, have been isolated from the feces of cranes (Windingstad et al. 1977; Langenberg and Dein 1992). The origin and significance of fecal *Salmonella* isolated from clinically healthy birds is not known. However, *Salmonella* can kill chicks, may affect fertility, and is a concern in birds for release into the wild. For these reasons, an effort is often made to stop or control shedding by *Salmonella* carriers. Frequently, these infections are transient and self-limiting with temporary isolation and surveillance all that is necessary. However, some infections are persistent and antibiotic therapy is useful. Birds treated with ampicillin, tetracycline, trimethoprim-sulfa, or metoprim-sulfa antibiotics, or with an experimental *S. typhimurium* bacteria, (see Table 8.1 for dosages) have tested negative on several subsequent cloacal cultures. Antibiotic therapy is controversial because it may only temporarily stop shedding or even cause a permanent carrier state.

Escherichia coli is routinely cultured from the gastrointestinal tract of both young and adult cranes in low to moderate levels and can be considered normal flora. Even crane chicks raised indoors on carpeting will have *E. coli* as part of their gastrointestinal flora by day 6 post-hatch. However, *E. coli* can be pathogenic and has been the cause of diarrhea and subsequent death in young chicks (see Chapter 5, Veterinary Techniques section).

Mycobacterium avium infections (avian tuberculosis) are widely reported in captive cranes. In fact, some feel that cranes are more susceptible than many other avian groups to this usually fatal infection found in the gastrointestinal tract, liver, spleen, and other internal organs. Clinical signs include weight loss, anorexia, abdominal organ enlargement, the presence of masses on radiographs, and an elevated WBC count. Diagnosis can be difficult: the tuberculin skin test used in poultry does not work in cranes, so a combination of laparoscopy, liver biopsy, and fecal culture are often used (Langenberg and Dein 1994). Treatment using ethambutol and rifampin have been attempted in two cases (Snyder and Richard 1994), but the success rate is not yet clear.

Other bacteria isolated from cranes include *Pasteurella multocida*, *Arizona* spp., *Clostridium* spp., *Erysipelothrix* spp., and various Enterobacteriaceae such as *Pseudomonas* sp. and *Klebsiella pneumoniae*. See Table 8.1 for suggested antibiotics and dosages for treatment.

Respiratory Diseases

The bacterial agents most often associated with pneumonia in cranes include *Pasteurella multocida*, *Yersinia pseudotuberculosis*, *Escherichia coli*, *Klebsiella pneumoniae*, *Pseudomonas aeruginosa*, *Salmonella typhimurium*, and *Streptococcus* spp. Cultures can be obtained from the trachea or choana, from sinus exudate or air sac wash, or by air sampling in fledgling cranes (Shane et al. 1986). Even a positive bacterial culture may not diagnose a cause; it is possible that the bacteria are secondary to a viral infection or associated with another respiratory problem such as aspiration or scoliosis. Viral diseases including Newcastle disease (paramyxovirus group 1) are possible. Fungal infections, especially with *Aspergillus fumigatus*, are commonly seen in cranes although usually as a secondary problem after treatment for bacterial pneumonia or in a generally debilitated bird.

Fungal infections are difficult to treat, and are often diagnosed when a crane fails to respond to standard antibacterial therapy. Although buccal contamination of cultures is normal, the first step in the treatment of a respiratory disorder is culturing for bacteria and fungi. Radiographs can also be helpful in diagnosis.

Maintain the sick bird in a non-stressful, warm (70⁰-85⁰ F, 21⁰-29⁰ C) environment. Hospitalization is recommended for the first 7-10 days. Supportive care initially consists of systemic antibiotics. Gentamicin, trimethoprim/sulfadiazine, amikacin, piperacillin sodium, or enrofloxacin (see Table 8.1 for dosages) are all recommended as initial choices until the results of sensitivity tests from bacterial cultures are received. Other possible treatments include intramuscular (IM) injections of vitamins A, D, and E; intravenous (IV) or subcutaneous (SQ) fluids; and supplemental feedings if weight falls. If aspergillosis is suspected or confirmed by culture, use antifungal therapy such as fluconazole (oral), itraconazole (oral), amphotericin-B (intravenous or intratracheal), or a combination of the latter with one of the former. Tracheal flushes and nebulization are also recommended (see Table 8.1 and 8.4). Our best results in nebulizing chicks have come from clotrimazole, first used in 1995.

TABLE 8.4

Medications used for nebulization of cranes.

Medicine	Indications	Dosage
Enrofloxacin	Antibacterial	22 mg in 10 mL saline
Gentamicin	Antibacterial	50 mg in 10-30 mL saline or distilled water
Amikacin	Antibacterial	50 mg in 10 mL saline
Tylosin	Antibacterial	100 mg in 10 mL saline
Erythromycin	Antibacterial	200 mg in 10 mL saline
Polymyxin B	Antibacterial	333,000 U in 5 mL saline
Sulfadimethoxine	Antibacterial	200 mg in 15 mL saline
Piperacillin sodium	Antibacterial	200 mg in 10 mL saline
Amphotericin B	Antifungal	5-10 mg in 15 mL water ^{1,2}
Clotrimazole	Antifungal	30 mg, do not dilute ²
Acetylcysteine ³	Mucolytic	0.25-1.0 mL 10-20% solution in 10-15 mL saline or distilled water

¹ Can use distilled or sterile water for injection.

² Use disposable pediatric nebulizer and O₂.

³ Can be mixed with other medicines.

Nebulization therapy with antibiotics is best accomplished with an ultrasonic nebulizer commonly used in human respiratory therapy (see Appendix). This unit is used with a mobile bacterial filter to remove bacteria from the air supply. Ideally, the unit should have an adjustable output chamber from 0-7.5 mL and have an alert signal to indicate when the chamber is empty. The fan should have variable speed control and a aerosol output. The nebulization therapy should total 1 hour daily, divided into 2-3 equal treatments. A 1:10 or 1:20 mixture of gentamicin (or other water soluble antibiotic) to saline or distilled water is used (see Table 8.4). The crane must be housed in a relatively airtight chamber, such as a Snyder oxygen cage, during therapy. Nebulization therapy with antifungal agents (see Table 8.4) usually requires a pediatric nebulizer and forced air supplied by a tank (O_2) or air compressor.

Nebulization therapy is effective because it brings the antibiotic/antifungal directly to the affected respiratory membrane. Because there is usually little or no uptake of drugs into the crane's circulatory system from this application, standard systemic antibiotic therapy must still be used. The humidification of the respiratory epithelium during nebulization is soothing for the patient. In addition, a mucolytic agent such as acetylcysteine (0.25-1.00 cc) can be added to each 10-15 cc of nebulization fluid to help break up mucous in the respiratory tract.

Viral Diseases

Several viral diseases have been identified in cranes including avian pox (Simpson et al. 1975) and Newcastle disease (Kaleta and Marschall 1981). Inclusion body disease of cranes (IBDC, crane herpes virus) and eastern equine encephalitis (EEE) have had a major impact on captive cranes (Carpenter et al. 1987, 1989; Dein et al. 1986). The reovirus seen in Grey Parrots (*Psittacus erithacus*) (Graham 1987) is similar to a reovirus that caused chick mortality at Patuxent.

In 1978, IBDC caused the death of 18 cranes of 4 species at ICF (Docherty and Henning 1980). Signs were nonspecific but included anorexia, lethargy, weakness, and dyspnea. The pathology was typical of herpes virus infections with an inclusion body hepatitis and splenitis progressing rapidly to death. Liver and spleen are the best tissues for virus isolation. The status of IBDC in wild cranes is uncertain. Limited serotyping on six crane species from North America, Eurasia, and Africa has found only two positive Eurasian Cranes in

southwest Russia. However, similar Herpesviruses have been isolated from mill cranes in Austria, France, Japan, China, and Russia (Carpenter 1993 and ICF unpublished data). The IBDC virus may persist in a dormant state in an infected bird making detection by virus isolation difficult. Serological testing for IBDC antibody (a virus-neutralization test) is available only at the National Wildlife Health Center, Madison, Wisconsin. There is no specific treatment for this disease. Because of the danger posed by this virus to other cranes in a collection or in the wild and the potential for a carrier state, it is usually advisable to euthanize or completely isolate infected cranes. Trans-ovarian transmission of the virus has not been seen to date, so eggs may be safely taken from antibody-positive cranes.

The EEE virus, an arbovirus (arthropod borne virus), is native to the eastern and north-central North America, parts of Central and South America, and the Caribbean Islands. The virus is primarily carried by the mosquito *Culiseta melanura*, a species which breeds primarily in hardwood swamps. Nearly all exposed native birds develop antibody titers with no morbidity or mortality. However, some Whooping Cranes and some species introduced into North America often develop clinical signs and many die.

In 1984, EEE virus killed 7 Whooping Cranes at Patuxent (Dein et al. 1986; Carpenter et al. 1987, 1989). Three of the 7 birds showed lethargy, ataxia, and neck and leg paresis, while 4 showed no clinical signs. Of the 32 surviving Whooping Cranes in this captive flock, 14 (44%) developed antibodies to EEE. Subsequent to the 1984 outbreak, an inactivated EEE vaccine for humans was found to stimulate antibody titers in both Sandhill and Whooping Cranes (Clark et al. 1987). Since 1985, all Whooping Cranes held in areas where the disease is present have been vaccinated. In addition, there is a screening program at Patuxent to detect EEE by examining mosquitos and by monitoring exposure in sentinel Bobwhites (*Colinus virginianus*) and Sandhill Cranes (Pagac et al. 1992). This program documented an EEE incident in 1989 where the virus was present in mosquitos and spread to the quail, but no losses were recorded among 55 exposed but vaccinated Whooping Cranes, leading us to conclude that protective titers were established by the vaccination program. A variety of crane species at zoos have been vaccinated with commercial equine EEE vaccines with no reported ill effects. However, Clark et al. (1987) reported that two species of cranes had a poor antibody response to equine EEE vaccines.

Parasitic Diseases

Parasites opportunistically infect stressed or crowded cranes during migration or when in captivity. Clinical signs of parasitism are usually non-specific and may include weight loss, lethargy, diarrhea, or dyspnea. Heavy parasitic infections can also cause malnutrition and increase susceptibility to other diseases (Carpenter 1979).

A parasite monitoring and control program is critical to maintain and breed healthy cranes in captivity. Therapeutic and prophylactic administration of parasiticides (Table 8.5) is an important part of any medical program. In addition, reducing pen crowding, practicing pen rotation, cleaning facilities, quarantine and treatment of new birds, and separating birds by age are important parts of a control program. Prophylactic administration of anti-parasite medications is especially useful with young chicks (see Chapter 5), cranes being prepared for release or shipment to other collections, and when introducing cranes into new (parasite-free) facilities.

Cranes can be infected by a number of species of protozoa including the blood-born parasites *Hemoproteus* and *Leucocytozoon*, but the true significance of these infections has not been documented. *Hexamita* has been implicated as the cause of enteritis and death of captive Florida Sandhill Cranes (M. G. Spalding, University of Florida, Gainesville, Florida, personal communication).

The most common and best-documented protozoan parasites of cranes are the coccidia. Both *Eimeria gruis* and *Eimeria reichenowi* (Fig. 8.9a) are common parasites of Whooping, Demoiselle, and Sandhill Cranes, and are the likely species found in other cranes. In addition, *Adelina* sp. has been found in Sandhill Cranes (Courtney et al. 1975). *Isoospora lacazei* has been found in two captive Whooping Cranes, but was thought to have been due to contamination of food by fecal matter from passerines (Forrester et al. 1978).

Cranes, like several other bird groups including geese and turkeys, have an extra-intestinal form of parasitism by *Eimeria* in addition to the gastrointestinal infection (Carpenter 1993). In this extra-intestinal form, called disseminated visceral coccidiosis (DVC), endogenous stages of the parasite disseminate from the alimentary tract throughout the body carried by the blood or possibly lymphatic systems. This results in general inflammation of organs, seen as broncho-pneumonia, hepatitis, myocarditis, splenitis, and enteritis, in addition to the formation of discrete granulomatous

nodules in any of these organs. The disease can be devastating in crane chicks under 60 days of age. It is a serious problem at Patuxent, but has not been documented at ICF where winters are much colder.

In captivity, concentrations of *Eimeria* in the soil are often much higher than would be found in the wild. The parasites have been documented in wild cranes, but the role of DVC in survival of wild cranes has not been studied. In captive colonies, several anti-coccidial medications (including amprolium and monensin, see Table 8.5) have proven effective in controlling the incidence of DVC when they are continuously mixed with the food (Carpenter et al. 1992). Intestinal coccidial infections in individual birds have been successfully treated with amprolium, trimethoprim-sulfa, or metoprim-sulfa, and sulfadimethoxine.

Endoparasites, including acanthocephalans, cestodes, trematodes, and nematodes, have been found in cranes (Carpenter 1993). The overall effect of such parasites on both wild and captive cranes is not well documented. Acanthocephalans (spiny headed worms; Fig. 8.9b) occasionally cause perforation of the intestines leading to peritonitis and subsequent death in captive crane chicks. This parasite's significance in older captive birds and in wild birds is unknown. No known treatment is available for this parasite. Because the earthworm (*Lumbricus* sp.) may be an intermediate host, rearing chicks indoors may prevent infection.

Gapeworms (*Syngamus* sp. and *Cyathostoma* sp.) have caused severe tracheitis, bronchitis, and obstruction of the trachea through irritation and formation of mucous plugs, leading to death. Signs include dyspnea and open-mouth breathing (gaping). Sometimes gapeworms can be seen in the upper trachea of a symptomatic bird. Additionally, diagnosis is possible by examining tracheal washes and occasionally by fecal flotation. Ivermectin or fenbendazole (see Table 8.5 for dosages) have been effective in eliminating the parasite. Pen rotation will help prevent reinfection. Earthworms carry gapeworm eggs, therefore decreasing a crane's exposure to earthworms will help control infection rates.

Capillarids (*Capillaria* sp. and *Eucoleus* sp.; Fig. 8.9c) and *Ascaridia* sp. (Fig. 8.9d) can cause debilitation and occasionally contribute to death. Both are readily diagnosed on fecal flotation. Treatment with ivermectin or fenbendazole (see Table 8.5 for dosages) is effective, but reinfection is frequently possible unless birds are moved at least annually to a fallow pen. Treatment success has been highest when both ivermectin and fenbendazole are used concurrently.

TABLE 8.5

Antiparasitic medications used in cranes.¹

Drug	Indications	Route of Administration	Dosage	Treatment Schedule Per Day
Anticoccidials				
Amprolium	Anticoccidial	Food	0.0125 mg/kg (Prophylactic) 0.025 mg/kg (Therapeutic)	Continuous for 2 weeks, minimum
Amprolium	Anticoccidial when other forms of this drug are not appropriate	Drinking water	0.006%	Continuous
Monensin sodium	Anticoccidial	Mixed in feed	90 ppm	Continuous or seasonally
Triple Sulfa Soluble Powder ²	Used when clinical evidence of coccidiosis	Drinking water	1.5 tsp/gal	2 days on; 3 days off; 2 days on; 2 days off; 1 day on
Trimethoprim sulfa	Used when clinical evidence of coccidiosis	Oral IM	See Table 8.1	1-2/day
Ormethoprim sulfadimethoxine	Used when clinical evidence of coccidiosis	Food	0.015% ormethoprim 0.026% sulfa	continuous for 3 weeks
Sulfa dimethoxine	Used when clinical evidence of coccidiosis	Oral	50 mg/kg	1/day for 2 weeks
Antinematodals				
Fenbendazole	Capillariasis, other nematodes	Oral	100 mg/kg	5 days, then repeat in 10-14 days
Ivermectin	Broad-spectrum	IM	0.2 mg/kg	2 doses 10-14 days apart or as needed
Levamisole	Safe, efficacious broad-spectrum anthelmintic	Oral	40 mg/kg (25 mg/kg for chicks)	Bi-weekly or as needed
Piperazine	Treating individuals or groups of cranes for ascarids	Drinking water	15-20 g/gal	3 days; repeat in 2 weeks
Pyrantel pamoate	Intestinal nematodes	Oral	4.5 mg/kg	2 doses 10-14 days apart
Thiabendazole	Wide range of antiparasitic action with a high degree of efficacy and safety	Oral	100 mg/kg	Weekly or bi-weekly as needed
Anticestodals and Antitrematodals				
Albendazole	Effective in treating some trematodes	Oral	20 mg/kg	2 doses 1 week apart
Praziquantel	Effective in treating cestodes; potentially toxic	Oral	6 mg/kg	Bi-weekly or as needed
Ectoparasiticals				
Carbaryl	Control of most ectoparasites	Topical	5% powder	Weekly or bi-weekly or as needed
Pyrethrins	Control of most ectoparasites	Topical	0.10% powder	Weekly or bi-weekly or as needed

¹ Based on Olsen and Carpenter 1996.

² Active drug ingredients: sulfamerazine sodium 27.20%, sulfamethazine sodium 27.20%, and sodium sulfathiazole sesquihydrate 29.85%.

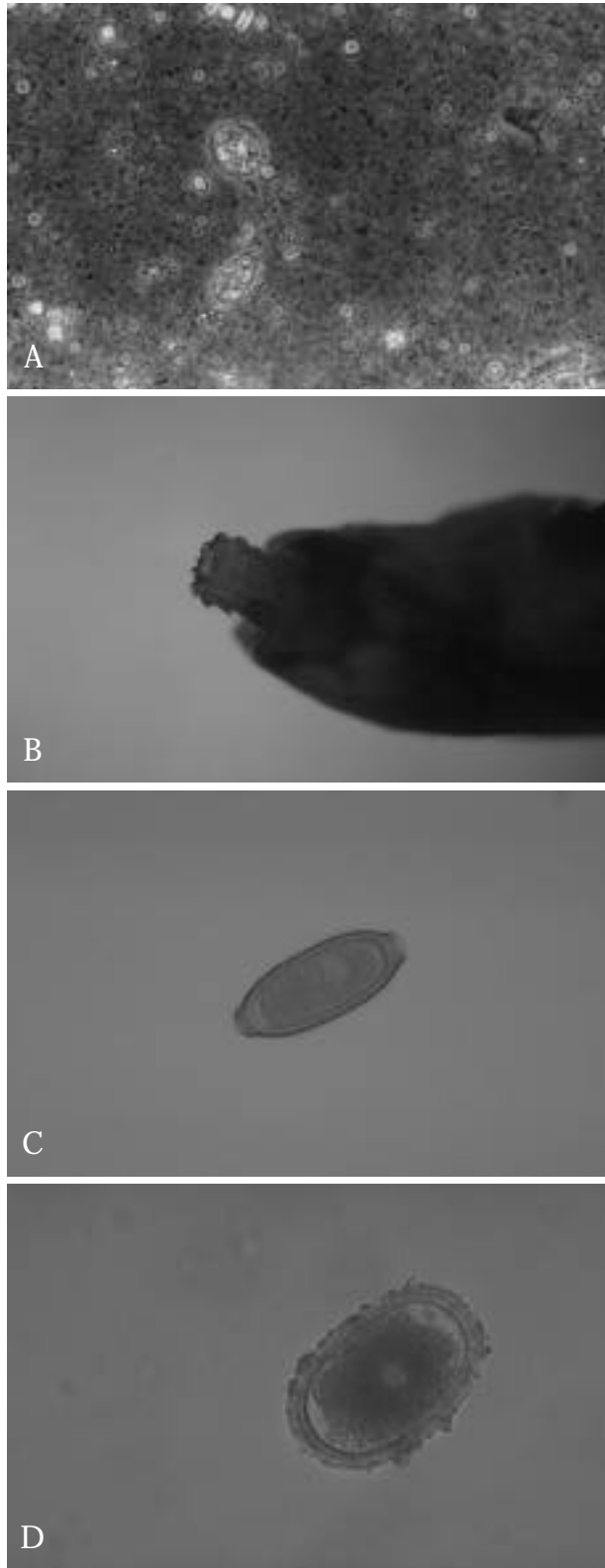


Fig. 8.9. Photomicrographs of parasites: A, ova of *Eimeria gruis* (pyriform, $10 \times 14 \mu$) and *Eimeria reichenowi* (round, $11 \times 13 \mu$); B, head of acanthocephalan (proboscis 0.5 mm); C, *Capillaria* egg ($25 \times 50 \mu$); D, *Ascaridia* egg ($50 \times 90 \mu$).

Photos Harry Danforth (A) and Glenn H. Olsen (B-D)

Ectoparasites including 5 mite species (Order Acarina) and 4 biting lice species (Order Mallophaga) (Forrester et al. 1976; Atyeo and Windingstad 1979) are seen in cranes. No pathological significance, except possibly in young or debilitated birds, has been noted in ectoparasite problems. A dusting with 5% carbaryl or 0.10% pyrethrins (Table 8.5) is very effective as a treatment. In addition, dusting can be done during an annual health check as a prophylactic measure.

Problems with biting and stinging insects such as black flies (*Simulium* sp.), bees (*Apis* sp.), and wasps (*Vespa* and others) sometimes cause minor skin irritation, excessive preening, and behavioral signs of discomfort and stress. At ICF, equine insect repellents containing pyrethrins or carbaryl have been somewhat effective (see also Chapter 11F).

Non-Infectious Diseases

Trauma

The most frequent causes of trauma will vary between institutions, but include collisions with pen structures and during capture, handling, and shipping. Even with good husbandry and excellent facilities, aggression associated with establishing dominance hierarchies, mate selection, and defense of territory, food, or water remains important (see Chapter 6; Carpenter et al. 1976; Carpenter and Derrickson 1981). **Dangerous situations** are: (1) pair formation, especially in a community (group) pen with 3 or more birds, (2) when introducing new cranes into an established social group, and (3) the escape of a crane into a neighbor's pen. Generally, the intruder will be the victim of the aggression. At Patuxent, 7.3% of the Whooping Crane deaths occurring during the 15 year period from 1966 to 1981 were from aggression (Carpenter and Derrickson 1981).

Most aggression-related injuries are to the neck and head (Fig. 8.10). Depending on how soon the crane is found, the victim will often be in a deep state of shock. **Standard shock treatment** is given: corticosteroids, fluids (IV and SQ), and antibiotics (Table 8.1) are administered. The wounds are cleaned, and following stabilization, the bird is radiographed to assess skull damage. Supportive care, especially during the first 24 hours, is critical. Patients that survive the initial trauma and the first 48 hours normally make a complete recovery, but many cranes carry scars for life.



Fig. 8.10. *Head and neck injuries due to aggression.*

Photo David H. Ellis

Occasionally, a parent will attack a chick (see Chapter 5), but not normally without an underlying cause, such as chick lethargy, the parent reacting to some disturbance, or the parent not being prepared for a chick. Normally such chicks are killed.

An important source of trauma for captive cranes is collisions with fences and buildings. Anatomically and behaviorally, cranes seem predisposed to damage their long legs, beaks, necks, and wings. Intensive management of cranes for production can exacerbate this problem. Careful attention to pen and facilities design (see Chapter 12) and capture, handling, and shipping techniques (see Chapter 2) will significantly reduce trauma cases.

Trauma to the neck, head, or beak is second only to leg and wing injuries. Neck injuries generally occur when a crane runs or flies into a pen fence or building wall. The resulting damage to the cervical vertebrae and spinal cord is often fatal, but if not, it often results in signs of ataxia, paresis, or abnormal neck position. Treat for shock using corticosteroids (dexamethasone has been used successfully in several cases; see Table 8.1 for dosage). Methocarbamol (see Table 8.1 for dosage), a muscle relaxant, was useful in one successful case (Done et al. 1993).

Ocular injuries that have been seen in captive cranes include lacerations of the third eyelid and the primary lids, and corneal lacerations, abrasions, and punctures. These injuries can be repaired with standard ophthalmic surgical procedures described for birds and mammals (Magrane 1965; Karpinski and Clubb 1986; Murphy 1987). Prophylactic treatment

with aminoglycoside ophthalmic antibiotics is recommended to prevent secondary infection.

Beak fractures are very common in captive cranes though fortunately the majority are minor. Especially during cold winter weather when probing behavior is impeded by frozen soil, fractures of the tip (1-4 cm) of either the upper or lower beak are seen. Trimming of over-long beak tips and providing safe substrates for probing help decrease this problem. More severe fractures also occur with the most common site being immediately in front of the nares on the upper beak. It is unclear why breaks at this site are common. Initial treatment includes control of hemorrhage and therapy for shock. Options for surgical repair are described under Common Surgical Procedures, this chapter. Regrowth of the upper beak will be minimal when the fracture is more than 3-4 cm from the tip. However, fractures of the lower beak 8 cm (less in small-billed species) from the tip can regrow. Providing visual barriers between cranes and human activity and between crane pairs will decrease the behavior at fence lines that puts cranes at risk for beak damage.

Ruptured Air Sacs

Ruptured air sacs are infrequently seen in cranes. Adult cases are generally minor; chick cases can be more serious. In most cases, handling trauma was the suspected cause. Some cases resolved without medical intervention; some involved extensive subcutaneous emphysema on the thorax, neck, and head (Fig. 8.4). For these, air was withdrawn using a syringe and needle several times from several different locations over the body, but each time the condition would return within 24 hours. Finally, latex drains (6-13 mm diameter tubes) or setons (narrow gauze loops) were surgically inserted through the skin and into the air spaces. These birds were given antibiotics and the drains or setons were cleaned with a 1% povidone iodine solution twice daily. Within 10 days, the birds returned to normal and the drains were removed.

Osteomyelitis

Bone infections can occur secondarily in open fractures contaminated before or during surgical procedures or in pododermatitis (bumblefoot). In the early stages, osteomyelitis is not evident on radiographs, but rather shows up some days later. Usually the first evidence is a hazy appearance of the bone

structure with some roughening of the trabecular outline. As the condition progresses, evidence of bone absorption and reactionary bone formation appear simultaneously. The periosteum may be elevated with some new bone formation occurring underneath (Fig. 8.11). Osteolysis may occur, especially if the infection is in conjunction with a foreign body such as a bone screw or intermedullary pin. Signs of possible spreading of the infection include widening or increase in size of osteolytic area, active periosteal response, loss of joint space, or soft tissue swelling. **Avian tuberculosis** is known to occur in several of the wild populations of North American cranes, and any bird from these flocks showing limb problems should be radiographed to search for bone changes similar to osteomyelitis or neoplasia.

Treat osteomyelitis with antibiotics or antifungals chosen by culture and sensitivity testing (Table 8.1) and with surgical debridement (removal of necrotic tissue). Radiographically, response to treatment is indicated by cessation of osteolysis and gradual redevelopment of normal bone structure (Douglas and Williamson 1970:44). The most common complications are delayed union of fracture sites or bone sequestra. Sequestra are seen as areas of bone separated from other nearby bone by a radiolucent (non-bony) zone. Sequestra are necrotic pieces of bone and are best demonstrated by lack of any change in appearance on serial radiographs.

Arthritis, reportedly uncommon in birds (McMillan 1988), when seen in cranes is secondary to trauma, infection, developmental limb problems, or articular gout. Iatrogenic arthritis is one sequel to carpal tenotomy often used to limit flight capability in crane colonies.

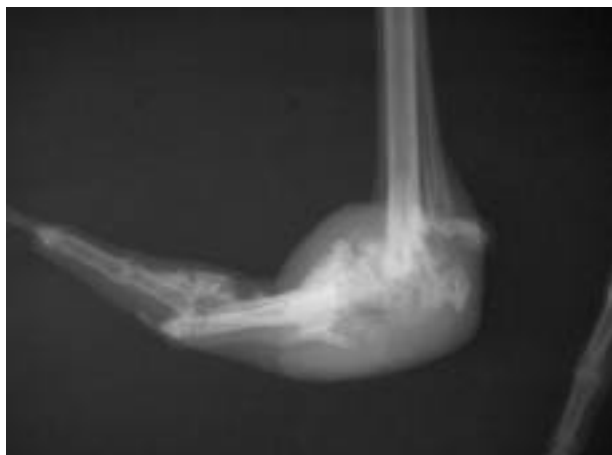


Fig. 8.11. *Osteomyelitis (radiograph).*

Photo Glenn H. Olsen

Egg Binding

Egg retention or egg binding is a rare condition usually associated with an abnormally formed egg (soft-shelled, abnormally large, or abnormally small), or a bird that has an abnormal pelvis or cloaca, is in poor condition, is hypothermic, or has low blood calcium. Occasionally an infection can lead to a retained egg. Clinical signs of egg binding include straining, lethargy, or depression. The egg is usually readily palpable in the caudal abdomen.

Initial therapy consists of providing a warm, quiet environment for the bird and lubricating the cloaca with surgical lubricant (see Appendix) or petroleum jelly. Medications should include fluid therapy, oxytocin, and intramuscular calcium (Calphosan, see Appendix) (see Table 8.1 for dosages and routes of administration). Occasionally, sedation is useful to relieve oviduct spasms. Antibiotic therapy should be considered in all cases where infection or contamination of the oviduct is suspected.

If the egg can be gently manipulated from the oviduct, this should be done. Breaking an egg is not recommended as the sharp fragments can lacerate the oviduct. If the egg does break, as many pieces as possible should be removed through the cloaca. If the egg is retained high in the abdomen (i.e., palpable for two days without movement), an exploratory laparotomy with surgical removal of the egg may be necessary.

Disorders of the Cloaca

Straining from egg retention, constipation, vent irritation, or diarrhea occasionally results in a prolapsed cloaca, oviduct, or part of the large intestine. Cloacal prolapses are seen in chicks with and without diarrhea. In some cases, stress is considered a factor. If possible, determine which organ is prolapsed and treat the cause before replacing the cloaca. Be sure to differentiate a prolapse from a protruding growth in the cloaca.

With a mild prolapse, it may be possible to replace the prolapsed tissues after lubricating them with surgical jelly. More severe cases may require bathing the tissues with a hypertonic solution (such as 50% dextrose) to reduce swelling prior to replacement. A gloved finger or smooth tube (syringe without needle) may help to reduce the prolapse. A purse-string suture may be required for several days to keep the tissues from prolapsing again. Once the purse-string suture is

in place, the crane must be watched carefully to make sure it can defecate normally. In laying females, a purse-string suture in the cloaca should be left in place only until about one day prior to oviposition. The underlying cause of the prolapse should also be treated. In severe cases, it may be necessary to remove necrotic tissue or amputate part of the prolapsed organ. Prognosis is guarded in these cases.

In some species, a tendency to prolapse appears to be inherited (Macwhirter 1987) although this has not been documented in cranes. Other cloacal abnormalities seen in cranes include a papilloma-like growth in a Sarus Crane and seasonal inflammation and vent soiling in laying females, for which no causative agent has been found.

Neoplasia

Several types of neoplasia have been reported in cranes including renal adenocarcinomas (Montali 1977; Decker and Hruska 1978), renal carcinoma (Montali 1977), lymphocytic leukemia (Montali 1977), granulocytic leukemia (Wei et al. 1986), and metastatic cholangiocarcinoma (Allen et al. 1985). A hematopoietic stem cell neoplasm occurred in one Florida Sandhill Crane at Patuxent. There is a higher incidence of adenocarcinomas in wild Mississippi Sandhill Cranes than in captive birds, but the cause of this situation is still under investigation.

Chondroma-like lesions have been seen in wild Florida Sandhill Cranes and one Whooping Crane (M. A. Spalding, University of Florida, Gainesville, Florida, personal communication). Generally the incidence of neoplasia in both captive and wild cranes appears to be low.

Toxicology

Only a few cases of crane morbidity or mortality due to specific toxins have been reported. A likely assumption is that most substances that are toxic to birds in general are also toxic to cranes. Cranes are considered to be low on the food chain, and are therefore not as likely to be affected by toxic compounds through biomagnification (Mullins et al. 1978). However, cranes are relatively long-lived birds and therefore have the opportunity to slowly accumulate significant amounts of persistent chemicals.

Examination of two Whooping Crane carcasses and one embryo for DDT, DDD, DDE, and dieldrin demonstrated very low levels in all tissues (Lamont

and Reichel 1970). **Pesticide levels** in fat from Sandhill Cranes from Florida and Nebraska were low (Lewis 1974). Samples from the same study in Texas contained high levels of heptachlor epoxide and smaller amounts of DDT, DDE, and dieldrin. Oklahoma samples one year had elevated DDT, DDE, dieldrin, and heptachlor epoxide levels. However, the next year, samples from the same Oklahoma site showed only low levels of DDE, with other pesticides not detected. Mullins et al. (1978) found low levels of pesticides DDT, DDD, DDE and dieldrin, and low levels of heavy metals, lead and mercury, in Greater Sandhill Cranes and eggs from Oregon and Idaho. Mercury levels were significantly higher in breeding age birds compared to eggs and young, indicating possible accumulation with age.

In 1989 and 1990, 58 Sandhill Cranes collected in Nebraska (as powerline mortalities) were analyzed for organophosphate and carbamate compounds and inorganics (Fannin 1992). Heptachlor epoxide, oxychlorane, DDE, and hexachlorobenzene were found in liver tissues. From opportunistic sampling of Whooping Crane carcasses and eggs since the 1960's, Lewis et al. (1992) reported that while DDT and mercury levels have declined following banning use of these substances as pesticides and fungicides, other compounds such as chlorinated hydrocarbons persist at low levels. Trace elements including aluminum, arsenic, cadmium, chromium, copper, selenium, and zinc were found at levels high enough to justify further monitoring.

Famphur, an organophosphate (o,o-dimethyl o-<p-(dimethylsulfamoyl) phenyl>phosphorothioate), was found at 69 ppm in the digestive tracts of two dead Sandhill Cranes in Gilmar County, Georgia (White et al. 1989). The brains from these two birds showed a 75% reduction in cholinesterase activity attributed to the effect of famphur. Organophosphorus compounds inhibit cholinesterase in the nervous system, disrupting synaptic transmission of nerve impulses. Death is usually the result of asphyxiation associated with failure of the brain respiratory center. Organophosphates and carbamates are known to be extremely toxic to wildlife, especially birds (Zinkal et al. 1978; Hill and Fleming 1982; Henny et al. 1985).

Both wild and captive Sandhill Cranes have died from **lead poisoning** when accidentally exposed. Mullins et al. (1978) found lead in samples of Greater Sandhill Cranes and eggs from Oregon and Idaho. Occasionally individual cranes, especially juveniles, have comparatively high levels (Franson and Hereford

1994). The source of lead has varied, but includes lead-based paints (Kennedy et al. 1977) and lead fishing weights and bullets (Windingstad et al. 1984). One wild Whooping Crane died after ingesting a small plastic encased battery or fish sinker (Snyder et al. 1992).

Zinc toxicosis has been seen in captive Whooping and Red-crowned Cranes after ingestion of zinc-containing metal objects, principally wire clippings from fence construction and zinc alloy coins. The clinical signs include depression, weakness, and lethargy. Recovery was rapid after surgical removal of the metal.

Mycotoxins produced by *Fusarium* sp. molds have caused death in wild and captive cranes. Between 1982 and 1987, an estimated 9,500 Sandhill Cranes in Texas and New Mexico died from an unspecified mycotoxin found on unharvested peanuts (*Arachis hypogaea*) (Windingstad et al. 1989). Cranes were observed standing or flying but unable to hold their necks straight or erect. Lesions observed at necropsy included multiple muscle hemorrhages and submandibular edema. Peanut-associated mycotoxicosis has also been seen in Demoiselle and Eurasian Cranes in India (ICF unpublished data). In 1987, deoxynivalenol toxicity, a grain-based mycotoxin present in pelleted feed, resulted in 4% (15 cranes) mortality and 80% morbidity at Patuxent (Olsen et al. 1995). Bioassays using quail or less valuable cranes are now in use with larger crane flocks to help detect unsuitable commercial feeds before they are fed. Carefully monitor food during long periods of warm, rainy weather.

Botulism, a paralytic disease caused by ingestion of *Clostridium botulinum* toxin, has killed cranes in at least one North American zoo. The toxin is produced anaerobically with the typical source being the decay of submerged carcasses of small animals. This kind of poisoning should be considered whenever cranes are housed in naturalistic water exhibits. Pay careful attention to water quality and the health of waterfowl using the exhibit. A commercial *Clostridium botulinum* Type C bacterin-toxoid has been used to protect cranes (Cambre and Kenny 1993).

Capture Myopathy

Capture (or exertional) myopathy, reported several times in cranes (Brannian et al. 1981; Hartman 1983; Windingstad et al. 1983; Carpenter et al. 1991), has been associated with trapping and restraint. It has

also been seen in captive cranes after serious, traumatic injuries or after prolonged restraint in a sling. Predisposing factors described for other species (Boever 1986) and most likely applicable to cranes include fear, anxiety, overexertion, repeated handling, transportation of an exhausted animal, prolonged transportation, constant muscle tension, and restraining the bird with muscles cramped in unusual positions or for a prolonged time. In some species of mammals, metabolic acidosis may play an important role in capture myopathy (Harthorn and Young 1974).

Clinical signs can range from peracute death due to cardiac failure, to painful, stiff movement, and swollen, hard muscles that are warm to the touch, and secondarily, trauma to limbs as the animal struggles. If the bird has survived for some days, there will be a reduction or loss of subcutaneous and abdominal fat. Serum chemistry levels, particularly creatinine kinase, lactic dehydrogenase, and aspartate aminotransferase, are often highly elevated and are useful in assessing the severity of changes in muscle tissue (Harthorn and Young 1974; Brannian et al. 1981). Elevation in uric acid values associated with renal failure can result from increased lactic acid production, myoglobinuria, or impaired mobility and subsequent dehydration.

Gross lesions consist of numerous pale, streaked areas in the skeletal muscle and heart. Renal lesions, such as urate nephropathy, are also common. Pale mottled kidneys are described in one case involving a Greater Sandhill Crane (Windingstad et al. 1983), and urate deposits were described on the kidneys of an East African Crowned Crane (Brannian et al. 1981). Microscopically, lesions are characterized by extensive areas of myocardial and skeletal muscle degeneration and necrosis, and secondary inflammation.

Prevent capture myopathy by minimizing handling, by properly handling and transporting cranes (see Chapter 2), and by maintaining adequate levels of vitamin E and selenium in the diet. Treatment of the condition is supportive and consists of intravenous fluids, corticosteroids, antibiotics, vitamin E, selenium, and good nursing care. It may be necessary to keep the bird in a sling. Physical therapy is also important for recovery. If blood pH levels are below normal, the crane should be treated with intravenous sodium bicarbonate. If blood pH levels are unavailable, but acidosis is suspected, sodium bicarbonate may be administered IV at the rate of 4-6 mEq/kg body weight.

Orthopedics

Leg, Foot, and Toe Disorders

Fractures, and less frequently **dislocations**, are common in cranes. Fractures are usually associated with trauma. Pathological fractures associated with nutritional imbalances are not generally seen in captive cranes fed a formulated pelleted diet. Likewise, the occurrence of secondary nutritional fractures in wild cranes has not been documented. Fractures should be evaluated for location, articular involvement, bone density, periosteal response, and soft tissue involvement (McMillan 1988, 1994). Generally, mid-shaft long-bone fractures have better prognosis than fractures close to the bone ends or involving articular surfaces. Most long-bone fractures of cranes require surgical fixation (Fig. 8.12). Some fractures of pneumatic bones result in subcutaneous emphysema in and around the fracture site. Tendon and ligament injuries around the hock and foot are also seen. These are difficult to diagnose and require weeks or months to heal.

The **healing of crane bones** is similar to the process seen in other birds and mammals (Bush et. al. 1976). Healing in uncomplicated cases occurs in 3-8 weeks with pneumatic bones generally healing slower than medullary bones. Osteoporosis associated with disuse is a possible complication. Often some form of physical therapy is required for a crane to begin using a limb after a fracture has healed.

Leg injuries are common in cranes (Carpenter 1986; Curro et al. 1992; Olsen 1994), and include long

bone fractures, stifle and tibial luxations, fractures and luxations of toes, spraddle legs, lateral rotation of the tibiotarsus, crooked or curled toes, and perosis. In chicks, rapid weight gain has been implicated, as have hatching problems and trauma. Exercise is a large factor in leg development. Crane chicks reared by parent cranes suffer much fewer leg abnormalities than do crane chicks raised by hand (see discussion of crane chick leg disorders in Chapter 5). Diets containing low levels of methionine or sulfur amino acids help reduce rapid growth and associated leg abnormalities in crane chicks (Serafin 1980, 1982). Correction of **crooked or curled toes** has been accomplished with splints (see Figs. 5.18 and 5.19).

Progressive osteoarthritis, especially in the hock joint of the legs, is seen in older cranes often after a long history of recurring mild lameness. Mineralization of tendon sheaths around the hock is a common radiographic finding. Medical therapy includes non-steroid anti-inflammatory drugs such as phenylbutazone and piroxicam to decrease the pain. Intra-articular and intramuscular injections of polysulfated glycosaminoglycans (Adequan) have been found ineffective in severe cases.

Bumblefoot (Pododermatitis)

Crane bumblefoot (pododermatitis) is an inflammation of the foot often associated with a bacterial infection (Fig. 8.13). Bumblefoot may start as pressure necrosis of the footpad, in many cases due to unequal weight bearing on the foot due to a lameness of the other leg. Generally, bacteria enter the foot by two routes, the first being an acute wound or laceration of the integument of the foot; the second route is

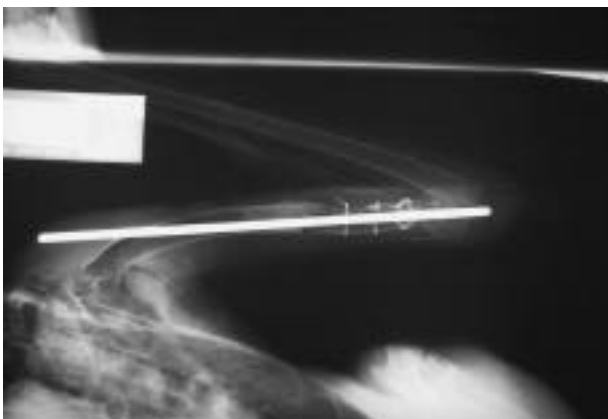


Fig. 8.12. Long-bone fracture (radiograph) showing internal fixation.

Photo Glenn H. Olsen



Fig. 8.13. Bumblefoot, Class IV-V

Photo Glenn H. Olsen

through the cracking or sloughing of dead (devitalized) skin or scales that expose the underlying tissues. The disease is progressive. Infection can spread into the tendons and bones of the foot and can be debilitating. Rarely, infections spread elsewhere in the body.

A classification system developed for use in raptors (Remple 1993) is also applicable to crane bumblefoot. Five classes are used to describe the progressive course of the disease and to grade prognosis. Class I represents the early inflammatory response to a lesion and is characterized by tissue bruising or callus (hyperkeratotic reaction) with an excellent prognosis for recovery. In Class II, there is infection in the underlying tissues, localized loss of blood supply and death of tissue (ischemia and necrosis). With modern developments in treatment, prognosis is now good. Class III is characterized by extensive infection and gross inflammatory swelling characterized by serous, fibrotic, or caseous tissue reaction; prognosis is good to guarded. Class IV represents lesions with infection and swelling of underlying tissues resulting in inflammation of the ligaments or tendons (tenosynovitis), joints (arthritis; Fig. 8.13), or bone (osteomyelitis; Fig. 8.11); prognosis is guarded to poor. In Class V, the lesions of Class IV have worsened resulting in deformity, crippling of the limb, and loss of normal function; prognosis is grave.

Treatment should start with correcting any management problem or concurrent leg injuries contributing to the bumblefoot. Abrasive surfaces, such as concrete, can lead to foot injury. Medical care includes ensuring good nutrition through adequate diet plus vitamin and mineral supplementation. Vitamin A injections are given (30,000 IU/kg body weight or 1.0 mL/kg of Aquasol A intramuscularly once weekly). Surgery under general anesthetic is recommended in severe cases. The goals of surgery are to debride (cut away) necrotic and infected tissues to reduce antigen load. Skin edges should be brought together to enable first intention healing to occur (Riddle 1980). Prior to surgery, the site is thoroughly scrubbed with a stiff brush. After incising the skin, a culture for bacterial and fungal identification is taken from the deep tissues. If surgical debridement is not done, a sample for culture can be collected by fine-needle aspiration. Remove fibrotic and infected exudate and irrigate the wound with chymotrypsin and a broad-spectrum penicillin product such as piperacillin (Riddle 1980). Close the wound with a

4-0 or 3-0 nylon, simple interrupted suture pattern or use vertical or horizontal mattress sutures to relieve tension.

After surgery, the foot is kept bandaged until the wound is healed. If the surgical wound is on the plantar (bottom) surface of the foot, the bandage should be designed to reduce pressure on the surgery site and surrounding inflamed tissues. In raptors, a preformed styrene cast or ball bandage is used to accomplish this (Remple 1993). Variations on these techniques, modified for the anatomy of the crane foot, have been used with varying success. Keeping the bandage and surgical site dry can be a problem with cranes, especially those housed outdoors. A top layer of waterproof tape helps. Antibiotic therapy should begin before surgery or, if not before, immediately after surgery. A broad-spectrum penicillin, such as piperacillin, is recommended, often in combination with an aminoglycoside such as amikacin (see Table 8.1 for dosages). Antibiotic therapy should be modified based on the culture and sensitivity results, and should continue for a minimum of 14-21 days post surgery. The crane should be kept on a soft surface such as grass, padded indoor-outdoor carpeting, or deep bedding.

Bandaging and Splinting

Bandaging or splinting is frequently used in the treatment of orthopedic problems. To minimize stress, a bandage or splint should be completed as quickly as possible and should be as unobtrusive and lightweight as possible but still be adequate to protect and stabilize the body part. If the crane becomes agitated during the bandaging/splinting process, use sedation or a general anesthetic.

To **avoid damaging feathers**, especially remiges and rectrices, use Vetwrap (see Appendix, but avoid red or other brightly colored Vetwrap) or other self-adhesive tapes that do not adhere to the feathers. Masking tape or autoclave tape can be used, but they tend not to hold well to feathers, especially when wet. Adhesive tape should only be used if the feathers are first wrapped with gauze. On the scaled portions of the leg, adhesive tape is acceptable. In some situations, a layer of waterproof tape over the bandage is helpful to keep the underlying bandage dry. Check bandages and splints frequently to detect swelling, irritation, slippage, or removal by the crane. Bandages in young, growing birds should be changed every 48 hours.

The **Figure-8 bandage** (Fig. 8.14) is useful for fractures of the radius, ulna, and hand, or to support the wing during recovery from soft tissue injuries or developmental abnormalities. This bandage can be used alone, or if the bone ends are severely malaligned, in conjunction with internal fixation. The immobilization provided by the Figure-8 bandage can result in a temporary or permanent stiffening of wing joints preventing normal flight. For release birds, surgical fixation techniques are recommended to increase the chance of normal flight. Humerus fractures usually require internal fixation, and a temporary Figure-8 bandage and body wrap to limit wing movement.

In cranes, broken legs often result in death. **Leg splints** may be useful in simple and open (compound) fractures of the tibiotarsus and tarsometatarsus (Olsen 1994). Leg splints are used alone or in conjunction with internal fixation. External fixators (Fig. 8.15a) have been used with both tarsometatarsal and tibiotarsal fractures with some success. Femur fractures, rare in cranes, generally require internal fixation (Howard 1990), although a Type I Kirschner Ehmer apparatus has also worked.

Some other leg splinting techniques (Figs. 8.15b and 8.16) are hinge splints and Schroeder-Thomas splints for both tibiotarsal and tarsometatarsal fractures. Spoon splints (usually with the spoon-end removed), fiberglass, and plaster splints are also used. Because the joints above and below a fracture site

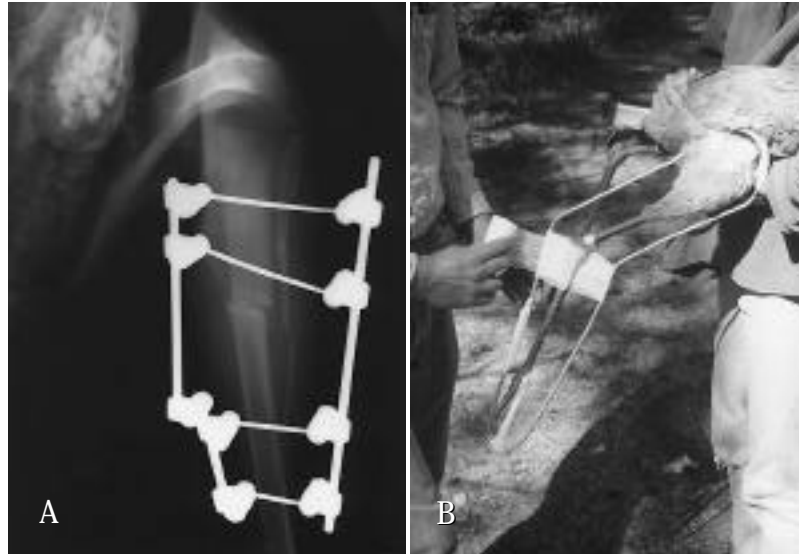


Fig. 8.15. *Two types of splints: A, Type II Kirschner Ehmer splintage for fracture of tibiotarsus in 41-day-old crane chick; B, Thomas splint (note wire makes a complete loop around thigh).*

Photos Glenn H. Olsen (A) and David H. Ellis (B)

should be immobilized during the healing process, the crane should not be allowed to move about normally.

Leg prostheses have been successfully used in four cases of amputation below the hock for irreparable tarsometatarsal fractures. In one case, a custom-designed “human” orthotic was used; in the others, a section of PVC pipe was attached to the stump. Custom fitted wood and bamboo prostheses have also been used.

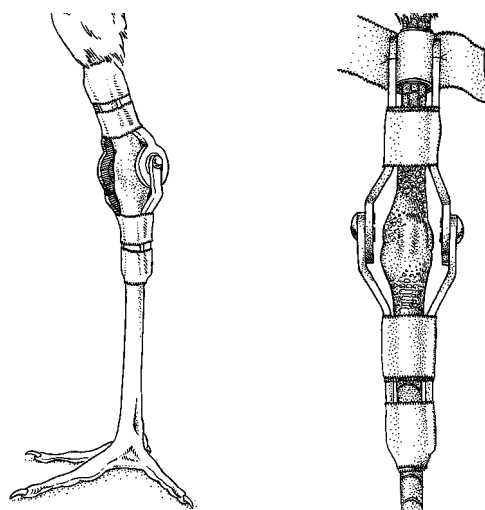


Fig. 8.16. *Hinge splint: The use of a hinge brace to provide medial-lateral support to joints with soft tissue injuries such as ligament tears has proven successful in the medical management of this type of injury in cranes (P. Klein, Humane Society, Washington, D.C., personal communication).*

Art Kate Spencer



Fig. 8.14. *Figure-8 bandage.*

Photo David H. Ellis

Because of crane anatomy, a flat or “shoe” type of splint is required to correct **phalangeal fractures** (Fig. 5.19), luxations, or to control knuckling during recovery from tendon or nerve injuries. Satisfactory **toe splints** are made of bent aluminum rod and of reheatable plastic splint material (Hexcelite, Orthoplast, or Polyflex II). Often the whole foot is encased in the splint, with a spine of splint material extending up the tarsometatarsus to help stabilize the foot. Splints for curled digits in crane chicks are discussed in Chapter 5.

When a serious leg or spinal injury occurs, supplemental support for the crane may be provided by a rigid framework with a **cloth sling** (Fig. 8.17). Some institutions use a cloth sling suspended by ropes from the ceiling. Cranes confined to slings often refuse to eat or drink, necessitating nutritional support by tube feeding. Some continually struggle with the constraining support system and require light sedation (midazolam, diazepam) to prevent self-injury. After removal of the splint or cast, another period of intensive management and therapy is required (Olsen 1994), and survival/success rates for cranes in slings are low.



Fig. 8.17. Lorie Shaull attends a Sandhill Crane suspended in a sling.

Photo David H. Ellis

Anesthesia

Gas anesthesia, using isoflurane, is the best technique for sedation or surgical anesthesia in cranes (Ludders et al. 1989). Induction and recovery are rapid and smooth which is critical for the safety of the crane and handler. Halothane can also be used, but has been associated with a higher incidence of cardiac and respiratory problems. Although pre-anesthetics such as

midazolam or tiletamine-zolezepam (Table 8.1) can be used, cranes are generally induced by mask and then intubated (2.5–6.0 uncuffed endotracheal tubes). Injectable anesthetics such as tiletamine-zolezepam or ketamine combined with diazepam, midazolam, or xylazine (Table 8.1) can be used, but respiratory and cardiac complication rates are higher and the crane must be monitored carefully and preferably held in a small padded room during recovery. Yohimbine has been used on several occasions to speed recovery when xylazine has been used. Local anesthesia in cranes is possible using small amounts of lidocaine (0.25–0.5 mL in adult cranes) or another local anesthetic.

Common Surgical Procedures

Laceration Repair

Lacerations are common in cranes. Cranes occasionally cut themselves (especially in the neck) with their sharp toenails (especially the inner nail) when they struggle during capture. This injury occurs 2–3 times each year among the 100+ crane chicks raised at Patuxent. Lacerations also occur from sharp objects in the pen and from aggressive pen-mates. Common sites for lacerations include the head, dorsal neck, carpal area, and the legs. Local or general anesthesia is used during repair of lacerations.

First, control hemorrhaging with compression on the wound site. For small wounds, ferric chloride hemostatic powder (Macwhirter 1987), ferric subsulfate, or Monsell's solution will stop bleeding. After the hemorrhage is controlled, gently pluck body feathers (not remiges or rectrices) around the wound site if needed, but avoid restarting the hemorrhage. Clean the wound with a dilute solution of antiseptic such as povidone iodine (1%), warm saline solution, or chlorhexidine. If the wound edges are not fresh, they should be debrided (cut back to fresh tissue). Suitable suture material can include 3-0 or 4-0 nylon or similar sized polyglycolic acid absorbable thread. Suture patterns generally reflect the nature and extent of the laceration with simple continuous, simple interrupted, and horizontal mattress patterns (Fig. 8.18) being the most common used in cranes. Cranes sometimes try to remove bandages with their bills, so reinforce accordingly. Non-absorbable sutures can be removed after 10 to 14 days; absorbable sutures are normally left in place.

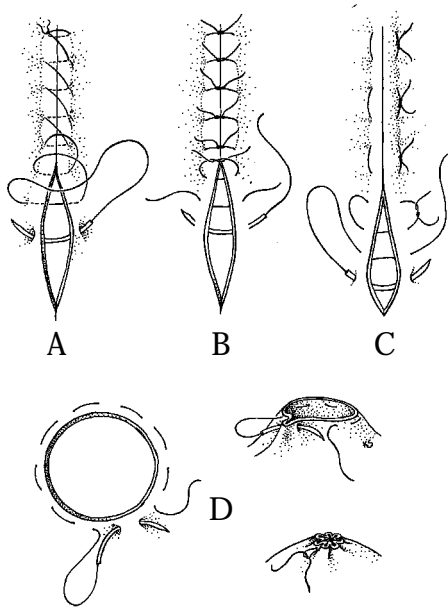


Fig. 8.18. Sutures: A, simple continuous; B, simple interrupted; C, horizontal mattress; D, purse string. Art Kate Spencer

Repair of Fractured Beaks

Minor fractures of the tip of a beak can be managed without anesthesia in two ways. The beak can be trimmed at the point of fracture. Trimming works well for fractures occurring on the distal 2-3 cm of the beak, but may result in profuse bleeding. Alternately, the fracture can be stabilized with cyanoacrylate glue (surgical glue, "super glue") while the blood vessels constrict. After 2-5 days, the fracture fragment is trimmed off. Cyanoacrylate glue or dental acrylics can also be used to provide homeostasis and protect the stump of a fractured beak. If only the upper or lower beak is fractured, the protruding section of the unfractured beak is gradually trimmed back using a hand-held grinding tool (Dremel tool; see Appendix) or surgically removed using radio-cautery for hemostasis (Ellman radio surgery unit; see Appendix).

More serious fractures require surgical management under general anesthesia. One method of repair uses self-curing dental acrylics or hoof acrylics (Fagan 1982; Altman 1984; Frye 1984; Wolf 1985; see Appendix). The fracture site is cleaned, and any open wound is treated. The fracture is then manually aligned, and the beak covered with a layer of acrylic (Fig. 8.19). Because acrylics generate heat when curing, we sometimes surround the acrylic with cold packs to minimize damage to soft tissues. Unless the fracture site is very stable, the acrylic material will require reinforcing

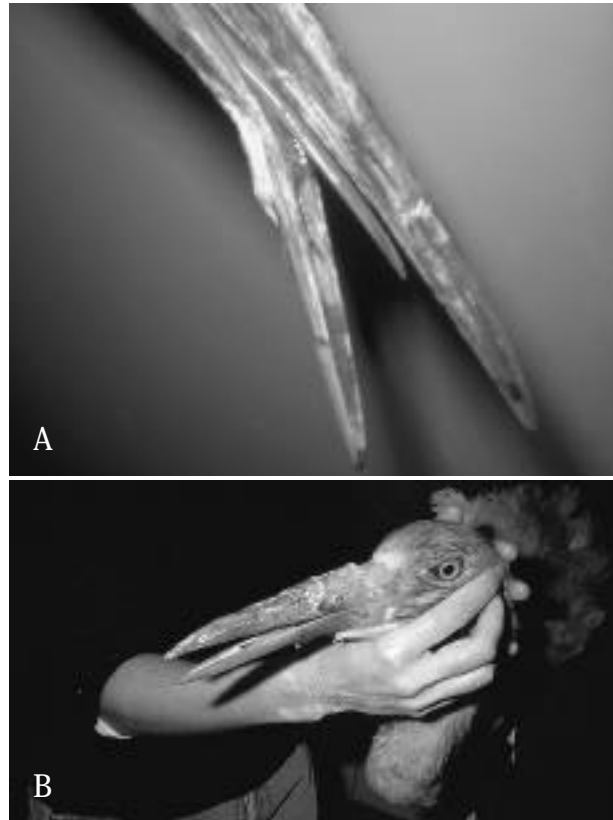


Fig. 8.19. Crane beak repairs: A, fractured lower mandible before repair; B, fractured maxillary after repair with dental acrylic.

Photos Glenn H. Olsen

with Kirschner wires or stainless steel plates. These are applied longitudinally along the sides, top, or bottom of the beak, over the first layer of dental acrylic, and held in place with another layer of acrylic. For added stability, one or more Kirschner wires are implanted at right angles through the acrylic splint and beak. The final step in the process is to remove sharp edges by smoothing and shaping the outside surface of the acrylic with a high-speed, hand-held grinder (Dremel tool). Acrylic splints are generally left on the beak for 4-6 weeks.

Other techniques that have been used for beak repair include bone plating, Kirschner-Ehmer-type external fixation, and intramedullary pinning (Howard 1989). Frequently, cranes will not eat for the first day or two after surgical repair. Feeding through a pharyngostomy or esophagostomy tube may be less stressful for these patients than repeated oral tube feeding and handling of the beak. Rates of successful repair with severely displaced fractures have been low, especially in species such as the Siberian Crane which does not stop using its beak for probing after surgery.

In cases where the fractured beak fragment has been lost, beak prostheses have been designed using moldable acrylics and attached with intramedullary pins, wire, or acrylics (Greenwell et al. 1989). In cranes, a beak prosthesis generally needs to be replaced every 3-6 months due to the wear associated with the crane's probing activities. For these reasons, a beak prosthesis is probably more appropriate for an exhibit crane, not a breeding crane. Several captive cranes have fed normally and have survived for years in captivity with beaks shortened unilaterally or bilaterally by as much as half the length of the normal beak (Howard 1989).

Endoscopic Examinations

The first avian endoscopic examinations (using rod-lens systems) were in the early 1980's (McDonald 1982) for determining sex in monomorphic psittacines. The technique is useful in cranes both for determining sex and for diagnosing a variety of abdominal and respiratory disorders. A rigid endoscope (1.9-2.7 mm outside diameter, 30° view, 17-19 cm length; see Appendix for source) is most useful. A 150-watt light source is attached via a flexible fiberoptic cord. A system using a handle-mounted, battery pack (ophthalmoscope/otoscope handle) with a focusing ocular piece on a rigid tube (Medical Diagnostic Systems, see Appendix) has proven useful in some field situations and is about one sixth the price of the rod-lens endoscopes. The disadvantages of this more portable system are reduced light transmission and poorer optics. Flexible endoscopes have also been used for gastrointestinal and tracheal examinations and for foreign body retrieval from the upper gastrointestinal tract and trachea (Howard et al. 1991). A human bronchoscope with extra channels (i.e., for insertion of other instruments) is recommended. We have used such units coupled with laser cautery to remove tumors from the accessible portions of the gastrointestinal tract and the trachea.

The **endoscope is sterilized** before use. One method is to expose it to ethylene oxide gas. After exposure to the gas, the instrument needs to be aired for 8-12 hours before use. Ethylene oxide also poses a human health hazard and manufacturer's safety recommendations should be followed. A second, safer and easier, method is to soak the endoscope for 15-20 min in 2% glutaraldehyde (Glutarex, see Appendix). Soaking for more than 2 hours may damage optics.

After soaking, the endoscope is rinsed with sterile, distilled water prior to use. Two separate rinses of 3-5 min each are recommended. Basic endoscopy is described in Taylor (1994).

The site is prepared for the surgical procedure after the patient is anesthetized (isoflurane and ketamine have been successfully used for endoscopic examinations). We strongly advise against performing endoscopic examinations on physically restrained, but non-anesthetized cranes.

There are at least six commonly used **endoscopic entry sites** on each side. Choice depends on the organ system to be view. The sites are: (1) the ventral midline or just lateral to the midline at the posterior margin of the sternum for examination and biopsy of the liver, (2) one side of the ventral midline near the pelvis for examination of the gastrointestinal and urogenital tracts, (3) lateral to the cloaca also for the gastrointestinal and urogenital tracts, (4) the flank behind the last rib for the genital organs and posterior lungs, (5) the flank in front of the last rib also for the genital organs and posterior lungs, and (6) dorsally between the second- and third-to-last ribs, slightly below the vertebral column (and just anterior to the leg) for examination of the lungs, genital organs, and heart. In Siberian Cranes, hemorrhage has frequently occurred when the entry site is behind the last rib. Therefore, an entry site between the last two ribs is preferred for this species. Rigid and flexible endoscopes have also been used to visualize lesions in the trachea, esophagus, and cloaca.

Contraindications for endoscopic examination include severe disease conditions that would prevent general anesthesia, obesity, fluid in the abdomen (ascites), and the presence of a large developing egg. Possible complications include trauma to organs; laceration of a blood vessel, liver, or spleen resulting in serious hemorrhage and occasionally death; subcutaneous emphysema from air leaking from an air sac through the hole made in the body wall; and the possibility of sepsis at the surgery site. The risk of subcutaneous emphysema can be lessened by placing an absorbable suture in the body wall and another in the skin upon exiting an endoscopic site, or by using tissue glue (cyanoacrylic glue) to seal the surgery site. Trauma to organs, hemorrhage, and sepsis are all controlled best by using proper techniques. We recommend prior training to avoid mistakes and to enable proper interpretation of tissues viewed through the endoscope.

Ventriculotomy/Foreign Body Removal

Cranes frequently pick up bright or novel objects and occasionally swallow them. These objects frequently lodge in the ventriculus. Clinical signs reported in Sarus Cranes include lethargy, labored standing, hock sitting, and diarrhea (Bush and Kennedy 1978). Gastrointestinal bleeding and signs of heavy metal toxicosis (lead, zinc, etc.; see Poisoning) have been seen. One Whooping Crane in Maryland died from ingesting a nail that punctured the ventriculus. In another case, a Florida Sandhill Crane that ingested a gold earring suffered severe clinical signs. Two Whooping Cranes that swallowed wire survived. In one case, the wire penetrated the gizzard and became walled off in a necrotic mass in the abdomen. In the second case, the wire penetrated the gizzard mucosa and was found in the gizzard muscle layers. Operations to remove the wire were successful in both cases.

If clinical signs appear, take **radiographs** to confirm the presence of a metallic foreign body (Fig. 8.20). Unfortunately, not all foreign bodies are evident on radiographs. **Surgery** can be performed to remove the object (if it is likely to poison the crane or pierce the gastrointestinal tract). Surgery (a ventriculotomy or proventriculotomy) is performed under general anesthesia using isoflurane. If possible, fast the crane 12–24 hours to reduce gut contents. Place the crane in right lateral recumbency and incise the skin and muscle parallel to the last rib. The ventriculus is exteriorized, packed off, and incised through the *musculus intermedin* at the posterior end of the ventriculus (or through the less muscular proventriculus). The foreign object is removed and then the ventriculus is closed with

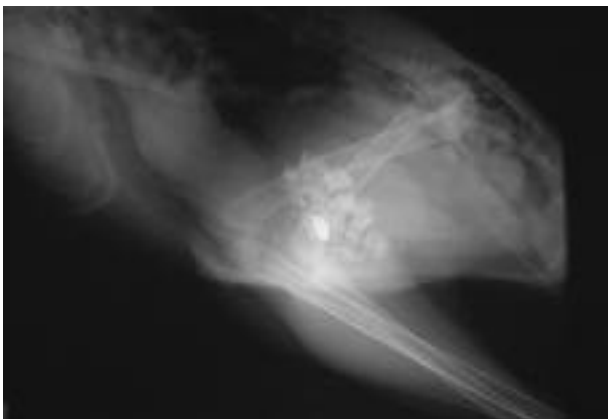


Fig. 8.20. *Ingested object (radiograph).*

Photo Glenn H. Olsen

4-0 absorbable suture material in a 3-layer closure. First, close the mucosa/submucosa with simple interrupted sutures. Second, close the ventriculus muscle wall with both horizontal and vertical mattress sutures. Third, close the adventia with interlocking continuous sutures. The abdominal muscle wall and skin are closed with 4-0 absorbable sutures. Reduce food intake for 5 days post surgery and treat the crane with antibiotics (Table 8.1). Feces will return to normal within 10 days after surgery (Bush and Kennedy 1978). Only recently has endoscopy proven useful for retrieval of stomach foreign bodies in adult cranes.

Preventive Medicine

Preventive medicine should include annual health checks. Each bird needs a physical exam, a blood count and blood chemistry profile, a screening for likely or common infections (e.g., Salmonella, EEE, IBDC, TB), and a fecal parasite analysis. For cranes entering or leaving the colony, impose a 30–60 day quarantine with disease screening. Prophylactic treatment (for parasites, etc.) and vaccination (EEE, botulism) schedules should be developed to meet the needs of the flock. The Whooping Crane Health Advisory Team has published detailed preventive medicine protocols (Langenberg and Dein 1992) that are useful for other species.

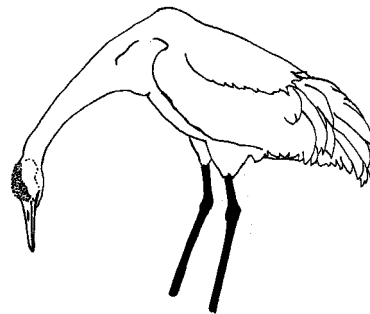
Literature Cited

- Allen, J. L., H. D. Martin, and A. M. Crowley. 1985. Metastatic cholangiocarcinoma in a Florida Sandhill Crane. *Journal of the American Veterinary Medical Association* 187:1215.
- Altman, R. B. 1984. Use of temp-plus dental product for beak repair and prosthesis. *Association of Avian Veterinarians Newsletter* 5(4):101.
- Atyeo, W. T., and R. M. Windingstad. 1979. Feather mites of the Greater Sandhill Crane. *Journal of Parasitology* 65:650–658.
- Boever, W. J. 1986. Noninfectious diseases. Pages 962–964 *in* M. E. Fowler, editor. *Zoo and wild animal medicine*. W. B. Saunders, Philadelphia, Pa.
- Brannian, R. E., D. L. Graham, and J. Creswell. 1981. Restraint associated myopathy in East African Crowned Cranes. Pages 21–23 *in* M. E. Fowler, editor. *Proceedings of the American Association of Zoo Veterinarians*, Seattle, Wash.

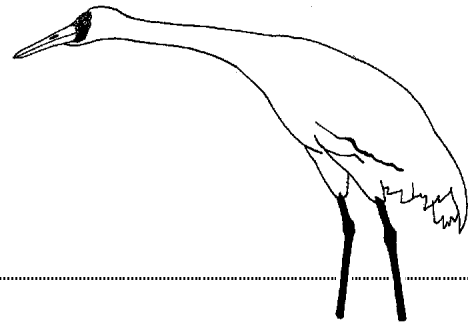
- Bush, M., and S. Kennedy. 1978. Ventriculostomy for removal of foreign bodies from Sarus Cranes. *Journal of the American Veterinary Medical Association* 173:1107-1110.
- Bush, M., D. Locke, L. A. Neal, and J. W. Carpenter. 1981a. Pharmacokinetics of cephalothin and cephalexin in selected avian species. *American Journal of Veterinary Research* 42:1014-1017.
- Bush, M., D. Locke, L. A. Neal, and J. W. Carpenter. 1981b. Gentamicin tissue concentrations in various avian species following recommended dosage therapy. *American Journal of Veterinary Research* 42:2114-2116.
- Bush, M., R. J. Montali, G. R. Novak, and A. E. James. 1976. The healing of avian fractures: a histological xeroradiographic study. *Journal of the American Animal Hospital Association* 12:768-773.
- Bush, M., L. A. Neal, and R. S. Custer. 1979. Preliminary pharmacokinetics studies of selected antibiotics in birds. Page 45 *in* Proceedings of the American Association of Zoo Veterinarians.
- Cambre, R. C., and D. Kenny. 1993. Vaccination of zoo birds against avian botulism with mink botulism vaccine. Page 383 *in* Proceedings of the American Association of Zoo Veterinarians, St. Louis, Mo.
- Campbell, T. W. 1988. Avian hematology and cytology. Iowa State University Press, Ames. 101 pp.
- Campbell, T. W. 1994. Chapter 9, Hematology. Pages 176-198 *in* B. W. Ritchie, G. J. Harrison, and L. R. Harrison, editors. *Avian medicine: principles and applications*. Wingers Publishing, Lake Worth, Fla.
- Carpenter, J. W. 1979. An outline of the treatment and control of crane parasites. Pages 101-108 *in* J. C. Lewis, editor. *Proceedings 1978 Crane Workshop*. Colorado State University Press, Fort Collins.
- Carpenter, J. W. 1986. Cranes (order Gruiformes). Pages 316-325 *in* M. E. Fowler, editor. *Zoo and wild animal medicine*. W. B. Saunders, Philadelphia, Pa.
- Carpenter, J. W. 1993. Infections and parasitic diseases in cranes. Pages 229-237 *in* M. E. Fowler, editor. *Zoo and wild animal medicine: recurrent therapy* 3. W. B. Saunders, Philadelphia, Pa.
- Carpenter, J. W., G. G. Clark, and D. M. Watts. 1989. The impact of eastern equine encephalitis virus on efforts to recover the endangered Whooping Crane. Pages 115-120 *in* J. E. Cooper, editor. *Disease and threatened birds*. ICBP Technical Publication 10.
- Carpenter, J. W., F. J. Dein, and G. Clark. 1987. An outbreak of eastern equine encephalitis virus in captive Whooping Cranes. Pages 123-127 *in* J. C. Lewis, editor. *Proceedings 1985 Crane Workshop*. Platte River Whooping Crane Habitat Maintenance Trust, Grande Island, Nebr.
- Carpenter, J. W., and S. R. Derrickson. 1981. Whooping Crane mortality at the Patuxent Wildlife Research Center, 1966-1981. Pages 175-179 *in* J. C. Lewis, editor. *Proceedings 1981 Crane Workshop*. National Audubon Society, Tavernier, Fla.
- Carpenter, J. W., L. N. Locke, and J. C. Miller. 1976. Mortality in captive Sandhill Cranes at Patuxent Wildlife Research Center, 1966-1975. Pages 268-283 *in* Proceedings of 1975 Crane Workshop. Oklahoma State University Press, Stillwater.
- Carpenter, J. W., M. N. Novilla, and J. S. Hatfield. 1992. The safety and physiologic effects of the anticoccidial drugs monensin and clazuril in Sandhill Cranes (*Grus canadensis*). *Journal of Zoo and Wildlife Medicine* 23:214-221.
- Carpenter, J. W., N. J. Thomas, and S. Reeves. 1991. Capture myopathy in an endangered Sandhill Crane (*Grus canadensis pulla*). *Journal of Zoo and Wildlife Medicine* 22:488-493.
- Chappell, S. A., and R. E. Brannian. 1984. Serum chemistry in East African Crowned Cranes. *Journal of Zoo Animal Medicine* 15:32-34.
- Clark, G. G., F. J. Dein, C. L. Crabbs, J. W. Carpenter, and D. M. Watts. 1987. Antibody response of Sandhill and Whooping Cranes caused by eastern equine encephalitis (EEE) virus vaccine. *Journal of Wildlife Diseases* 23:539-544.
- Coles, E. H. 1980. *Veterinary clinical pathology*. W. B. Saunders, Philadelphia, Pa. 562 pp.
- Connetta, J., E. Garner, E. Dolensek, H. Kobrin, and R. Strebel. 1974. Hematologic and biochemical profiles in normal Demoiselle Cranes (*Anthropoides virgo*). *Laboratory Animal Science* 24:105-110.
- Cook, R. A., R. A. Moretti, and J. A. Duffelmeyer. 1989. Hematology of the White Naped Crane (*Grus vipio*) (abstract). Page 417-418 *in* Proceedings First International Conference on Zoological and Avian Medicine, September 6-11, 1987, Oahu, Hawaii.
- Courtney, C. H., D. J. Forrester, J. V. Ernst, and S. A. Nesbitt. 1975. Coccidia of Sandhill Cranes, *Grus canadensis*. *Journal of Parasitology* 61:695-699.
- Curro, T. G., J. Langenberg, and J. Paul-Murphy. 1992. A review of lameness in long-legged birds. Pages 265-269 *in* Proceedings of the Association of Avian Veterinarians, New Orleans, La.
- Custer, R. S., M. Bush, and J. W. Carpenter. 1979. Pharmacokinetics of gentamicin in blood plasma of quail, pheasants, and cranes. *American Journal of Veterinary Research* 40:892-895.
- Decker, R. A., and J. C. Hruska. 1978. Renal adenocarcinoma in a Sarus Crane (*Grus antigone*). *Journal of Zoo Animal Medicine* 9(1):15-16.
- Dein, F. J. 1984. *Laboratory manual of avian hematology*. Association of Avian Veterinarians, East Northport, N.Y. 38 pp.
- Dein, F. J., J. W. Carpenter, G. G. Clark, R. J. Montali, C. L. Crabbs, T. F. Tsai, and D. E. Docherty. 1986. Mortality of captive Whooping Cranes caused by eastern equine encephalitis virus. *Journal of the American Medical Association* 189:1006-1010.
- Docherty, D. E., and D. J. Henning. 1980. The isolation of a herpesvirus from captive cranes with an inclusion body disease. *Avian Diseases* 24:278-283.
- Done, L. B., D. M. Ialeggio, and M. Cranfield. 1993. Therapeutic use of methocarbamol in a Demoiselle Crane (*Anthropoides virgo*) with severe ataxia and lateroflexion of the neck. Page 137 *in* Proceedings of the American Association of Zoo Veterinarians, St. Louis, Mo.
- Douglas, S. W., and H. D. Williamson. 1970. *Veterinary radiological interpretation*. Lea and Febiger, Philadelphia, Pa. 303 pp.
- Fagan, D. 1982. Beak repair in a South American Black Eagle. *American Association of Veterinarians Newsletter* 3(2):41.
- Fannin, T. E. 1992. Contaminant residues in Sandhill Cranes killed upon striking powerlines in Central Nebraska. *Proceedings North American Crane Workshop* 6:166-170.

- Forrester, D. J., J. W. Carpenter, and D. R. Blankinship. 1978. Coccidia of Whooping Cranes. *Journal of Wildlife Diseases* 14:24-27.
- Forrester, D. J., F. H. White, and C. F. Simpson. 1976. Parasites and diseases of Sandhill Cranes in Florida. Pages 284-290 *in* J. C. Lewis, editor. Proceedings of the International Crane Workshop. Oklahoma State University, Stillwater.
- Franson, J. C., and S. G. Hereford. 1994. Lead poisoning in a Mississippi Sandhill Crane. *Wilson Bulletin* 106:766-768.
- Frye, F. L. 1984. Prosthesis enhance quality of life. *Veterinary Medicine* 7:931-935.
- Gee, G. F., J. W. Carpenter, and G. L. Hensler. 1981. Species differences in hematological values of captive cranes, geese, raptors, and quail. *Journal of Wildlife Management* 45:463-483.
- Graham, D. L. 1987. Characterization of a reo-like virus and its isolation from and pathogenicity for parrots. *Avian Diseases* 31:411-419.
- Greenwell, M., J. J. Robertson, and G. M. Constantinescu. 1989. Avian beak prosthesis. *The Compendium on Continuing Education for the Practicing Veterinarian* 11:1365-1371.
- Greiner, E. D., and B. W. Ritchie. 1994. Parasites. Pages 1007-1029 *in* B. W. Ritchie, G. J. Harrison, and L. R. Harrison, editors. Avian medicine, principles and applications. Wingers Publishing, Lake Worth, Fla.
- Harthoorn, A. M., and E. Young. 1974. A relationship between acid-base balance and capture myopathy in zebra (*Equus burchelli*) and an apparent therapy. *Veterinary Record* 94:337-342.
- Hartman, L. M. 1983. Summary of mortality of 14 species of cranes at the International Crane Foundation, 1972-1982. Pages 555-570 *in* G. W. Archibald and R. F. Pasquier, editors. Proceedings 1983 International Crane Workshop, Bharatpur, India. International Crane Foundation, Baraboo, Wis.
- Hawkey, C., J. H. Samour, D. G. Ashton, M. G. Hart, R. N. Cindery, J. M. Finch, and D. M. Jones. 1983. Normal and clinical haematology of captive cranes (Gruiformes). *Avian Pathology* 12:73-84.
- Hawkey, C. M., and T. B. Dennet. 1989. Comparative veterinary hematology. Iowa State University Press, Ames. 192 pp.
- Henny, C. J., L. J. Blus, and C. S. Hulse. 1985. Trends and effects of organochlorine residues on Oregon and Nevada wading birds, 1979-83. *Colonial Waterbirds* 8(2):117-128.
- Hill, E. F., and W. J. Fleming. 1982. Anticholinesterase poisoning of birds: field monitoring and diagnosis of acute poisoning. *Environmental Toxicology and Chemistry* 1:27-38.
- Hochleithner, M. 1994. Biochemistries. Pages 223-245 *in* B. W. Ritchie, G. J. Harrison, and L. R. Harrison, editors. Avian medicine: principles and applications. Wingers Publishing, Lake Worth, Fla.
- Howard, P. 1989. Management of beak fractures in cranes. Page 24 *in* Proceedings of the 1989 Exotic Animal Dentistry Conference, Philadelphia, Pa.
- Howard, P. E. 1990. The use of bone plates in the repair of avian fractures. *Journal of the American Animal Hospital Association* 26:613-622.
- Howard, P. E., F. J. Dein, J. A. Langenberg, K. J. Frischmeyer, and D. B. Brunson. 1991. Surgical removal of a tracheal foreign body from a Whooping Crane (*Grus americana*). *Journal of Zoo and Wildlife Medicine* 22:359-363.
- Kaleta, E. F., and H. J. Marschall. 1981. Newcastle disease in a zoo affecting Demoiselle Cranes (*Anthropoides virgo*), Greater Flamingos (*Phoenicapterus ruber*) and a Pied Imperial Pigeon (*Ducula bicolor*). *Avian Pathology* 10:395-401.
- Karpinski, L. G., and S. L. Clubb. 1986. Clinical aspects of ophthalmology in caged birds. Pages 616-621 *in* R. W. Kirk, editor. Current veterinary therapy IX, small animal practice. W. B. Saunders, Philadelphia, Pa.
- Kennedy, S., J. P. Crisler, E. Smith, and M. Bush. 1977. Lead poisoning in Sandhill Cranes. *Journal of the American Veterinary Medical Association* 171:955-958.
- Klein, P., K. Charmatz, and J. Langenberg. 1994. The effect of flunixin meglumine (Banamine R) on the renal function in Northern Bobwhite (*Colinus virginianus*): an avian model. Page 128 *in* Proceedings of the American Association of Zoo Veterinarians, Pittsburgh, Pa.
- Lamont, T., and W. Reichel. 1970. Organochlorine pesticide residues in Whooping Cranes and Everglade Kites. *Auk* 87:158-159.
- Langenberg, J., and F. J. Dein. 1992. Whooping Crane health management workshop report. International Crane Foundation, Baraboo, Wis. 47 pp.
- Langenberg, J. A., and F. J. Dein. 1994. Diseases and disease management in North American cranes. *Wildlife Rehabilitation* 12:43-53.
- Lewis, J. C. 1974. Ecology of the Sandhill Crane in the southeastern central flyway. Ph.D. dissertation, Oklahoma State University, Stillwater. 213 pp.
- Lewis, J. C., R. C. Drewien, E. Kuyt, and C. Sanchez, Jr. 1992. Contaminants in habitat, tissue, and eggs of Whooping Cranes. Proceedings North American Crane Workshop 6:159-165.
- Locke, D., M. Bush, and J. W. Carpenter. 1982. Pharmacokinetics and tissue concentrations of tylosin in selected avian species. *American Journal of Veterinary Research* 43:1807-1810.
- Ludders, J. W., J. Rode, and G. S. Mitchell. 1989. Isoflurane anesthesia in Sandhill Cranes (*Grus canadensis*): minimal anesthetic concentration and cardiopulmonary dose-response during spontaneous and controlled bleeding. *Anesthesia and Analgesia* 68:511-516.
- MacWhirter, P. 1987. Reproductive and genetic disorders. Pages 143-159 *in* P. MacWhirter, editor. Everybird, a guide to bird health. Inkata Press, Melbourne, Australia.
- Magrane, W. G. 1965. Canine ophthalmology. Lea & Febiger, Philadelphia, Pa. 240 pp.
- McDonald, S. E. 1982. Surgical sexing of birds by laparoscopy. *California Veterinarian* 5:16-22.
- McMillan, M. C. 1988. Avian radiographic diagnosis. Pages 301-308 *in* 1988 Proceedings of the Association of Avian Veterinarians, Houston, Tex.
- McMillan, M. C. 1994. Imaging techniques. Pages 246-326 *in* B. W. Ritchie, G. J. Harrison, and L. R. Harrison, editors. Avian medicine: principles and applications. Wingers Publishing, Lake Worth, Fla.

- Montali, R. J. 1977. An overview of tumors in zoo animals. *Journal of the American Veterinary Medical Association* 171:531.
- Mullins, W. H., E. G. Bizeau, and W. W. Benson. 1978. Pesticide and heavy metal residues in Greater Sandhill Cranes. Pages 189-194 in J. C. Lewis, editor. *Proceedings 1978 Crane Workshop*. Colorado State University Press, Fort Collins.
- Murphy, C. J. 1987. Raptor ophthalmology. *Compendium of Small Animal Medicine* 9:241-258.
- Olsen, G. H. 1994. Orthopedics in cranes: pediatrics and adults. *Seminars in Avian and Exotic Pet Medicine* 3(2):73-80.
- Olsen, G. H. and J. W. Carpenter. 1996. Chapter 9. Cranes. In R. Altman, K. Quesenberry, and S. Clubb, editors. *Avian Medicine and Surgery*. W. B. Saunders Co., Philadelphia, Pa. In press.
- Olsen, G. H., J. W. Carpenter, G. F. Gee, N. J. Thomas, and F. J. Dein. 1995. Mycotoxin caused disease in captive Whooping Cranes (*Grus americana*) and Sandhill Cranes (*Grus canadensis*). *Journal of Zoo and Wildlife Medicine*. In press.
- Olsen, G. H., and S. D. Gaunt. 1985. Effect of hemoprotozoal infections on rehabilitation of wild raptors. *Journal of the American Veterinary Medical Association* 187:1204-1205.
- Pagac, B. B., M. J. Turell, and G. H. Olsen. 1992. Eastern equine encephalomyelitis virus and *Culiseta melanura* activity at the Patuxent Wildlife Research Center, 1985-90. *Journal of the American Mosquito Control Association* 8(3):328-330.
- Puerta, M. L., J. C. Alonso, V. Huecas, J. A. Alonso, M. Abelenda, and R. Munoz-Pulido. 1990. Hematology and blood chemistry of wintering Common Cranes. *Condor* 92:210-214.
- Quesenberry, K. E., G. Mauldin, and E. Hillyer. 1989. Nutritional support of the avian patient. Pages 11-19 in *Proceedings of the 1989 Annual Meeting of the Association of Avian Veterinarians*, Seattle, Wash.
- Redig, P. T. 1984. Fluid therapy and acid base balance in the critically ill avian patient. Pages 59-73 in *Proceedings Annual Meeting of the Association of Avian Veterinarians*, Toronto, Canada.
- Remple, J. D. 1993. Raptor bumblefoot: a new treatment technique. Pages 154-160 in P. T. Redig, J. E. Cooper, J. D. Remple, and D. B. Hunter, editors. *Raptor biomedicine*. University of Minnesota Press, Minneapolis.
- Riddle, K. E. 1980. Surgical treatment of bumblefoot in raptors. Pages 67-73 in J. E. Cooper and A. G. Greenwood, editors. *Recent advances in the study of raptor diseases*. Chiron Publications, Keighley, West Yorkshire, England.
- Ritchie, B. W., C. M. Otto, K. S. Latimer, and D. T. Crowe. 1991. A technique of intraosseous cannulation for intravenous therapy in birds. *The Compendium on Continuing Education for the Practicing Veterinarian* 12:55-58.
- Schalm, O. W., N. C. Jain, and E. J. Carroll. 1975. *Veterinary hematology*. Lea and Febiger, Philadelphia, Pa. 807 pp.
- Serafin, J. A. 1980. Influence of dietary energy and sulfur amino acid levels upon growth and development of young Sandhill Cranes. Page 30 in *Proceedings of the American Association of Zoo Veterinarians*, Washington, D.C.
- Serafin, J. A. 1982. The influence of diet composition upon growth and development of Sandhill Cranes. *Condor* 84:427-434.
- Shane, S. M., G. H. Olsen, G. D. Smith, and W. S. Bivin. 1986. The use of a particle sampler to diagnose lower respiratory tract infection in companion birds. *Avian/Exotic Practice* 3(2&3):5-9.
- Simpson, C. F., O. J. Forrester, and S. A. Nesbitt. 1975. Avian pox in Florida Sandhill Cranes. *Journal of Wildlife Diseases* 11:112-115.
- Snyder, S. B., and M. J. Richard. 1994. Treatment of avian tuberculosis in a Whooping Crane (*Grus americana*). Page 167-169 in *Proceedings of the American Association of Zoo Veterinarians*, Pittsburgh, Pa.
- Snyder, S. B., M. J. Richard, J. P. Thilsted, R. C. Drewien, and J. C. Lewis. 1992. Lead poisoning in a Whooping Crane. Pages 207-211 in D. A. Wood, editor. *Proceedings 1988 North American Crane Workshop*. Florida Nongame Wildlife Program Technical Report 12, Gainesville.
- Taylor, M. 1994. Endoscopic examination and biopsy techniques. Pages 327-354 in B. W. Ritchie, G. J. Harrison, and L. R. Harrison, editors. *Avian medicine: principles and applications*. Wingers Publishing, Lake Worth, Fla.
- Wei, Y. C., et al. (Copy unavailable). 1986. Three cases of granulocytic leukemia in the Red Crowned Crane. *Chinese Journal of Zoology* 21(5):32-33.
- White, D. H., L. E. Hayes, and P. B. Bush. 1989. Case histories of wild birds killed intentionally with famphur in Georgia and West Virginia. *Journal of Wildlife Diseases* 25:184-187.
- Windingstad, R. M., R. J. Cole, P. E. Nelson, T. J. Roffe, R. R. George, and J. W. Dorner. 1989. *Fusarium* mycotoxins from peanuts suspected as a cause of Sandhill Crane mortality. *Journal of Wildlife Diseases* 25(1):38-46.
- Windingstad, R. M., S. M. Kerr, and L. N. Locke. 1984. Lead poisoning of Sandhill Cranes. *Prairie Naturalist* 16:21-24.
- Windingstad, R. M., S. S. Hurley, and L. Sileo. 1983. Capture myopathy in a free-flying greater Sandhill Crane (*Grus canadensis stabida*) from Wisconsin. *Journal of Wildlife Diseases* 19(3):289-290.
- Windingstad, R. M., D. O. Trainer, and R. Duncan. 1977. *Salmonella enteritidis* and *Arizona hinshawii* isolated from wild Sandhill Cranes. *Avian Diseases* 21:704-707.
- Wolf, L. 1985. Prosthetic bill technique in a Canada Goose. Pages 177-179 in 1985 *Proceedings of the Association of Avian Veterinarians*, Boulder, Colo.
- Zinkal, J. G., J. Rathert, and R. R. Hudson. 1978. Diazinon poisoning in wild Canada geese. *Journal of Wildlife Management* 42:406-408.



Genetic Management



Claire M. Mirande, George F. Gee,
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Due to the trends toward extinction of many crane species in the wild, the continued development of cooperative management programs for captive cranes is a critical component of recovery strategies. Significant progress has been made in the last ten years toward the improvement of techniques for preserving genetic diversity. Successful regional programs are under development and cooperation within regions is increasing. It is now important to develop mechanisms for coordination between these regions toward world conservation strategies. New research is needed to evaluate and refine management programs.

Efforts to preserve endangered species should promote self-sustaining wild populations. The establishment of captive “species banks” can be critical to ensure the survival of some species, and to promote the preservation of genetic diversity so populations are able to respond to change (Mettler and Gregg 1969; Wilcox et al. 1986). Lack of diversity often reduces resistance to disease, decreases fertility, increases embryo mortality, and reduces growth rates (Frankel and Soule 1981).

This chapter discusses the management of genetic diversity in captive populations of cranes. We present the basic principles and tools of genetic and demographic management of small captive populations, and review cooperative management programs and useful contacts. Finally, we summarize genetic research needs and projects underway.

Genetic and Demographic Management

Every individual in a population represents a unique combination of alleles (alternative forms of a gene). The population itself, however, can be described by measuring the **frequency of each allele** at each locus. Some alleles will be common, shared by most of the

population’s members; other alleles will be rare, found in only a few animals. The object of captive management is to preserve, so far as is possible, the genetic description of the wild population—to preserve the highest diversity possible. Achieving this goal requires controlled propagation.

Any single non-inbred individual represents 50% of the total genetic diversity in a population (Denniston 1978). However, the **alleles** in one animal do not represent the distribution of alleles in the population, unless that population contains essentially no variability. A captive population founded by only a few individuals may lack rare alleles or over represent them. The larger the number of individuals contributing to a captive population, the more accurately total genetic diversity of the species will be represented. The genetic diversity represented, however, does not increase linearly with the number of contributing individuals. In theory, the larger the number of wild individuals used to start the captive population, the better. In practice, captive managers are constrained by the lack of individuals of rare species, a lack of space, and frequently by the characteristics of existing captive populations. These populations may contain animals of unknown origins or inbred individuals and have suboptimal distributions of age, sex, and parentage.

A population’s genetic diversity is only partly dependent upon its actual size, N . The effective genetic size of a population is estimated by its **effective population number**, N_e (Crow and Kimura 1970). N_e measures the way in which the population reproduces, transmitting its genes to the next generation (Foose and Ballou 1988). Specifically, N_e is the number of individuals that would be required in a hypothetical, random breeding population of constant size, equal sex ratio, and with non-overlapping generations to retain the same amount of genetic diversity as was retained in the original population.

N_e increases when: (1) the number of breeders increases, (2) the number of offspring per breeder increases, (3) the number of offspring per breeder becomes more equal (the variance in the number of

offspring per breeder decreases [Frankel and Soule 1981]), and (4) the sex ratio is even (at least in most species). Populations with larger N_e values lose genetic diversity and rare alleles more slowly than populations with smaller N_e values (Denniston 1978). When N_e reaches a certain size, somewhere around 500 animals, the population may gain genetic variation when the rate of mutation exceeds genetic drift (Frankel and Soule 1981).

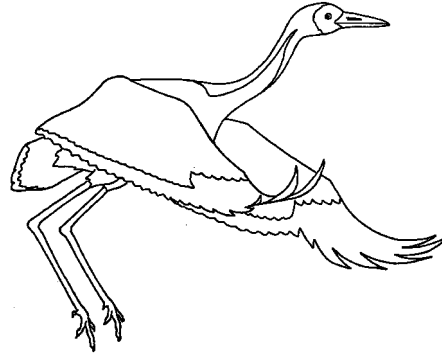
The first priority of genetic management is to breed as many of the founders in a population as possible. N_e is linearly related to the number of founders in the population. A **founder** is usually defined as an animal that is taken from the wild and has no relatives in captivity except its own descendants. Potential founders that die without leaving offspring contribute nothing to the gene pool unless their genetic material has been cryopreserved in a way that allows them to produce offspring later.

The next priority is to **increase the number of offspring per founder**. Quickly breeding a rare species is important to increase its chances of long-term survival (Soule 1983) and is genetically beneficial as well (Flesness 1977). However, skewing the genetic representation in favor of a few foundersto achieve this growth can be harmful. Also, fluctuations in growth rates should be avoided because they can skew the age distribution (see below) and thereby decrease the population's stability. Program managers must also determine size of a **minimum viable population (MVP)**. MVP can be estimated using CAPACITY software (see Studbook section below). The **carrying capacity** for this population in captivity needs to be equal or larger than the MVP to achieve program goals.

Once a captive population is large enough to withstand extinction, it should be managed to **equalize the genetic representation** of its founders. Increasing the number of offspring of poorly represented pairs can dramatically increase the genetic diversity of a population without increasing the number of individuals (Denniston 1978; Swengel 1987). Sometimes this also requires curtailing breeding or culling offspring for highly represented pairs.

Culling involves dispersing cranes or euthanasia. If a captive population is at carrying capacity, N_e can be increased by culling offspring of over-represented lineages. Culling can thereby correct some of the genetic harm due to unequal breeding or differential survival. Generally, only second or subsequent generation animals are culled.

The genetic diversity of a population is greatest when each founder has the same number of offspring. When each pair has the same number of chicks, N_e is twice that of a random breeding population (Crow and Kimura 1970; Frankel and Soule 1981). Captive crane pairs generally have very unequal numbers of chicks (Swengel 1987; Sheppard 1988) ($N_e < N$). Long reproductive life spans also enable a few pairs to produce most of the next generation.



Demographic management involves the examination and manipulation of population characteristics toward achieving **stable age structure** and a **stable population size** near carrying capacity. **Age specific fertility** and **age specific mortality** are the most important data. Demographic analysis reveals the number of offspring required from each breeding-age crane to maintain a stable population. Managers must also know the age at first reproduction, longevity, reproductive life span, and sex ratio before enacting long-term demographic management. From this information, **mathematical models** aided by the computer (see Studbook section below) can predict population growth and the rate of loss of genetic material. Such models establish numbers of offspring needed per pair (Odum 1994). Lifetime reproductive goals can then be translated into annual breeding objectives for each bird.

Optimally, the **sex ratio** for captive crane populations should be even. Some captive populations have uneven sex ratios. For example, in the past, Red-crowned Cranes in North America have been skewed toward females and Siberian Cranes toward males. This skewing is probably an artifact of small population size.

During each generation, some genetic diversity is lost because each parent contributes only half of its genes to an offspring. On average, it requires six offspring to represent 98.4% of each parent's genetic

information. To reduce the rate of loss of genetic diversity, it is important to **maximize generation time** (the average age at which an individual produces offspring). One possible strategy for a population near carrying capacity is to allow each pair to breed once or twice when they reach sexual maturity to ensure retention of some genetic information of the breeders. Then allow the pair to produce only one offspring every five years until the desired number of offspring are produced. An alternate strategy is to delay breeding, cull older offspring, and breed the youngest offspring.

Inbreeding, the breeding of genetically related individuals, decreases heterozygosity (proportion of loci measured which have two different alleles), increases homozygosity (proportion of loci measured which have the same alleles), and is generally harmful (Flesness 1977; Frankel and Soule 1981; Ralls et al. 1988). Inbreeding reduces fertility and hatchability in Red-crowned Cranes (Swengel 1985), leads to greater expression of the population's **genetic load** (i.e., increases the rate of expression of harmful recessive alleles), and decreases population fitness especially for reintroductions (Frankham et al. 1986). Some alleles influence survival and fitness more than others. It may be possible to design breeding programs to prevent the loss of highly advantageous alleles. Flesness (1977) describes how to avoid inbreeding. **Inbreeding coefficients** for all potential pairings can be obtained using SPARKS and GENES software (see Studbook section below).

Populations which go through a **genetic bottleneck** (i.e., major reduction in size), and are therefore derived from a few individuals, are more likely to be at greater risk for expression of genetic load through inbreeding. Because we normally cannot assess the number of deleterious genes in the founding individuals, it is best to avoid unnecessary inbreeding. Bottlenecks also result in the loss of allelic and gene diversity (related to, but not the same as heterozygosity), which estimate the presence of rare alleles. This loss decreases the ability of the population to adapt to changes in its environment (Mettler and Gregg 1969). Breeding strategies should be designed to preserve three types of genetic diversity: heterozygosity, allelic diversity, and gene diversity (Willis and Wiese 1995).

In captive breeding programs, it is important to avoid **artificial selection** (Miller and Hedrick 1991). We should not select for birds adapted to captivity (e.g., tame birds are often more productive). Natural selection has selected favorable traits for millions of years, and the best we can do is minimize evolutionary

change while the birds are in captivity. Geneticists debate the use of artificial selection to reduce genetic load. Frankham et al. (1986) recommend preventing cranes with harmful traits from breeding. Natural selection after release will remove harmful traits, so artificial selection is generally avoided.

To aid readers, a summary of guidelines to maximize genetic diversity is presented in Table 9.1. Table 9.2 summarizes procedures for selecting mates and targeting the number of offspring per pair.

Studbooks

The studbook, the basic tool for genetic management, contains a genealogy of all animals living or dead (see Table 9.3 for studbook managers, and Table 9.4 for a sample studbook). Although several species have international studbooks, regional studbooks also are kept to facilitate local management decisions.

Each studbook contains identification numbers, date of hatch, sex, parentage, date and cause of death, and dates and locations where the cranes have been

TABLE 9.1.

Summary of genetic and demographic management guidelines to maximize genetic diversity.¹

1. Start the population with an adequate number of founders (at least 20 founders which effectively reproduce).
2. Expand the population to captive "carrying capacity" as quickly as possible. Carrying capacity should be larger than the minimum viable population size.
3. Equalize the sex ratio (number of breeding males: number of breeding females).
4. Equalize family size (the breeding animals should have equal numbers of offspring contributing to the next generation).
5. Stabilize the size and growth rate of the captive population once it reaches "carrying capacity" (generally 100-500 cranes). Avoid fluctuations in population size.
6. At this stage, manage for longer generation times.
7. Minimize inbreeding at all stages.
8. Manage for stable age structure at all stages.

¹ Adapted from DeBoer (1989).

TABLE 9.2.

Recommended procedures for selecting mates and targeting number of offspring per pair.²

1. Assign genetic values to birds. (GENES software provides an ordered list of mean kinship by sex and a measure of rare alleles in the "proportion of genome unique" report.)
2. First, breed birds with highest genetic value (lowest mean kinship). These birds should produce the largest number offspring.
3. Second, breed birds with lower genetic value, but whose alleles may be lost soon. (Knowledge of managers and kinship value in the SPARKS masterplan report are sources of this information.)
4. Pair individuals according to the following criteria:
 - a. Mate individuals with similar genetic value (e.g., mean kinship) to avoid combining rare and common alleles.
 - b. Mate individuals whose offspring will have low inbreeding coefficients.
 - c. Maximize pairing success based on age, behavior, and physical condition.
 - d. Adjust for logistical considerations such as transfers, quarantine, and cost.
 - e. Adjust, if needed, based on wishes of individuals or institutions.

² Adapted from Wiese and Willis (1993).

held. It also includes mates, inbreeding coefficients, living offspring and siblings, and summary tables of holdings by institution, births, deaths, and transfers. Some detail may also be given on status in the wild and captive management efforts.

A studbook-like report for species which do not have an official studbook can now be provided by the International Species Information System (ISIS) (see Chapter 10). ISIS has developed a valuable new software program entitled Single Population Analysis and Record Keeping System (SPARKS). This software is designed for the production of studbooks and to conduct basic genetic and demographic analysis on population data. Supporting software (GENES, DEMOG, and CAPACITY) provides for most of the analysis described in this section. Individuals and institutions with captive cranes are encouraged to join ISIS. Further information can be obtained by contacting ISIS (address in Appendix) or ICF.

Cooperative Captive Management Programs

Considerable effort has been focused, both regionally and internationally, on **coordinating captive management efforts** to preserve genetic diversity. For these programs to succeed, individual animals must be paired and bred (or not bred) according to genetic and demographic management strategies. To a degree, participating institutions have less autonomy in determining the fate of individual cranes, but are committed to a larger goal (long term preservation of the gene pool).

In 1992, global priorities for captive propagation of all cranes were established as part of a **Conservation Assessment and Management Plan (CAMP)** which summarized management in the wild, recovery/management plans, research, and the size and type of captive programs needed to support field efforts.

At a **Global Captive Action Plan (GCAP)** workshop for cranes in 1993, the status of captive populations was examined including estimates of global and regional population sizes, degree of difficulty in breeding, existence of international or regional studbooks or management programs, and release programs. Topics examined included management of founders, research needs, studbook and management program needs, and methods for coordinating global and regional programs. Target populations were established for the world.

Regional crane **Taxon Advisory Groups (TAGs)** are being formed to determine regional roles in captive management, coordinate allocation of limited space and resources between taxa, and coordinate programs with other regions. Crane TAGs have been established for global populations in North America, Europe, Africa, and Australia (see Table 9.3). Chinese and Japanese TAGs are being developed.

Global cooperation for individual species is organized under **Global Animal Survival Plans (GASPs)**. Some species, such as the Red-crowned Crane, can be effectively managed as regional metapopulations with periodic exchange of bloodlines. Other species, such as the Siberian Crane, have a lower number of founders and must be managed globally to insure genetic health. GASP workshops have been held for the Red-crowned and Siberian Cranes and are recommended for the Black-necked, Hooded, White-naped, and Wattled Cranes.

TABLE 9.3.

Summary of studbooks and management programs for cranes.

Regional Taxon Advisory Group (TAG)**Coordinators for Cranes**

Conservation Breeding Specialist Group (CBSG)

Captive Crane Working Group:

Claire Mirande, International Crane Foundation

North America:

Claire Mirande, International Crane Foundation

Europe:

Gunter Schlessner, Wilhelma Zoological Gardens

U.K. and Ireland:

Nick Lindsay, Whipsnade Zoo, and Dave Coles,
Child Beale Trust

Africa:

Alan Abrey, Umgemi Bird Park

China:

To be determined

Japan:

Kazuaki Nippashi, Saitama Children's Zoo

White-naped Crane

International Studbook Keeper and SSP

(North America) Coordinator:

Christine Sheppard, Wildlife Conservation Society

EEP (Europe) Coordinator:

Peter Muhling, Nuremberg Zoo

JMSP (U.K.) Studbook Keeper:

Nick Lindsay, Whipsnade Zoo

SSCJ (Japan) Coordinator, Studbook Keeper and

Regional Coordinator:

Kazuaki Nippashi, Saitama Children's Zoo

Wattled Crane

International Studbook Keeper and SSP Coordinator:

Fred Beall, Franklin Zoological Park

Global Animal Survival Plan (GASP) Coordinators:

Fred Beall, Franklin Zoological Park

Linda Rodwell, The Highlands Crane Group

JMSP Studbook Keeper and JMSP Coordinator:

Nick Lindsay, Whipsnade Zoo

SSCJ Studbook Keeper and Coordinator:

Masanori Kobayashi, Chiba Zoo

Hooded Crane

International Studbook Keeper and SSP Coordinator:

Bruce Bohmke, Phoenix Zoo

JMSP Studbook Keeper and JMSP Coordinator:

Nick Lindsay, Whipsnade Zoo

SSCJ Studbook Keeper & Regional Coordinator:

Takeshi Sakoh, Hira Kawa Zoo

Siberian CraneInternational Studbook Keeper and International Global
Animal Survival Plan (GASP) Coordinator:

Vladimir Panchenko, Oka State Nature Reserve, Lakash

Chinese Studbook Keeper:

Zhao Qingguo, Chinese Association of
Zoological Gardens**Red-crowned Crane**

Global Animal Survival Plan:

no coordinator designated

International Studbook Keeper and SSCJ Coordinator:

Teruyuki Komiya, Tokyo Ueno Zoo

North American Studbook Keeper:

Scott Swengel, International Crane Foundation

SSP Coordinator:

Claire Mirande, International Crane Foundation

Chinese Studbook Keeper and Regional Coordinator:

Liu Dajun, Shenyang Zoo

EEP Coordinator and Regional Studbook Keeper:

Rob Belterman, Rotterdam Zoo

JMSP Studbook Keeper and JMSP Coordinator:

Nick Lindsay, Whipsnade Zoo

Blue Crane

International Studbook Keeper:

Ferdi Schoeman, National Zoological Gardens of
South Africa

North American Studbook Keeper:

Susan Scott, North Carolina Zoological Park

JMSP Studbook Keeper and JMSP Coordinator:

Whipsnade Zoo

West African Crowned Crane

North American Studbook Keeper:

Susan Haeffner, Denver Zoo

JMSP Studbook Keeper:

Roger Wilkinson, Chester Zoo

Black-necked Crane

Chinese Studbook Keeper:

Zhao Qingguo, Chinese Association of
Zoological Gardens

TABLE 9.4.**Sample studbook produced by SPARKS software.**SIBERIAN CRANE STUDBOOK (*Grus leucogeranus*)

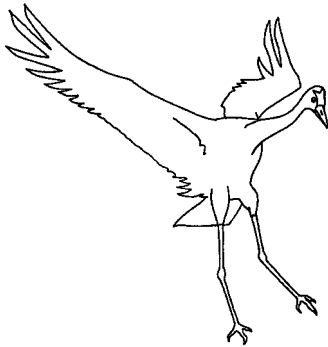
Stud No.	Sex	Hatch Date	Sire	Dam	Location	Date	Local ID	Event	Birth/Origin	Death Date	Rearing	Name
58	M	19 May 84	UNK	4	Baraboo	19 May 84	6-016	Hatch	Captive Born		Hand	A. Wright
59	M	26 May 84	18	4	Baraboo	26 May 84	6-017	Hatch	Captive Born	25 Sep 87	Hand	Avery
						25 Sep 87		Death				
60	F	18 Jun 84	5	12	Baraboo	20 Jun 84	6-018	Hatch	Captive Born		Hand	Dr. Saab
61	M	20 Jun 84	18	4	Baraboo	20 Jun 84	6-019	Hatch	Captive Born		Hand	Ferguson
					Walsrode	16 Mar 88	UNK	Transfer				
62	M	10 Jun 85	18	4	Baraboo	10 Jun 85	6-020	Hatch	Captive Born		Hand	Gole
					Tokytotama	12 Mar 86	UNK	Transfer				
63	M	20 Jun 85	5	12	Baraboo	20 Jun 85	6-021	Hatch	Captive Born		Hand	Samar
					Tokytotama	12 Mar 86	UNK	Transfer				
64	F	17 May 86	18	4	Baraboo	17 May 86	6-022	Hatch	Captive Born		Hand	Ranjit
65	M	27 May 86	18	4	Baraboo	27 May 86	6-023	Hatch	Captive Born		Hand	Leif
						27 Nov 90		Death		27 Nov 90		
66	F	17 Jun 86	43	12	Baraboo	17 Jun 86	6-024	Hatch	Captive Born		Hand	Spitzin
						21 May 88		Death		21 May 88		
67	M	2 Jul 86	Wild	Wild	Oka	2 Jul 86	OKA-30	Hatch	Captive Born		Hand	Kieng
68	F	3 Jul 86	Wild	Wild	Oka	3 Jul 86	OKA-31	Hatch	Captive Born		Hand	Banyl
69	F	3 Jul 86	Wild	Wild	Oka	3 Jul 86	OKA-32	Hatch	Captive Born		Hand	Igel
						7 Aug 86		Death		7 Aug 86		
70	M	3 Jul 86	Wild	Wild	Oka	3 Jul 86	OKA-33	Hatch	Captive Born		Hand	Kulumer
					Moscow	13 Oct 86	UNK	Transfer				
						24 Nov 86		Death		24 Nov 86		
71	M	4 Jul 86	Wild	Wild	Oka	4 Jul 86	OKA-35	Hatch	Captive Born		Hand	Uol
						30 Jul 86		Death		30 Jul 86		
72	M	4 Jul 86	Wild	Wild	Oka	4 Jul 86	OKA-34	Hatch	Captive Born		Hand	Kjuel
						3 Nov 86		Death		3 Nov 86		

Compiled by: V. Panchenko, Oka State Reserve, through International Crane Foundation.
 Data current through: 7 Sep 1993 World

SPARKS v1.2
 8 Oct 1993

Species management programs have been formed for regional coordination of captive management including recommendations for transfers, pairings, and pair-by-pair productivity. Table 9.3 summarizes management programs for cranes.

The **Population and Habitat Viability Analysis** (PHVA) process uses computer simulation modelling (VORTEX software) to predict the probability of survival or extinction of wild and captive populations under current and potential conditions. PHVAs are a valuable tool in the development of recovery plans. Workshops have been held for Whooping, Red-crowned, Siberian, Mississippi Sandhill, and Wattled Cranes. For information contact: Conservation Breeding Specialist Group (CBSG) (address in Appendix) or Claire Mirande at ICF.



Genetic Research

Significance

Studies of **genetic diversity and relatedness** are particularly relevant to management of species such as the Whooping Crane where the captive flock was established from a very small wild population (i.e., following a genetic bottleneck). Although eggs from this population were collected from known nest sites, the relatedness of wild pairs and the continuity of nest site use are unknown (Gee et al. 1992). Diversity can also be used to evaluate **divergence** between populations important for setting management goals. For example, we need to know how much the Mississippi Sandhill Crane differs from the other Sandhill subspecies to evaluate our investment in its preservation.

Accurate pedigree information is essential for genetic management. Unfortunately, **parentage** is unknown in some captive cranes (either because

semen from more than one donor has been used to inseminate a female or because more than one female occupies the pen where an egg is found). Sometimes poor record keeping clouds parentage information.

Ongoing Genetic Research

Research on diversity and relatedness in cranes encompasses many techniques including protein electrophoresis, restriction fragment length polymorphisms (RFLP), competitive binding immunoassays, and blood typing. Details on these techniques are presented by Gee et al. (1992).

Protein electrophoresis reveals a small part of the entire genome by examining blood or tissue homogenates for banding patterns associated with specific enzymes. These bands represent phenotypes at the enzyme locus. This gives information on the number of alleles segregating at the locus in a population and the genotypes of the individuals tested (Gee et al. 1992). Early work with electrophoresis on cranes by Morgan, at the University of Maryland in 1975, indicated a limited amount of variation.

A recent technique in DNA analysis or **genetic fingerprinting** examines variations in DNA structure (Jeffries et al. 1985; Vassart et al. 1987). Enzymes (restriction endonucleases) are used to cleave DNA. The resulting fragments contain tandemly repeated sequences that are highly polymorphic (**RFLP**). Radiolabelled complementary probes have been developed to identify these fragments, providing fingerprints that are unique to each individual tested. These fingerprints provide an excellent means for identifying relatedness between individuals and to estimate population diversity (Jeffries et al. 1991; Geyer et al. 1993).

Several RFLP studies were conducted on cranes. Longmire et al. (1992) used a species-specific probe developed from Whooping Crane red blood cell DNA to examine relatedness and diversity in this species. Love and Dessauer used a species-specific probe to examine differences between Whooping Cranes and other closely related species (Love 1990). RFLP techniques have also provided a new technique for **sexing** cranes by developing a probe to identify repeat sequences characteristic to the W chromosome (see Chapter 11C).

Competitive binding immunoassays use a labelled antibody or antigen to detect immune reactions characteristic of individuals or groups of animals. Although this technique has been successfully used in

other species, attempts on cranes have proven unsuccessful to date (Gee et al. 1992).

Blood typing of the Major Histocompatibility Complex (MHC) is being used to determine relatedness and diversity in cranes. In this technique, antibodies are used to identify antigens which are controlled by individual gene loci. These loci segregate independently and can be used to estimate heterozygosity and relatedness.

Work is being conducted by W. E. Briles of Northern Illinois University, M. M. Miller at the Beckman Research Institute, and S. I. Jarvi at the Smithsonian Institution on the MHC in cranes (Jarvi et al. 1992). By absorbing (treating) known chicken specific reagents with the blood of a crane, it is now possible to prepare reagents capable of detecting individual forms of the MHC in a species of interest. Patuxent is collaborating to develop crane specific reagents using Sandhill Cranes. This study has been able to help elucidate the paternity of individual cranes. M. M. Miller is conducting molecular analysis of the MHC utilizing chicken MHC chromosomal DNA probes and developing additional species-specific probes through polymerase chain reaction and cloning techniques.

Information is also being collected on the relationship between MHC diversity and **disease resistance** (Allan and Gilmour 1962; Benacerraf 1981; Briles et al. 1983). MHC molecules bind antigens and activate the T cell response to foreign pathogens (Kurlander et al. 1992) playing an important role in the immune response. Maintaining MHC diversity may play a significant role in the survival of some endangered cranes. Breeding objectives based on MHC should be carefully integrated into strategies for maintaining genetic diversity across the entire genome (see Tables 9.1 and 9.2; Hughes 1991).



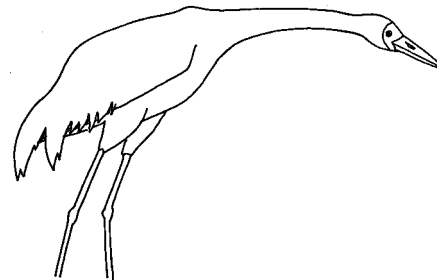
Lastly, genetic research can reveal **taxonomic relationships**. DNA-DNA hybridization (Sibley and Ahlquist 1983) and allozyme (Ingold 1984) studies supported the morphological and behavioral classification of 15 crane species. Allozyme analysis allowed genetic diversity estimates for Sandhill, Sarus, Siberian, and Whooping Cranes (Dessauer et al. 1992). Krajewski (1988), using DNA-DNA hybridization, found one group of five species so closely related that they could not be differentiated. In a later study, he separated the group into 5 distinct species using a highly polymorphic region of mtDNA (Krajewski and Fetzner 1994). Krajewski is also using mtDNA to evaluate the relationship of crane subspecies. Sheri Snowbank at Southern Illinois University is using mtDNA to determine maternal lineages of Whooping Cranes which survived the 1942 population bottleneck. Also, Travis Glenn at the Smithsonian Institution is using microsatellite DNA fingerprinting (RFLP) in museum specimens to estimate Whooping Crane genetic diversity before the 1942 bottleneck.

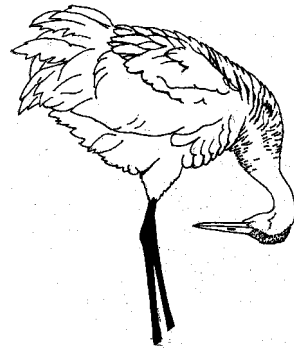
Future research needs include continued examination of relatedness of wild caught birds, completion of paternity analysis, and refinement and application of MHC studies.

Literature Cited

- Allan, C. P., and D. G. Gilmour. 1962. The B blood group system of chickens. III. The effects of two heterozygous genotypes on the survival and egg production on multiple crosses. *Genetics* 47:1711-1718.
- Benacerraf, B. 1981. Roles of MHC gene products in immune regulation. *Science* 212:1229-1236.
- Briles, W. E., R. W. Briles, R. E. Taffs, and H. A. Stone. 1983. Resistance to a malignant lymphoma in chickens is mapped to subregion of major histocompatibility (B) complex. *Science* 219:977-979.
- Crow, J. F., and M. Kimura. 1970. An introduction to population genetics theory. Harper and Row, New York. 591 pp.
- DeBoer, L. E. M. 1989. Founder representation: how to deal with it. *In* EEP Coordinator's Manual. National Foundation for Research in Zoological Gardens and EEP Executive Office, Amsterdam Zoo.
- Denniston, C. 1978. Small population size and genetic diversity: implications for endangered species. Pages 281-289 *in* S. A. Temple, editor. *Endangered birds: management techniques for preserving threatened species*. University of Wisconsin Press, Madison.
- Dessauer, H. C., G. F. Gee, and J. S. Rogers. 1992. Allozyme evidence for crane systematics and polymorphisms within populations of Sandhill, Sarus, Siberian, and Whooping Cranes. *Molecular Phylogenetics and Evolution* 1:279-288.

- Flesness, N. R. 1977. Gene pool conservation and computer analysis. *International Zoo Yearbook* 17:77-81.
- Foose, T. J., and J. D. Ballou. 1988. Population management: theory and practice. *International Zoo Yearbook* 27:26-41.
- Frankel, O. H., and M. E. Soule. 1981. Population genetics and evolution. Pages 32-77 in O. H. Frankel and M. E. Soule, editors. *Conservation and evolution*. Cambridge University Press, Cambridge, U.K.
- Frankham, R., H. Hemmer, O. A. Ryder, E. G. Cothran, M. E. Soule, N. D. Murray, and M. Snyder. 1986. Selection in captive populations. *Zoo Biology* 5(2):127-138.
- Gee, G. F., H. C. Dessauer, J. Longmire, W. E. Briles, and R. C. Simon. 1992. The study of relatedness and genetic diversity in cranes. Pages 225-230 in D. A. Wood, editor. *Proceedings 1988 North American crane workshop*. Florida Nongame Program Technical Report #12.
- Geyer, C. J., O. A. Ryder, L. G. Chemnick, and E. A. Thompson. 1993. Analysis of relatedness in the California Condors from DNA fingerprints. *Molecular Biology and Evolution* 10:571-589.
- Hughes, A. L. 1991. MHC polymorphism and the design of captive breeding programs. *Conservation Biology* 5:249-251.
- Ingold, J. L. 1984. Systematics and evolution of the cranes. Ph.D. dissertation, Miami University, Oxford, Oh. 62 pp.
- Jarvi, S. I., G. F. Gee, W. E. Briles, and M. M. Miller. 1992. Identification of segregating haplotypes of the major histocompatibility complex in cranes. *Proceeding North American Crane Workshop* 6:171.
- Jeffries, A. J., V. Wilson, and S. L. Thein. 1985. Individual-specific "fingerprints" of human DNA. *Nature* 316:76-79.
- Jeffries, A. J., M. Turner, and P. Debenham. 1991. The efficiency of multilocus DNA fingerprint probes for individualization and establishment of family relationships, determined from extensive casework. *American Journal of Human Genetics* 48:824-840.
- Krajewski, C. 1988. Phylogenetic relationships among cranes (Aves: Gruidae) based on DNA hybridization. Ph.D. dissertation, University of Wisconsin, Madison. 342 pp.
- Krajewski, C., and J. W. Fetzner, Jr. 1994. Phylogeny of cranes (Gruiformes: Gruidae) based on cytochrome-b DNA sequences. *Auk* 111:351-365.
- Kurlander, R. J., S. M. Shawar, M. L. Brown, and R. R. Rich. 1992. Specialized role for a murine class I-b MHC molecule in prokaryotic host defenses. *Science* 257:678-679.
- Longmire, J. L., G. F. Gee, C. L. Hardekopf, and G. A. Mark. 1992. Establishing paternity in Whooping Cranes (*Grus americana*) by DNA analysis. *Auk* 109:522-529.
- Love, J. 1990. Avian repetitive DNA. Ph.D. dissertation, Louisiana State University, New Orleans. 136 pp.
- Mettler, L. E., and T. G. Gregg. 1969. Population genetics and evolution. Prentice-Hall, Inc., Englewood Cliffs, N.J. 212 pp.
- Miller, P. S., and P. W. Hedrick. 1991. MHC polymorphism and the design of captive breeding programs: simple solutions are not the answer. *Conservation Biology* 5(4):556-558.
- Odum, R. A. 1994. Assimilation of new founders into existing captive populations. *Zoo Biology* 13:187-190.
- Ralls, K., J. Ballou, and A. Templeton. 1988. Estimates of lethal equivalents and the cost of inbreeding in mammals. *Conservation Biology* 2:185-193.
- Sibley, C. G., and J. E. Ahlquist. 1983. Phylogeny and classification of birds based on the data of DNA-DNA hybridization. *Current Ornithology* 1:245-292.
- Sheppard, C. 1988. White-naped Crane masterplan, September, 1988. New York Zoological Society. 58 pp.
- Soule, M. 1983. What do we really know about extinctions? Pages 111-124 in C. M. Schonewald-Cox, S. M. Chambers, B. MacBryde, and L. Thomas, editors. *Genetics and conservation: a reference for managing wild animal and plant populations*. Benjamin/Cummings Publishing Company, Inc., Menlo Park, Calif.
- Swengel, S. R. 1985. Genetic and demographic management of captive Red-crowned Cranes *Grus japonensis*. *Avicultural Magazine* 91(1-2):41-48.
- Swengel, S. R. 1987. A proposed management plan for captive Red-crowned Cranes. Pages 513-523 in G. W. Archibald and R. F. Pasquier, editors. *Proceedings of the 1983 International Crane Workshop*. International Crane Foundation, Baraboo, Wis.
- Vassart, G., M. Georges, H. Brocas, H. S. Lequarre, and D. Christophe. 1987. A sequence in M13 phage detects hyper-variable minisatellites in human and animal DNA. *Science* 235:683-684.
- Wiese, R. J., and K. B. Willis. 1993. American Association of Zoological Parks and Aquariums Species Survival Plan Central Dogma. In *Small Population Management Advisory Group Development Workbook*. American Association of Zoological Parks and Aquariums, Bethesda, Md.
- Wilcox, B. A., P. F. Brussard, and B. G. Marcot. 1986. The management of viable populations. Center for Conservation Biology, Stanford University, Stanford, Calif. 188 pp.
- Willis, K. B., and R. J. Wiese. 1995. Genetic variation maintenance strategies for captive breeding programs. *Journal of Aquaculture and Aquatic Science*. In press.





Records

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Fundamental to good management of a captive flock is keeping detailed records of important life history events and genealogy. The knowledge of family relationships is essential to genetic management in long-term captive breeding programs. Record keeping systems should organize and store this information in an easily accessible format. This allows evaluation of current and historical management practices. Standardization of data collection and reporting between centers can increase effective sample size and simplify data analysis.

Crane record keeping systems have been evolving at Patuxent since 1966 and at ICF since 1974 (Ellis et al. 1991). A detailed description of Patuxent's manual (non-computerized) system is available in published form (Ellis et al. 1991) and examples of the most recent data forms are available from the authors. Herein we outline the information that we consider essential for the long-term management of a captive crane colony. We also include information that will be helpful for some collections involved with more specialized uses. Our presentation is of a manual system (Fig. 10.1) organized to allow for limited redundancy while promoting ready access to each form. We have included Patuxent's form sheets (Figs. 10.2-10.13) and information cells (IC's) to provide useful information from other systems (cited later) and our own observations.

ARKS and Other Computerized Records Systems

The need for a complex computerized records system for general use by many institutions was identified by the Committee on Laboratory Animal Records (1979). To facilitate the keeping of good records on animal colonies, several systems for efficiently maintaining specimen inventory, health, productivity, and vital records have appeared (Brown 1975; Seal and Makey 1975). Most widely used in the avicultural community

is the computerized International Species Information System (ISIS) designed to promote uniform reporting of basic demographic information for individual specimens throughout the network of subscribing institutions (Seal et al. 1976, 1977). With over 460 institutions participating (1995), ISIS is rapidly becoming the most common animal record keeping system worldwide.

Animal Record Keeping System III (ARKS III) is the computer software provided to ISIS participants (utilizing an IBM compatible microcomputer with at least 512K RAM and a hard disk with a minimum of 10 megabytes). ARKS III enables maintenance of accurate up-to-date records. It is also user friendly. The ARKS III system provides useful summary reports and statistics on current inventory, births, deaths, and transfers. Details of events for individual animals are not included, so it is often desirable to augment the system with more detailed records on reproduction, behavior, etc.

The inventory of a zoological facility is easily edited and checked using the ARKS III program. By merely selecting the ISIS report function and inserting a disk, the data can be transcribed onto the disk and made ready for mailing to ISIS or other institutions. This procedure allows for a regularly updated international inventory of all individuals of any species (or other taxon) for all member institutions. From this information, ISIS produces valuable taxon reports summarizing basic genetic and demographic information. Institutions can also easily provide diskette copies of an individual animal's record, allowing efficient transfer of information when an animal is shipped.

Information entered for each specimen is coded for easy retrieval of various reports. ARKS III is currently able to provide 12 reports which are useful for in-house management. They are as follows: Collection inventory; Specimen report; Taxon report; Transaction reports; Enclosure log; Reproductive history; Sibling tables; Pedigree (for animals within the institution); Inbreeding (local); Age pyramid; Fecundity and Mortality report; and International Zoo Yearbook (IZY) report.

ISIS has produced three other programs which aid in record keeping and data retrieval. MedARKS software provides medical records and reports for anesthesia, parasitology, clinical pathology, serum and tissue banking, text for clinical notes, and medication and vaccination records. There is flexibility in designing additional in-house modules. A pathology module is under development. MedARKS is a sufficiently complete system for medical records for a crane collection and is presently used that way at ICF.

SPARKS software enables coordinators of breeding programs to develop individual species studbooks and facilitates the management of breeding programs through genetic and demographic analyses and reports.

EGGS software, an egg log database, is currently under development. It will enable the complex reporting required for the management of a colony of breeding cranes. Currently functioning as relational databases, both MedARKS and EGGS cannot perform without up-to-date records in ARKS III.

Other computerized records systems are also available which can accommodate detailed information on productivity, health, and many life history events (e.g., Sciabarrasi and London 1974) and can be used for management of veterinary data (e.g., Castleberry et al. 1966; SNOMED 1976, 1977).

The Patuxent Records System

The Patuxent records system is presented schematically in Fig. 10.1. In designing records, each form is given a descriptive title and is assigned a letter which is referred to in the text. Using the alphabetical designators, that figure serves as an index to the text.

The crane colonies at Patuxent and ICF are characterized by few species and many breeding adults. The breeding birds in our collections are often endangered species or subspecies. Because of the need to closely monitor all pertinent aspects of growth and productivity for each individual, other computerized records systems presently available do not fulfill all of these needs.

Below, we describe a manual system designed at Patuxent to record the detailed information we require for each bird. We will also provide some details of the ICF system where it differs significantly from the Patuxent system. The system can also be modified for use with smaller collections and for colonies of other

avian species. We provide details on the use of the system including filing instructions and a table of appropriate responses for a sample of life history events. We indicate, for example, which records are created or modified when an egg is laid, a chick hatches, etc. The system is designed to allow for limited redundancy, for ease in manual filing and retrieval, and for ease in conversion to automated processing. Portions of the manual system have been automated in DOS and require an IBM compatible computer (minimum capacity 286) with 640K RAM. The automation process is continuing.

In designing the records listed on Fig. 10.1, we followed several conventions. First, to limit redundancy and for ease in record retrieval, whenever possible, use the individual identification number (ID, which also codes the leg band number in the Patuxent collection) as an index to all records where the specimen appears as an individual. In Patuxent's system, the ID also indicates the hatch year, so records filed by ID are also in chronological order. We recommend placing the year, taxon, and ID along the top or in the upper right corner of each form sheet. To avoid confusion in reporting reproductive success, we count all chicks reaching 70 days of age (hatch day is day 0) as fledged. Finally, we reserve a column on many forms for the initials (or name) of the person recording each entry. In some countries, this last provision is required by law for medical records.

In Fig. 10.1, the records are arranged from left to right chronologically by life history events. Individual and pair records are near the top followed by daily working records, summary records, planning records, and visual aids. Most of the records contain information about individual birds. Some, however, give readings from machines, and one, the Annual Time Line, provides a visual display of all regularly scheduled husbandry activities and the general phenology of the breeding season. Most of the records follow a highly structured format; an exception is the Daily Log. In all, 40 to 45 records and aids are treated in the narrative.

Individual and Pair Records

Individual File. A file, maintained for each adult crane, is created at fledging (70 days) when a Rearing Record (B, C) is closed and an Individual Log (R) is created. The file serves as a folder for all pertinent records on an individual. Records included in the file are the Individual Record (L), the Individual Log (R),

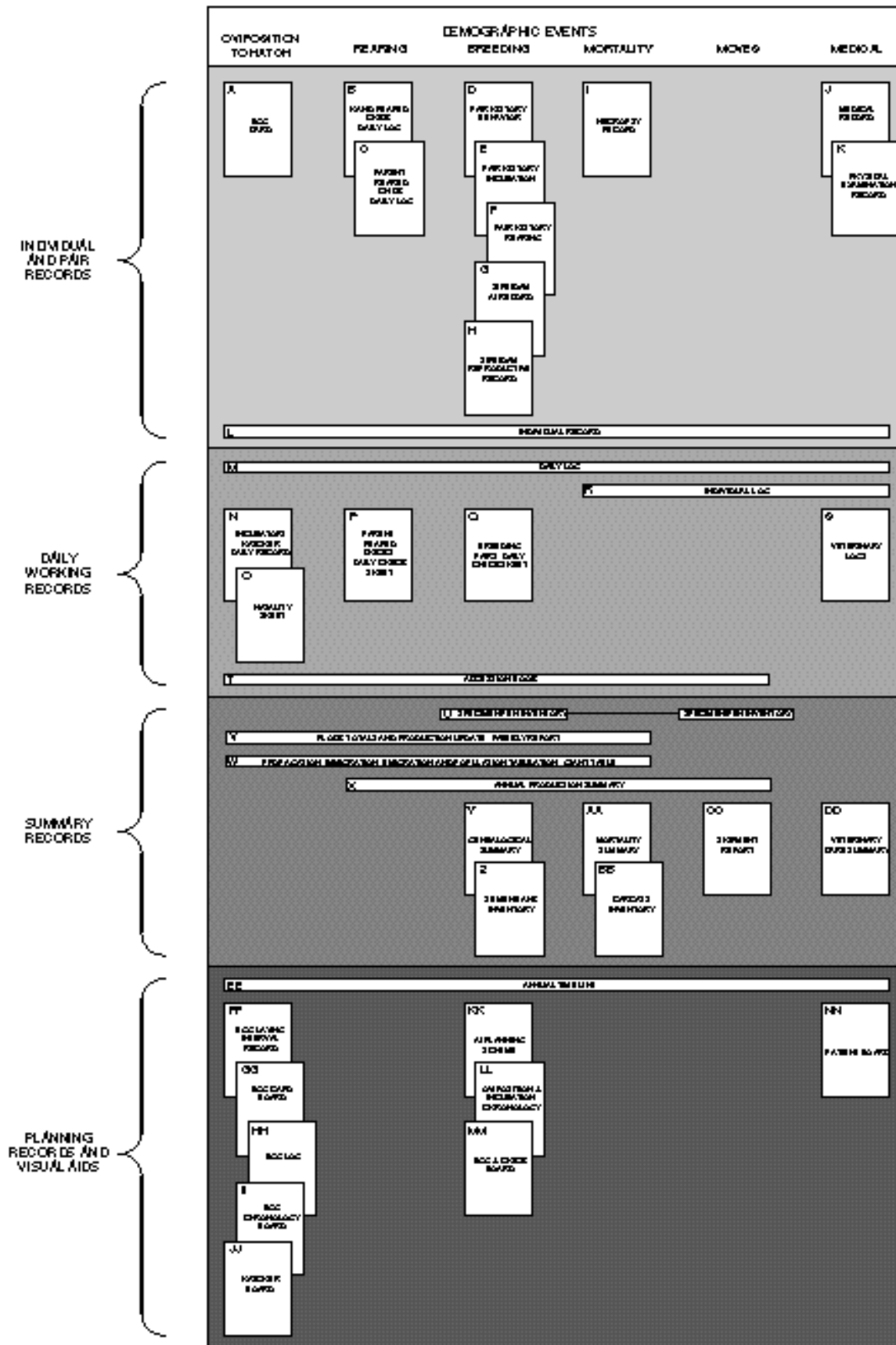


Fig. 10.1. Schematic of records system.

the Rearing Records (B, C), and the Genealogical Summary (Y). At Patuxent when an individual dies, all Medical Records (the Medical Record [J], the Physical Examination Record [K], and the Necropsy Record [I]) are transferred to the Individual File which is then transferred to the Mortality File. If an individual is transferred to another institution, all of the above records including breeding records are copied and transferred with the crane. The original file is then filed by taxon and ID, with files of other dispersed birds.

A. Egg Card. An Egg Card (Fig. 10.2) records all events from laying to hatching. The Egg Card is also used to record weight changes and to plot these against a normal weight loss curve so adjustments can be made in the incubation conditions. Hatching events are recorded on a separate Chick Hatching Record. Information recorded includes strength of calls and movements, and times of pip, rotation, and emergence. Abnormalities in position, egg waste, or the hatched chick are also noted. It is important to record the chick's ID number, tattoo, or name for reference on the Egg Card. Egg Cards are filed by year, taxon, and dam ID.

B. Hand-Reared Chick: Daily Log. Hand-reared chicks are weighed and inspected regularly. This record (Fig. 10.3), a running log on the progress of each chick through fledging, is posted on the door of the chick's pen. Weight gain is emphasized in this record because toe and leg deformities frequently result when growth is too rapid (Carpenter 1977, 1986). Data on physical condition, medical treatments, behavior, food consumption, hydration, socialization with other birds, and weight are recorded. Weight is also plotted against a normal weight change curve for the species and is used to decide if food withholding is necessary to decrease the likelihood of deformities. Once a chick has fledged, this record is transferred to the Individual File. Duplicate records may also be grouped by taxon and filed by year.

C. Parent Reared Chick: Daily Log. For many chicks reared by their own or foster parents, this record (Fig. 10.3) provides a running list of examinations, medical treatments, and behavioral observations through fledging. X's in the rows and columns of the form indicate the normal schedule for exams, treatments, and medications. Once the chick has

fledged, this record is transferred to the Individual File which is filed by taxon and ID. These records may also be grouped and filed by year.

D. Pair History: Behavior. For some pairs, especially those of endangered species, detailed behavioral notes are made throughout the breeding season. As a minimum, a running log should be kept indicating the degree to which a pair behaves as a social unit.

Included are annual notes on the general frequency of unison calling (Archibald 1976), the distance routinely maintained between birds, the presence or absence of key social and agonistic displays (Ellis et al. 1996) that indicate compatibility of mates, and peculiarities useful in signalling change in compatibility. Other behavior patterns useful in evaluating a pair are discussed in Chapter 6.

TAXON: <u>FSHC</u>		DAM ID: <u>05-83035</u>		YEAR: <u>1994</u>	
DATE LAID: <u>02/24</u>		PEN: <u>Y-44</u>		EGG NO: <u>2</u>	
TIME FOUND: <u>0715</u>		FOUND BY: <u>JMN</u>			
FERTILITY: <u>F</u> IF UNK			HATCHING DATE		
EMBRYO COND.: <u>ED MD LD</u>			PROJECTED: <u>03/26</u>		
DATE EXAMINED: <u>03/04</u>			ACTUAL: <u>03/26</u>		
METHOD: <u>CANDLED</u>			CHICK ID: <u>05-94002</u>		
EGG TRANSFERS					
DATE	LOC.	TO LOC.	COMMENTS (WEIGHT)		
<u>02/27</u>	<u>Y-44</u>	<u>to Petersime B</u>			
<u>02/28</u>	<u>Petersime B</u>	<u>to R12</u>			
<u>03/10</u>	<u>R12</u>	<u>to Petersime B</u>			
<u>03/24</u>	<u>Petersime B</u>	<u>to Hatcher A</u>			

TAXON	<u>Florida SHC</u>	EGG ID	<u>Y44 #2</u>	YEAR	<u>1994</u>
Date	Time	Hatching	Comments	Initials	
<u>03/24</u>	<u>2130</u>	<u>Star pipped.</u>	<u>Moved to Hatcher A</u>	<u>JMN</u>	
<u>03/25</u>	<u>0740</u>	<u>Hole pip.</u>		<u>JT</u>	
	<u>1630</u>	<u>Chick hatching;</u>	<u>peeping strongly</u>	<u>MSE</u>	
<u>03/26</u>	<u>0700</u>	<u>Hatched.</u>	<u>Sprayed umbilicus with</u>		
		<u>Betadine</u>		<u>KOM</u>	
CHICK ID <u>05-94002</u>					

Fig. 10.2. Two sides of the Egg Card, Form A.

Pg. 1

HAND-REARED CHICK: DAILY LOG: YEAR 1994

TAXON Mississippi SHC ID 03-94070 BAND COLOR/ID Green 70

DATE HATCHED 05/31/94 LOCATIONS CCB-4 ORIGIN LFP 1 #1

Date	Age	Time	Wgt	Fec	Treatments/Comments	Int
5/31	0	2000	113		.11 cc Gentocin. Moved to CCB. Betadine on umbilicus.	KOM
6/1	1	0825	110		Looks good. Worked with mounted head (in costume). Good response. Ate a little.	KOM
6/2	2				1.2 cc Gentocin. Diphtheria exam. No problems.	KOM

PARENT-REARED CHICK: DAILY LOG (SIDE 1): YEAR 1994

TAXON WHOOPING CRANE
ID 02-94060

Pen B-23 Date Hatched 06/12/94

Date	Age (d)	Wt. (g)	P. Exam	Meds/Treatments			Fecal Exam	Comments
				Gent	Ipa	Other		
6/12	0	X	X	X				
6/13	1	X117	Xv	Xv		3cc LRS		Sprayed umbilicus. Administered .12cc Gent.
6/14	2	X120	Xv	Xv		3cc LRS		Dehydrated. Administered .12cc Gentacin
6/15	3	X129	Xv	Xv		3cc LRS		
6/16	4	X138	Xv	Xv		3cc LRS		
6/17	5							
6/18	6	X193	Xv	Xv				
6/19	7							
6/20	8							
6/21	9	X287	Xv					
6/22	10							
6/23	11							
6/24	12	X428	Xv		Xv		X yes	
6/25	13							
6/26	14							
6/27	15	X	X					
6/28	16							
6/29	17							
6/30	18	X770	Xv	Xv			X(-)	
7/01	19							
7/02	20							
7/03	21							
7/04	22							
7/05	23							
7/06	24							
7/07	25	X310	Xv	Xv			X yes	
7/08	26							
7/09	27							
7/10	28							
7/11	29	X560						
7/12	30							
7/13	31							
7/14	32	X720	Xv	Xv			X	
7/15	33							
7/16	34							
7/17	35							
7/18	36							
7/19	37							
7/20	38							
7/21	39	X290	Xv	Xv			X	
7/22	40							

*Special drugs or prophylactic treatments as prescribed by veterinarian.

PARENT-REARED CHICK: DAILY LOG (SIDE 2): YEAR 1994

TAXON WHOOPING CRANE
ID 02-94060

Pen B-23 Date Hatched 06/12/94

Date	Age (d)	Wt. (g)	P. Exam	Meds/Treatments			Fecal Exam	Comments
				Gent	Ipa	Other		
7/21	41							
7/22	42							
7/23	43							
7/24	44							
7/25	45							
7/26	46							
7/27	47	X3010	Xv					2290(on day 39) Looks good
7/28	48							
7/29	49							
7/30	50							
7/31	51							
8/01	52							
8/02	53	X3.3k	Xv	Xv		X no		3010(on day 47) wing check 6cm blood feathers
8/03	54							
8/04	55							
8/05	56							
8/06	57							
8/07	58							
8/08	59							
8/09	60	X3.5k	Xv					wing check 4cm blood feathers
8/10	61							
8/11	62							
8/12	63							
8/13	64							
8/14	65							
8/15	66							
8/16	67	X3.56	Xv	Xv		X yes		wing check. 2cm blood feathers. taped right wing. PE. drew lcc blood for sexing
8/17	68							
8/18	69							

*Special drugs or prophylactic treatments as prescribed by veterinarian.

Fig. 10.3. Chick rearing records, Forms B and C.

This form should include details of pair formation. For example, the record should indicate whether the birds were removed from a flock as a result of naturally choosing each other as mates, or whether the pair resulted from penning a male and female side by side until favorable behavior was observed. Details of pen numbers and duration of stay should be indicated here. This form is filed by taxon and male ID. Pair histories of extant pairs are filed separately from records of former pairs.

E and F. Pair History: Incubation and Rearing. These forms (Fig. 10.4) provide a year-by-year evaluation of a pair's performance in incubation and chick rearing for the duration of the pair's existence. File by taxon and male ID.

G. Sire/Dam Artificial Insemination Record. A detailed record (Fig. 10.4) is made for each bird involved in the AI program. This form is used to evaluate responses to AI and to provide the raw data for investigating topics such as timing of semen

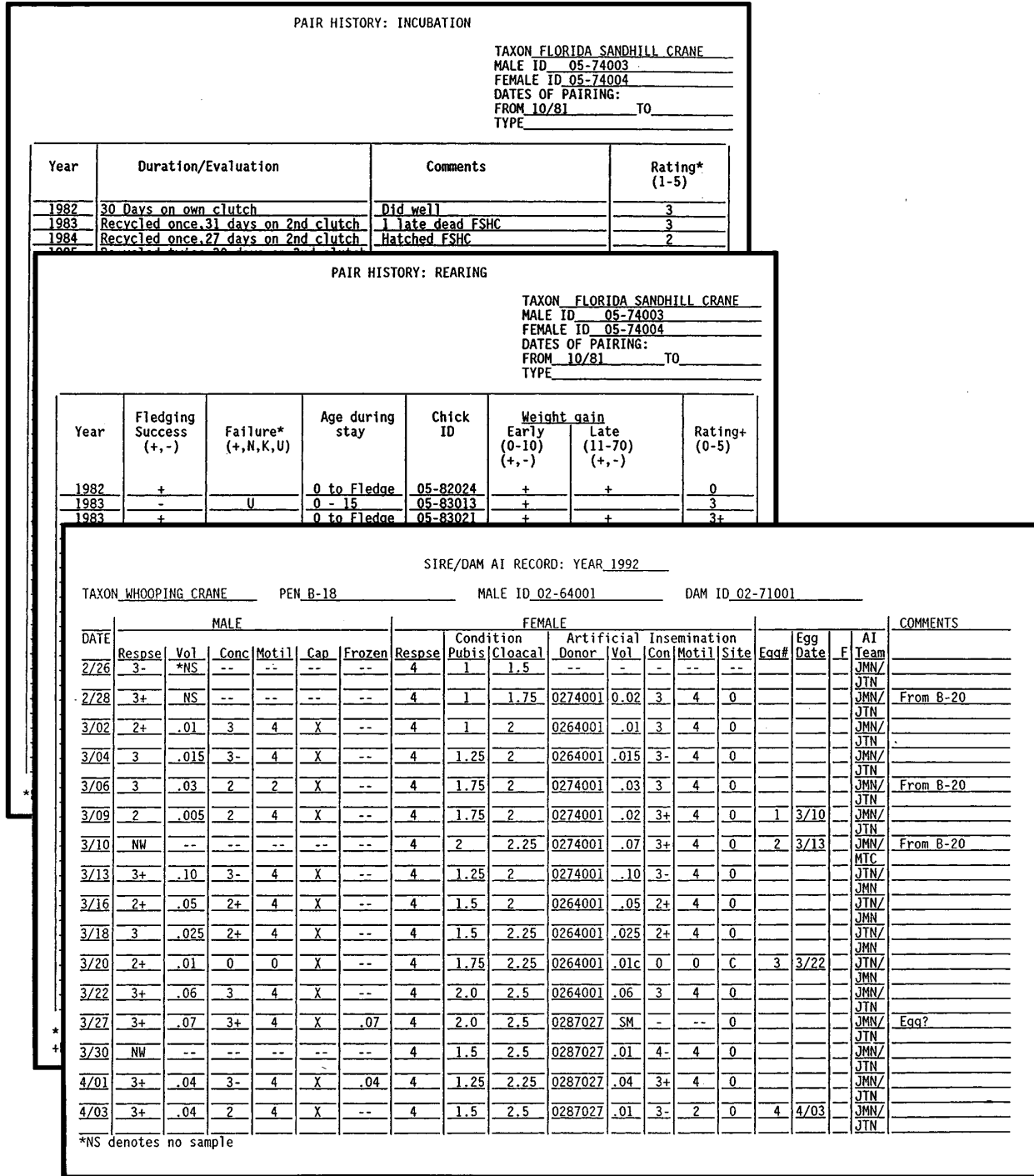


Fig. 10.4. Some breeding records, Forms E-G.

production, synchrony of mates, and suitability of AI techniques and paternity. Data is recorded for responses to AI, semen quality and quantity, and seasonal changes in the distance between pubic bones and condition of cloaca, both of which are indicators of the approach of egg laying (see Gee 1983 and Chapter 11A). Sire/Dam AI Records are filed by year,

taxon, and ID. The original copies of these forms, arranged by pen number, are included in a loose-leaf notebook that serves as an annual field log for AI.

H. Sire/Dam Reproductive Record. The Sire/Dam Reproductive Record (Fig. 10.5) provides a cumulative list, egg by egg, of the reproductive performance of each breeding pair. Not only can fertility

SIRE/DAM REPRODUCTIVE RECORD

PEN B-11
 DAM ID 02-85002
 TAXON WHOOPIING CRANE
 YEAR 1994

Egg No.	Clutch No.	Mo-Day Laid	Egg Measurements			SIRE ID				PROGENY						Comments	
			Length	Wid.	Init Mass	Fert/Emb Condition	AI (Y.N)	Positive	Others Possible	Mo-Day Hatch	Hatch Wt.	Chick ID	Rear Meth	Loc.	Flg. (+,-)		Mo-Day Death
1	1	3/14	102.2	61.5	212.4	IF	N										
2	1	3/17	106.0	60.9	218.3	F	N	02-86027	none								Died during hatching
3	2	3/22	103.8	63.1	225.4	F	N	02-86027	none	4/21	115.4	02-94019	HR	CCB	+		
4	3	3/29															
5	3	4/1															
6	4	4/5															
7	4	4/9															
8	4	4/13															Pulled from egg -- died

MEDICAL RECORD: YEAR 1991

Clinic Cage _____ Cage HOSP-1
 Attending Clinician DR. OLSEN
 Student _____
 Fluids: _____
 Type to Administer 2 1/2% Dextrose Amt/24 hrs 240 cc Rate 120/am
 Medications Amt Rate Start Time
Amikacin 0.23cc BID 0800
Piperacillin 2.8cc BID 0800

Special Procedures _____ Instructions _____
Radiographs, surgery, post-op care
Blood work-up
Monitor food/weight loss
Repeat radiographs in 2 weeks

Medications/Treatments/Observations

Date	Time	Body Wt.	Feedr Wt.	
3/22	0700	5.75		
3/23	0815	5.85	6.0	
3/24	0830	5.60	6.0	
3/25	0800	5.4	5.95	
3/26	0815	5.15	5.85	

PHYSICAL EXAMINATION FORM

Date 9/18 Year 1994 Taxon Mississippi Sandhill Crane
 ID 03-94086
 Name _____ Pen W-15 Age YOY Sex U

Pertinent History _____

GENERAL
 Body Condition Index (BCI) 3 Weight 4.4 kg
 Attitude/Conformation/Posture OK

EXAMINATION

Head: Eyes OK
 Eyelids _____
 Cere _____
 Nares _____
 Tongue/Mouth _____
 Beak _____
 Upper Respiratory _____
 Ears _____

Neck: _____
 Thorax: Sternum _____
 Auscultation _____
 Respiration _____

Abdomen: Palpation _____
 Vent _____

Limbs: Wings _____
 Legs Scrape on R hock (anterior); sprayed with Furazolidine
 Feet OK
 Nails _____

Integument: Skin _____
 Feathers _____

SAMPLES AND TREATMENTS
 Blood: Amount 8 ml, Site: Jugular X, Other _____, Serum _____ ml,
 EDTA _____ ml, Heparin _____ ml, Other _____ ml
 Feces: Pen or Individual, Color and Consistency _____
 Treatments: Internal Parasites: Product Ivomec Route Sq Amount 0.09ml
 External Parasites: Product _____
 Other Treatments/Comments Cloacal Swab to test for Salmonella
Took lcc blood for sexing

CLINICAL PATHOLOGY
 Blood: Hematocrit 42 % Total Solids 5.5 H/E Count 7520
 Heter _____ % Lymph _____ % Mono _____ % Eosin _____ % Baso _____ %
 WBC Count _____ Chemistry Results Attached _____
 Fecal: Flotation Capillaria, ascarids Direct Smear _____

Fig. 10.5. Some breeding and medical records, Forms H, J, and K.

and hatchability be evaluated from this form, but any aberrance in progeny survival is also apparent. This form is prepared throughout the breeding season in conjunction with the Egg Cards (A), Egg Logs (HH), and Sire/Dam AI Records (G), and is filed by year, taxon, and ID.

I. Necropsy Record. On this form, record details of the post-mortem examination and histopathological tests. IC's provide: location and time of death, weight, general body condition, a brief medical history, anatomical abnormalities, and name of performing pathologist. The form lists tissues and other materials retained, carcass deposition, information on diagnosis, and, where possible, cause of death. We recommend the tabular separation of the most frequent causes of death for ease in data extraction. This form is filed by taxon and ID. A copy may also be placed in the Mortality File in the Individual File.

J. Medical Record. For chicks, the Hand-reared (B) or Parent-reared Chick: Daily Log (C) serves as a

medical record. For a fledged bird, a separate Medical Record (Fig. 10.5) is created when the bird first shows clinical signs requiring veterinarian care. Medical Records include treatment instructions, a summary of treatments, and personnel involved in patient care as well as a description of the injury or disease. File by taxon and ID, with live bird records filed separately from dead. Copies of the Medical Record of dead birds are included in the Mortality File, which becomes part of the Individual File, and also with other medical records.

K. Physical Examination Record. In the fall of the year, each bird is given a physical examination (annual health check). This record (Fig. 10.5) is filed by taxon and ID for living birds, and like other medical records of dead birds, it is placed in the Mortality File in the Individual File.

L. Individual Record. The Individual Record (Fig. 10.6) provides an overview of the life of an individual crane as well as an index to all other

INDIVIDUAL RECORD					
Hatch date <u>06/02/71</u> or		ID <u>02-71001</u>			
Date of acquisition <u>05/18/71</u>		Name <u>Mrs. C.</u>			
Age on acquisition <u>Egg</u>		Taxon <u>Whooping Crane</u>			
Origin <u>WBNP (Klwi 1)</u>		Sex <u>Female</u>			
TRANSACTION RECORD:		Method/Behav. <u>Date5/73</u>			
Transaction Date		Sire ID <u>Wild</u>			
Former Owner		Dam ID <u>Wild</u>			
Former ID		Other IDs _____			
New Owner					
New ID					
Transaction Type					
INCUBATION METHOD: _____					
ARTIFICIAL INCUBATORS:	Type:	Dates			
	<u>Suitcase</u>	<u>5/17 -- 5/18</u>			
	<u>Petersime B</u>	<u>5/18 -- 6/02</u>			
FOSTER PARENT INCUB:	Location/ID's	Dates			
	_____	_____			
REARING METHOD:	Type	Age of Chick	Foster Parents		
	<u>Hand</u>	<u>0 -- 90 days</u>	(Species and ID)		
FLEDGE DATE (70 Days) <u>08/12/71</u>					
FLIGHT RESTRAINT METHODS	Type	Date			
	<u>Tenotomy</u>	<u>08/15/71</u>			
POST FLEDGING LOCATION and MATE					
Pen No.	Dates	Pen No.	Dates and Mate ID		
<u>BYP 1</u>	<u>09/10/71</u>				
<u>G1/2</u>	<u>10/15/76</u>	<u>G1/2</u>	<u>10/15/76</u>	<u>02-64001</u>	
<u>B17/18</u>	<u>11/10/85</u>	<u>B17/18</u>	<u>11/10/85</u>	<u>02-64001</u>	
MAJOR MEDICAL PROBLEMS: Date _____ Type _____					
PHYSICAL ABNORMALITIES <u>Tenotomized left wing</u>					
BEHAVIORAL CHARACTERISTICS					
<u>Not aggressive, turns on well during AI, fair incubator, good at parenting</u>					

INDIVIDUAL RECORD (CONTINUED)										
REPRODUCTIVE HISTORY IF FEMALE										
Year	Eggs			Fertility (F/F+I) (%)	Hatched (%)		No. Fledged (> 70 days) (%)			
	Total laid	No. fert.	No. infert.		No. unkn.	No.	(%)	No.	(%)	
<u>1980</u>	<u>3</u>	<u>1</u>	<u>1</u>	<u>1</u>	<u>1/2</u>	<u>50.0%</u>	<u>1</u>	<u>100%</u>	<u>0</u>	<u>0%</u>
<u>1981</u>	<u>4</u>	<u>2</u>	<u>2</u>	<u>2/4</u>	<u>50.0%</u>	<u>1</u>	<u>50%</u>	<u>1</u>	<u>100%</u>	
<u>1982</u>	<u>6</u>	<u>4</u>	<u>1</u>	<u>4/5</u>	<u>80.0%</u>	<u>2</u>	<u>50%</u>	<u>1</u>	<u>50%</u>	
<u>1983</u>	<u>6</u>	<u>5</u>	<u>0</u>	<u>5/5</u>	<u>100.0%</u>	<u>3</u>	<u>60%</u>	<u>1</u>	<u>33.3%</u>	
<u>1984</u>	<u>5</u>	<u>5</u>	<u>0</u>	<u>5/5</u>	<u>100.0%</u>	<u>4</u>	<u>80%</u>	<u>3</u>	<u>75%</u>	
SEMEN PRODUCTION SUMMARY IF AI MALE										
Year	No. attempted collections	No. samples collected	No. good samples	Date of 1st samp	Date of last samp	No. of days semen prod	Average volume			
ID OF PROGENY: _____, _____, _____, _____, _____, _____										
REPRODUCTIVE HISTORY IF NATURALLY BREEDING MALE										
Year	Mate ID	Eggs			Fertility (F/F+I) (%)	Hatched (%)		No. Fledged (> 70 days) (%)		
		Total Laid	No. Fertile	No. Infert.		No. Unknown	Ratio (%)	No.	Ratio (%)	
DEATH										
Date	Necropsy No.		Deposition of Carcass							
Cause of Death _____										

Fig. 10.6. Individual Record, Form L.

records where this bird appears as an individual. Details of various phases of the crane's life are kept on more specific records (discussed later). This and several other records are filed by taxon and ID in the Individual File. Separate files are maintained for dead or dispersed birds.

Daily Working Records

Daily logs or reports are useful for immediate communication between staff and to temporarily record information while working at sites remote from the records room. At ICF, a daily log is used year round and supplemented with a chick report during the breeding season. At Patuxent, the following daily work records are used.

M. Daily Log. Animal caretakers make a preliminary record of husbandry activities in the Daily Log. Thereafter, many details (such as pen-to-pen moves, unusual behavioral observations, and injuries or illnesses) are transferred to the Individual Log (R) or specific record sheets, but the Daily Log from each year is retained and filed chronologically. This is the only record of many routine activities.

N. Incubator/Hatcher Daily Record. This form (Fig. 10.7) provides a log of mechanical incubator and hatcher temperature and humidity conditions. Typically, readings are taken 2-4 times per day. These reports are filed chronologically and by machine number.

O. Natalty Sheet. This form (Fig. 10.7), a chronological list of all natalities, is used to assign ID numbers to chicks at hatching time. Entries are made manually as each chick hatches and the form can be generated by computer from Sire/Dam Reproductive Records. Filing is by year with the Egg Cards (A) or chick hatching records.

P. Parent-Reared Chicks: Daily Check Sheet. One copy of this form (Fig. 10.7) is used each day as the chick care teams travel through the colony to examine and/or provide medical treatments to all chicks being reared by crane pairs. After the tour through the collection, the information for each chick is transferred to the appropriate Parent-Reared Chick: Daily Log (C). Thereafter, a Daily Check Sheet is prepared for the next day by transferring information from the previous day's Daily Check Sheet and the examination/medication schedule on the Parent-Reared Chick: Daily Log (C). These forms are filed chronologically by year.

Q. Breeding Pairs: Daily Check Sheet (Walk Through Sheet). This check sheet (Fig. 10.8) is prepared daily for the caretaker to use while walking through the crane colony to inspect each pair, record nest condition, determine the number of eggs or chicks, and to evaluate nest attendance and other adult behavior. One copy of the form is sufficient for 70 pairs. At the end of the day, information on egg and chick numbers is transferred to the form for the following day. This form is used in planning egg moves and in rating each pair's incubation and rearing performance at the end of the breeding season.

R. Individual Log. A running log of events, from fledging to death, pertaining to the individual is kept on this form (Fig. 10.8). The information is later transferred to more specific records such as Individual Record (L), Pair History: Behavior (D), and Medical Record (J). This log is filed in the Individual File.

S. Veterinary Logs. Three log books provide a chronological record of veterinary activities. First, for each mortality, the Necropsy Log includes postmortem findings, samples (cultures and other materials), fate of carcass, and diagnosis. Second, the Radiology Log indexes the radiograph files, and third, the Laboratory Log reports detailed results of microbial cultures, fecal examinations, blood tests, etc. Filing is chronological by year for each log.

T. Accession Book. The Accession Book (Fig. 10.8) is the most fundamental record kept for an animal colony. Akin to the specimen catalog for a museum, it is a cumulative log of all cranes that have been or are a part of the collection. Various taxa can be logged in different books, or more commonly, all are logged in the same book, chronologically, according to arrival date or hatching or fledging date (for chicks originating at the facility). All birds that reach fledging age are included, but chicks that die before fledging may or may not be included. Birds that are owned by the institution but housed elsewhere are also included. Accession book records can be easily maintained on ARKS III.

INCUBATOR/HATCHER RECORD: YEAR 1993

Machine name: Petersime B

Set at 99.5 °F dry bulb and 87.0 °F wet bulb.

Date	Time	Dry Bulb (F)	Wet Bulb (F)	Tray Position	Comments	Init
3/18	1510	99.7	85	Down		KB
3/19	0800	99.3	86.5	Down	Cleaned/fumigated	KB
	1530	99.5	88	Up	Opened vent slightly	KB
3/20	0900					
	1630					
3/21	0815					
	1530					
3/22	0825					
	1530					
3/23	0800					
	1540					
3/24	0820					
	1530					
3/25	0900					
	1630					
3/26	0730					
	1700					
3/27	0835					
	1515					
3/28	0800					
	1535					
3/29	0820					
	1600					
3/30	0750					
	1540					
3/31	0715					
	1510					
4/1	0725					
	1530					
4/2	0700					
	1610					
4/3	0815					
	1545					
4/4	0900					
	1700					
4/5	0845					
	1510					
4/6	0800					
	1645					
4/7	0810					
	1550					
4/8	0755					
	1530					
4/9	0800					

NATALITY SHEET: YEAR 1991

Hatch Order	Taxon	ID	Origin	Hatch Date	Rearing Methods	Locations	Comments
1	FSHC	05-91001	Y44 #1	3/26	HR	CCB	Died 7/13
2	FSHC	05-91002	Y44 #2	3/26	HR	CCB	
3	FSHC	05-91003	R12 #1	3/29	HR	CCB	
4	FSHC	05-91004					
5	FSHC	05-91005					
6	WC	02-91006					
7	MSHC	03-91007					
8	MSHC	03-91008					
9	MSHC	03-91009					
10	WC	02-91010					
11	MSHC	03-91011					
12	WC	02-91012					
13	FSHC	05-91013					
14	FSHC	05-91014					
15	GSHC	04-91015					
16	WC	02-91016					
17	GSHC	04-91017					
18	GSHC	04-91018					
19	MSHC	03-91019					
20	GSHC	04-91020					
21	MSHC	03-91021					
22	GSHC	04-91022					
23	FSHC	05-91023					
24	MSHC	03-91024					
25	GSHC	04-91025					

PARENT-REARED CHICKS: DAILY CHECK SHEET: YEAR 1994

Date 05/18/94

Pen	Chick Tx-ID	Age (d)	Wt. (g)	P. Exam	Water	Int*	Meds/Treatments	Other Treat.	Fecal Exam	Comments
R43	03035	20			X					
R35	03067	3	X 120	X	X	Xy		1.5ccF1		110g day 2. checked hydration: ok
R33	05050	12	X 300	X	X		Xy		X	205g day 9. ewewash R eye. grass: ok
R31	05064	3	X 128	X	X					120g day 2
R27	03058	5			X					
R25	05053	9	X 290	X	X					190g day 6
R23	03057	6	X 150	X	X		X			110g day 3
R17	03071	1	X 110	X	X	X		1.5ccF1		110g day 0
R06	03036	19			X					
B11	02054	8			X					ok
B20	02051	11			X					ok
B22	05048	12	X 330	X	X		X		X	230g day 9
B15	05038	18	X 740	X	X		X		X no	550g day 15
B04	04047	12	X 470	X	X		X		X no	300g day 9
B02	05050	12	X 300	X	X		X		X	205g day 9; ewewash L eye - grass. PE ok
G11	03060	5	X 135	X	X					110g day 4; check wt. loss. hairy R hock
G11	03068	3	X 120	X	X			1.5ccF1		115g day 2. check vent
G01	03065	3	X 120	X	X					110g day 2; check vent; clean. borderline hydration. 1.5ccF1
Y21	03062	4			X					
Y41	03055	8			X					
Y25		0	X 90	X	X	X		1.5ccF1		
Y27	03063	4	X 95	X	X		X	AmPipBid tube fd fluids		95g on day 3; 2cc tube food
Y29	03061	4			X					
Y07	05049	12	X 460	X	X		X		X	370g day 10
Y05	03066	3	X 110	X	X					100g day 2
Y24	03055	4	X 95					AmPipBid 2ccTF 2ccF1		

Specific Medication Instructions:

Fig. 10.7. Some daily working records, Forms N-P

BREEDING PAIRS DAILY CHECK SHEET: YEAR 1994

Date: 05/13 Weather: 75 degrees, mild.

Pair	Comments	0600	1130	1530	eve.	Pair	Comments	0600	1130	1530	eve.
R43	E1 D1	FI DM	-1E+1E	FI		Y1					
R41	E2	FI		mI		Y3					
R39	E1 HF	CH		FB Cf		Y5	E2	FI E2	-2+1D	FU D1	

Page 1

INDIVIDUAL LOG

TAXON WHOOPING CRANE
ID 02-88051
NAME Tarzan
SEX Female

ORIGIN_WB 21A FLIGHT CAPABILITY FW REARING TYPE PR OTHER ID WT 88051

NOTE:

Date	Pen	BCI	Wgt.	Fecals	Observations/Comments	Init
08/16/88	B21	3	4.6		70 days: officially fledged	JMN
08/23/88					Blood taken, Vaccinated for EEE, Pulled blood	
				(-)	feather for sexing, Braided, Wing tagged	BC

Page 1

ACCESSION BOOK

ID	Taxon	Other ID	Name	Sex	Date	Hatch Place	Acquisition				
							Dam No. Sire No.	Names	Former Owner	Transaction Date	Type
8-022	G.a. Sharp11	130881	Pilai	0.1	08/13/81	ICF	08-006	Gloria	1	ICF	
							08-007	Painless	2		
8-023	*	170881	Atsuo	0.1	08/17/81	ICF	08-006	Gloria	1	ICF	Breeding Loan
							08-007	Painless	2		
8-024	*	020981	Sam Hammer-	1.0	09/02/81	ICF	08-006	Gloria	1	ICF	John Henry Dick Donation
							08-007	Painless	2		
							08-006	Gloria	1	ICF	

Page 2

ACCESSION BOOK (Page 2)

Date	Destination	Disposition	Transaction Type	Date	Location	Death	
						Cause of Death	Deposition
--	--	--	--	--	--	--	--
13/24/83	Vogelpark, Walsrode	Breeding loan		--	--	--	--
13/14/83	John Henry Dick	Donation - establish breeding prog/reintro		--	--	--	--
--	--	--	--	06/24/82	ICF	Raccoon predation	--
--	--	--	--	09/24/81	ICF	Leg problems	--
1/09/84	Thailand Depart. of Forestry	Donation - establish breeding prog/reintro		--	--	--	--
--	--	--	--	11/19/84	ICF	Post-surgical infection from hardware injection	--
1/09/84	Thailand Depart. of Forestry	Donation - establish breeding prog/reintro		--	--	--	--

Fig. 10.8. Additional daily working records, Forms Q, R, and T.

Summary Records

These records provide an overview of the colony and facilitate long-term management of a crane flock. All collections should have a system that, at least annually, summarizes flock size, acquisitions (births and transfers in), and dispositions (or depositions, i.e., deaths and transfers out). Patuxent uses the following records.

U. Specimen/Pen Inventory (Monthly Report). For each taxon, an automated Specimen Inventory (Fig. 10.9) is updated and prepared monthly. Individual birds of a taxon are listed by ID number, except for paired females who are listed immediately after their mates. A copy of this inventory is carried by caretakers when returning birds to pens and when locating an individual in the colony. A second automated version of this data, the Pen

SPECIMEN INVENTORY: DATE/YEAR <u>March 14, 1995</u>										Page <u>1</u>
ID	TAG	NAME	HATCH	ORIGIN	REAR	SEX	FLIGHT	PEN	COMMENTS	
02-84001	-	TEN	1984	PWRC UR #2	PR	M	FW	B-01	NEEDS METAL BAND	
02-85007	8595	-	1985	PWRC UL #5	HR	F	FW	B-01	-	
02-84002	-	PIRATE	1984	WBNP K-4	PR	M	FW	B-03	-	
02-87043	87043	-	1987	WBNP SK-3	PR	F	FW	B-03	-	
02-88058	88058	-	1988	WBNP K-6	HR	M	FW	B-05	-	
02-88051	-	TARZAN	1988	WBNP SK-4	PR	F	FW	B-05	-	
02-89035	89035	DAMIEN	1989	PWRC B12 #1	HR	M	BW	B-07	-	
02-91066	-	-	1991	WBNP #12	HR	F	BW	B-07	-	
02-88022	-	C.J.	1988	PWRC B18 #3	PR	M	FW	B-10	-	
02-83001	8335	CLIP	1983	PWRC UL #4	HR	F	FW	B-10	-	
02-86027	86027	ALTA	1986	WBNP ALTA	HR	M	FW	B-12	-	
02-85002	8591	LAZARUS	1985	PWRC UL #2	HR	F	AMP	B-12	MISS'G TIP RT WNG/PR DAY 0-10	

FLOCK TOTALS AND PRODUCTION UPDATE: WEEKLY REPORT									
Week <u>12/19 - 12/25</u>		Year <u>1994</u>							
TAXON	Living Stock <1 yr	Living Stock >1 yr	Prod. Pairs	No. Eggs	No. Hatch	Mort. This Wk <1 yr	Mort. This Wk >1 yr	Mort. This Yr <1 yr	Mort. This Yr >1 yr
Florida SHC	15	122	36	171	39	0	0	24	4
Greater SHC	8	82	26	92	13	0	0	5	8
Miss SHC	29	26	16	66	39	0	0	10	2
Whooping C	16	40	9	48	21	0	0	5	1

Fig. 10.9. Two summary records, Forms U and V.

Inventory, is indexed by pen complex and pen number, with individuals in the same pen listed numerically by ID, or with paired females listed immediately after their mates. This is the version used each day while the caretaker walks through the colony to check that each bird is in the appropriate pen.

A copy of each monthly update is placed chronologically in the archives to act as a historical description of the colony. ARKS III provides its own Specimen Inventory as a report.

V. Flock Totals and Production Update (Weekly Report). A weekly summary of flock size is prepared by balancing natality and acquisitions with mortality and departures. This report (Fig. 10.9) provides a summary for internal review and for informing cooperators of recent changes in captive colonies. The Weekly Report is our most useful document for reviewing egg production and chick survival during the breeding season. Weekly reports are filed chronologically.

W. Propagation, Immigration, Emigration and Population Tabulation (Giant Table).

The demography of a taxon within the colony is summarized for its entire history in the Giant Table (Fig. 10.10). Row headings are years. In our version, nearly 60 columns are divided into 7 major column groups. This table is updated at the end of each breeding season and is our most useful document for quickly summarizing demographic trends for each taxon within the colony.

X. Annual Production Summary. This report (Fig. 10.11), prepared at the end of the breeding season, provides an annual summary of egg and chick production for each dam, whereas colony summaries are provided in W, above. This report combines all of the information from the individual Dam Reproduction Records (H) and Egg Cards (A). Filing is by taxon and year with the Sire/Dam Reproductive Records (H).

Production Through Natural Fertility

Production Through Artificial Insemination

Adult Totals

Years	Males > 3 yrs	Semen prod. qual.	Females > 3 yrs	Layers	AI Pairs	AI Layers	AI Eggs potentially fertile	AI Eggs fertile	AI Eggs infertile	AI Eggs unknown fertility	AI Fertility ratio	AI Fertility percent	AI Hatching ratio	AI Hatching percent	AI Fledged ratio	AI Fledged percent	NF Pairs	NF Layers	NF Eggs potentially fertile	NF Eggs fertile	NF Eggs infertile	NF Eggs unknown fertility	NF Fertility ratio	NF Fertility percent	NF Hatching ratio	NF Hatching percent	NF Fledge ratio	NF Fledge percent	
1988	14		18	6	5	4	13	8	2	3	8/10	80	7/8	88	37	43													
1989	16		15	5	7	5	19	14	4	1	14/18	78	5/5	84	8/8	89													
1990	10		11	4	3	3	7	5	1	1	5/8	83	4/5	80	3/4	86													
1991	13		17	5	3	3	9	6	2	1	6/8	75	3/8	50	1/3	33													
1992	13		17	8	3	3	7	5	1	1	6/8	83	3/5	60	2/3	67													
1993	14		18	8	2	2	7	5	0	2	5/5	100	4/5	80	4/4	100													
1994	16		18	9	2	2	9	6	1	2	8/7	86	4/6	87	3/4	75													

Eggs and Chicks Sent Outside

Eggs and Chicks From Outside Sources (OUT)

O.E.P.

Years	Other egg production	Tot. Egg production/AI & NF	Eggs received	Eggs rec'd fertile	Eggs rec'd infertile	Eggs rec'd unknown fertility	Hatch year	After hatch year	Total	Years	Comments
1988	2	15	14*	8	0	6	8/5	100	8/5	100	
1989	0	18	16	7	6	2	7/13	58	8/6	100	
1990	3	14	0	1	4	1	11/12	92	6/11	56	
1991	0	21	16	11*	0	1	9/9	100	9/9	100	
1992	1	48	10	9	0	0	16/16	100	15/18	84	
1993	0	35	16	18	0	0					
1994	0	48	0								

Summary: Excluding OUT : Year End Flock Totals

Years	Sum. fertility ratio	Sum. fertility percent	Sum. hatching ratio	Sum. hatching percent	Sum. fledge ratio	Sum. fledge percent	Year	Comments
1988	8/10	80	15/16	94	10/16	67	1988	
1989	14/18	78	16/20	75	11/15	73	1989	
1990	5/10	50	4/5	80	3/4	75	1990	% not incl is 1 fert. Canadian SHC
1991	8/14	64	5/9	56	2/5	40	1991	*2 embryos in WB eggs DOA
1992	11/21	52	15/20	80	15/18	83	1992	
1993	16/24	67	20/32	94	24/30	80	1993	
1994	28/37	76	21/28	72	18/21	86	1994	

FIG. 10.10. Propagation, Immigration, Emigration, and Population Tabulation (Giant Table): Patuxent Whooping Crane data, Form W.

10/07/94

1994 ANNUAL PRODUCTION REPORT
WHOOPING CRANE

Dam ID	Origin	Egg	Laid	Fert	AI	Hatch	Sire ID	Chick ID	Rear	RearLoc	Flg	Comment
02-85007	B-02	01	03/30/94	U	N	--	--	--	--	--	--	3/30 Broken
02-85007	B-02	02	04/04/94	F/ED	N	--	02-84001	--	--	--	--	--
02-85007	B-02	03	04/05/94	F	N	05/03/94	02-84001	02-94041	H	CCB	(+)	--
02-85007	B-02	04	04/11/94	F	N	05/10/94	02-84001	02-94054	P	B-11	(+)	--
02-85007	B-02	05	04/14/94	I	N	--	--	--	--	--	--	--
02-87043	B-04	01	03/17/94	U	N	--	--	--	--	--	--	3/21 Broken
02-87043	B-04	02	03/21/94	F	N	04/20/94	02-84002	02-94018	H	CCB	(+)	--
02-87043	B-04	03	03/26/94	F/LD	N	--	02-84002	--	--	--	--	--
02-87043	B-04	04	04/09/94	F	N	05/08/94	02-84002	02-94052	H	CCB	(+)	--
02-87043	B-04	05	04/11/94	I	N	--	--	--	--	--	--	--
02-83001	B-09	01	03/30/94	U	N	--	--	--	--	--	--	3/30 Broken
02-83001	B-09	02	04/04/94	U	N	--	--	--	--	--	--	4/04 Broken
02-83001	B-09	03	05/03/94	F/LD	N	--	02-88022	--	--	--	--	--
02-83001	B-09	04	05/03/94	U	N	--	--	--	--	--	--	5/03 Broken
02-85002	B-11	01	03/14/94	I	N	--	--	--	--	--	--	--
02-85002	B-11	02	03/17/94	F/LD	N	--	02-86027	--	--	--	--	--
02-85002	B-11	03	03/22/94	F	N	04/21/94	02-86027	02-94019	H	CCB	(+)	--
02-85002	B-11	04	03/29/94	I	N	--	--	--	--	--	--	--
02-85002	B-11	05	04/01/94	I	N	--	--	--	--	--	--	--
02-85002	B-11	06	04/05/94	F	N	05/04/94	02-86027	02-94045	H	CCB	(+)	--
02-85002	B-11	07	04/09/94	I	N	--	--	--	--	--	--	--
02-85002	B-11	08	04/13/94	F/LD	N	--	02-86027	--	--	--	--	5/09 Embryo pulled from egg

02-83004 B-13
02-83004 B-13
02-83004 B-13
02-83004 B-13

02-87042 B-15
02-87042 B-15
02-87042 B-15
02-87042 B-15
02-87042 B-15
02-87042 B-15

02-71001 B-18
02-71001 B-18
02-71001 B-18

MORTALITY SUMMARY: YEAR 1987

No.	Taxon	Loc	ID	Origin	Htch Yr.	Sex	Death Date	Necrop No.	Deposit	Cause of Death
1	FSHC	R11	05-87005	R12 #4	1987	U	09/19	87216	Incin.	Found dead in pen in sleeping position
2	MSHC	G15	03-87064	G02 #6	1987	U	09/20	87217	Butler Bldg.	Found dead in pen in sleeping position
3	FSHC	-02	05-85051	Y43 #2	1985	F	09/22	87221	Incin.	Found dead in pen
4	FSHC	Y17	05-87032	Y32 #3	1987	U	09/23	87222	Incin.	Found dead -- dehydration
5	FSHC	IP4	05-87018	R42 #4	1987	U	09/24	87224	Incin.	Found dead in pen
6	FSHC	IP4	05-87021	Rock#4	1987	U	09/25	87226	Incin.	Found dead in pen
7	WC	B01	02-67001	WBNPS2	1967	M	09/25	87225	Butler Bldg.	Died at Hospital
8	FSHC	-02	05-84035	R13 #1	1984	F	09/25	87228	Incin.	Found dead in pen
9	GSHC	Y11	02-82023	GL14C	1982	M	09/25	87227	Incin.	Died at Hospital
10	WC	B16	02-83009	Col-orado	1983	M	09/25	87229	Butler Bldg.	Died at Hospital
11	FSHC	-02	05-80004	Rock#3	1980	M	09/26	87230	Incin.	Found dead in pen
12	WC	B02	02-83007	UL #1	1983	F	09/26	87232	Butler Bldg.	Died at Hospital
13	FSHC	-02	05-84036	R08 #1	1984	U	09/27	87233	Incin.	Found dead in pen
14	GSHC	Y35	04-87040	GL6	1987	U	09/27	87234	Incin.	Ran into fence, broke neck
15	GSHC	QP2	05-50004	GLNWR	1987	F	09/27	87235	Incin.	Malnourished, dehydrated
16	FSHC	R15	05-82037	R04 #3	1982	M	10/02	87242	Incin.	Died at hospital
17	GSHC	QP2	05-50001	GLNWR	1987	M	10/05	87244	Incin.	Died from complications after asperating tube food
18	FSHC	Y34	05-87006	R44 #3	1987	U	10/16	87248	Incin.	Deteriorated physically after contaminated feed study
20	GSHC	CCB	05-85021	GL-01	1985	F	10/27	87250	Incin.	Deteriorated physically after contaminated feed study
21	FSHC	R39	05-87054	R42 #4	1987	U	11/04	87262	Incin.	Severe pneumonia
22	FSHC	CCB	05-85047	R24 #4	1985	F	11/15	87269	Incin.	Contaminated feed study
23	GSHC	CCB	05-85027	GL25B	1985	U	11/21	87270	Incin.	Contaminated feed study
24	FSHC	CCB	05-85038	R18 #2	1985	U	11/06	87263	Incin.	Contaminated feed study
25	GSHC	QP2	04-50002	GLNWR	--	M	12/16	87287	Incin.	Trauma -- dislocated lft leg
26	MSCH	IP2	03-87065	MSCNWR Weber	1987	U	12/21	97290	Butler Bldg.	Trauma -- found dead in pen

caretaker

ormed legs
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liosis; resp

Fig. 10.11. Two summary records, Forms X and AA.

Y. Genealogical Summary. The genealogical records for each crane taxon are derived from Sire/Dam Reproductive Records and AI Records. An ancestral chart, similar to that used for humans and available from regional genealogical societies, is useful as a visual aid. This and other genealogical records for each individual bird are filed within the Individual File. Genealogical summaries can be produced by SPARKS software. Genealogical records, kept for all known captive individuals of a species in a studbook (e.g., Sheppard 1985 and Chapter 9), frequently include inbreeding coefficients.

Z. Semen Bank Inventory. This inventory system consists of a running log of samples entering the semen bank and a semen inventory file wherein all samples from an individual male are listed as they enter the bank. Detailed records on the handling of each sample (see Chapter 11B) are also made, but the essential details of semen quality, volume, cane number, location, and source are recorded on the inventory sheet for each donor. A separate record is also maintained for each cryopreservation tank. Column headings are taxon, ID, pen, date the sample was frozen, volume, location in tank, and comments.

Mortality File. Whenever a mortality occurs, the Mortality Summary (AA) is updated, and the Necropsy Record (I), Medical Record (J), Physical Examination Records (K), and all records in the Individual File are transferred to a Mortality File. Mortality Files are filed by year.

AA. Mortality Summary. Crane mortality is tabulated chronologically from the Necropsy Records and the Necropsy Log (one of the Veterinary Logs). At the beginning of each year, a new record is begun (Fig 10.11). Filing is by year. ARKS III can generate an inventory of all deceased birds.

BB. Carcass Inventory. A running list is kept of all carcasses by taxon. In addition, a tag, including death date, ID, and cause of death, is attached to each carcass. The Carcass Inventory includes data from the specimen tag, plus storage site, final deposition site, and necropsy number. This report is kept in the permanent files, however, for ease in data entry, a copy can be kept at the carcass storage site.

CC. Shipment Report. This is a running list of birds sent to or received from other institutions. Because it duplicates entries in the Accession Book (T), it may be unnecessary to keep this record (especially if the Accession Book is automated in ARKS III or otherwise). Column headings indicate date sent, taxon, ID's, recipient, and purpose of transfer.

DD. Veterinary Care Summary. From the records listed in the "medical" and "mortality" columns in Fig. 10.1, a Veterinary Care Summary can be prepared annually. Included are totals by sex and age class for surgical, diagnostic, and radiographic procedures, and for medications used. This provides an overview of significant medical activities for the year and can be used to identify trends and make recommendations for improved collection management. The MedARKS system also creates summary records sorted by time period, taxon, procedure, or problem, and is retrieved by key words.

Planning Records and Visual Aids

Wall-mounted charts or diagrams are useful in work areas to provide current information for management decisions. Patuxent has found the following visual aids useful. In addition to Patuxent's forms, ICF uses erasable boards or bulletin boards to display maps (of bird, pen, and egg locations), examination and treatment schedules for chicks, and a list of ongoing medical cases.

EE. Annual Time Line. This display board (Fig. 2.2) is a valuable aid in planning activities that occur for only a portion of the calendar year. By using different colors for the horizontal bars for each taxon, it is possible to display scheduled events for several taxa on one time line.

FF. Egg Laying Interval Record. The duration and timing of the egg laying period for each year and the intervals between eggs are summarized for each female in this report (Fig. 10.12). The timing of egg removal, important in assessing maximum productivity, should be indicated for each egg on the form (see Chapter 3). This form is filed by taxon, ID, and year.

GG. Egg Card Board. On this display board, each pen containing a pair of cranes, and each single but potentially productive female, is given a label. There are also separate labels on the board for each mechanical incubator. As eggs are laid, introduced, or removed, the Egg Cards (A) are affixed beneath the appropriate labels on the Egg Card Board. This board provides a quick update on the location of each egg in the breeding colony and each incubator. In conjunction with the Egg and Chick Board (MM), this board serves as one of the most useful visual aids in planning egg moves and other aspects of incubation.

EGG LAYING INTERVAL RECORD															
TAXON <u>Whooping Crane</u>															
ID <u>02-71001</u>															
Year	Total Eggs	Season Length	Last Egg	Date 1st Egg	Days between Eggs Nos. 1 - 11										
					2	3	4	5	6	7	8	9	10	11	
1979	1	--	--	30 May	--	--	--	--	--	--	--	--	--	--	
1980	--	--	--	--	--	--	--	--	--	--	--	--	--	--	
1981	5	31 days	11 June	16 May	2	9	2	13	--	--	--	--	--	--	
1982	7	34 days	18 May	4 Apr	2	8	3	6	4	8	--	--	--	--	
1983	8	42 days	14 May	2 Apr	2	14	7	4	9	3	--	--	--	--	
1984	10	50 days	27 May	7 Apr	3	8	4	6	7	4	8	5	5	--	
1985	6	45 days	21 May	6 Apr	2	10	19	3	11	--	--	--	--	--	
1986	--	--	--	--	--	--	--	--	--	--	--	--	--	--	
1987	1	--	--	8 Apr	--	--	--	--	--	--	--	--	--	--	
1988	3	22 days	20 Apr	29 Mar	3	19	--	--	--	--	--	--	--	--	
1989	7	52 days	26 Apr	15 Mar	3	14	2	12	3	8	--	--	--	--	
1990	4	39 days	25 Apr	17 Mar	14	13	12	--	--	--	--	--	--	--	
1991	7	43 days	27 Apr	15 Mar	4	10	3	9	12	5	--	--	--	--	
1992	6	37 days	16 Apr	10 Mar	3	9	12	12	5	8	--	--	--	--	
1993	5	25 days	14 Apr	20 Mar	3	7	13	2	--	--	--	--	--	--	
1994	6	35 days	25 Apr	21 Mar	2	8	4	8	13	--	--	--	--	--	

Fig. 10.12. Egg Laying Interval Record, Form FF

HH. Egg Log. This record (Fig. 10.13) provides a chronological list of all eggs laid for each taxon. It is useful as a visual aid in the management of egg moves and in updating the Weekly Report (V). Data from these forms are used for productivity summaries.

II. Egg Chronology Board. This aid can be used to follow the progress of a selection of the most valuable eggs (usually the eggs of endangered taxa). Column headings are dates. Rows are individual eggs. Each egg is represented by a 30-day-long adhesive strip affixed to the board. General events in the incubation of all eggs (e.g., timing of earliest vocalizations, entry into the air cell, pipping, hatch date, and chick ID) are printed on each strip. Egg locations are written on the strip following each egg move. Using the Egg Chronology Board, a caretaker can quickly determine location and stage in development of each egg.

JJ. Hatcher Board.

When late-term eggs are moved from nests or artificial incubators to the hatcher, the Egg Cards (A) are affixed beneath the label for the appropriate hatcher on this display board.

KK. Artificial Insemination (AI) Planning Scheme.

A detailed breeding strategy for each individual in the colony is condensed into this form (Fig. 10.13). Copies are included for easy reference in the annual field log for AI. This record provides a useful, brief description of the pair history and AI recommendations. This form is filed by taxon and year with the Sire/Dam AI Records (G).

LL. Oviposition and Incubation Chronology (Giant Check Sheet).

This large display form depicts the chronology of egg laying and egg moves for a breeding season.

Column headings are days beginning with the date of the first egg of the season. Rows are either dam ID or pen numbers. All females of one taxa are grouped together with the first female to lay occupying the most elevated row. Data is transcribed to this check sheet directly from the Breeding Pairs: Daily Check Sheet (Q). An underlined check mark indicates the day oviposition occurred. A check mark indicates the pair is incubating. Eggs removed or added are indicated by plus or minus signs and numbers. For example, a two egg clutch removed to encourage a pair to lay again would be indicated by a “-2” in lieu of the check for that day. This form simplifies predicting when a pair is due to recycle (re-lay). The form is also useful in planning egg moves and in recycling pairs so that the best foster parents are in the proper reproductive state to receive eggs or chicks of endangered taxa. It is helpful to have each pair’s incubation performance rating next to the dam ID or pen number. Hatch date and chick ID numbers are placed at the end of

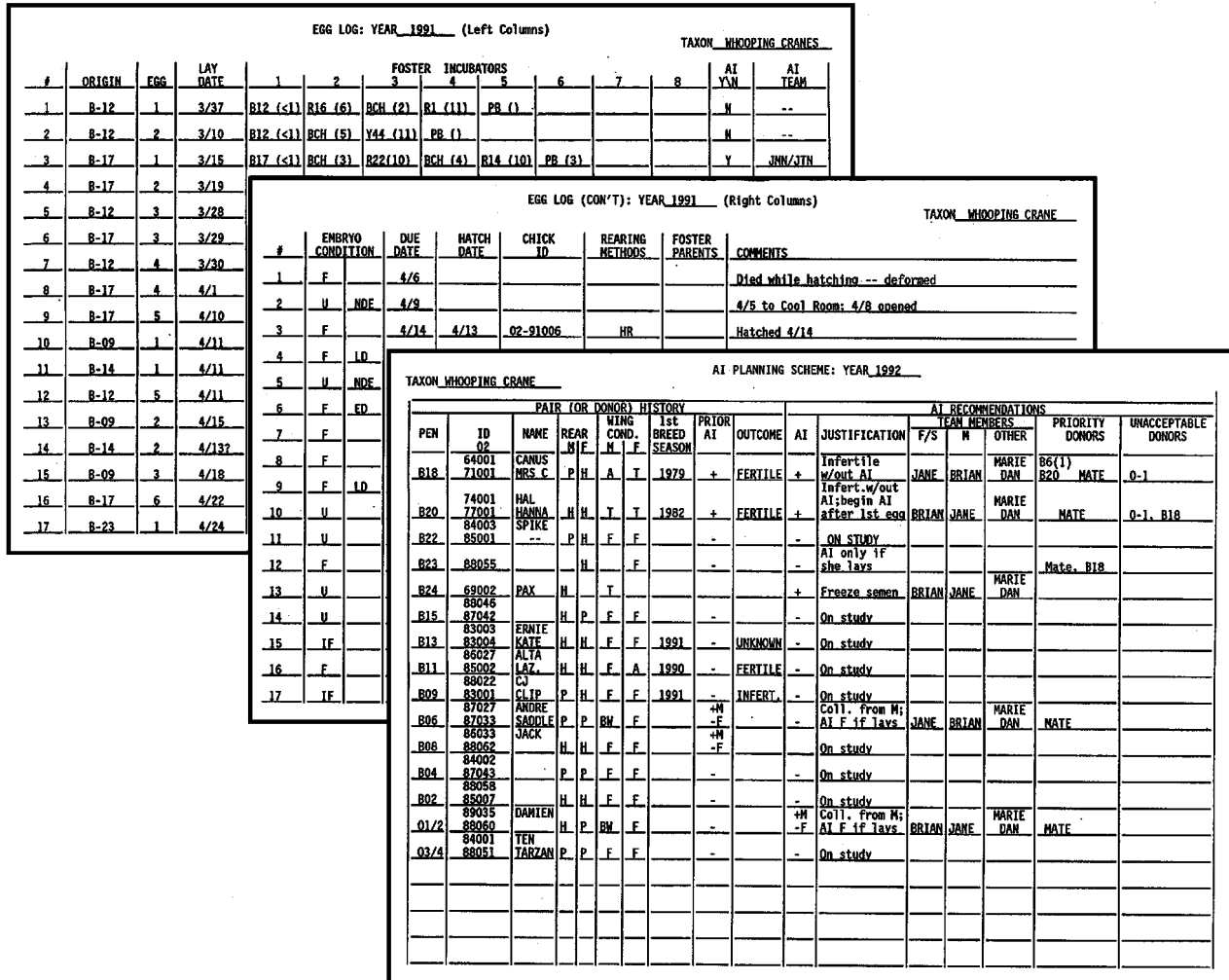


Fig. 10.13. Some planning records and visual aids, Forms HH and KK.

the row when the pair is no longer incubating. One form is generated each year and then filed chronologically.

MM. Egg and Chick Board. Using a ferric board and magnetic labels, this visual aid displays the number and taxon of eggs or chicks each pair is incubating or rearing. As for the Egg Card Board, each breeding pen is listed. Beneath each pen label, colored circles (eggs) and squares (chicks) indicate the appropriate number and taxa for eggs and chicks attended by each pair of foster parents. This visual aid is especially useful in planning egg and chick care during the period when many pairs of foster parents are in transition from incubation to chick rearing. By contrast, during incubation, the Oviposition and Incubation Chronology (LL) and the Egg Card Board (GG) are most useful.

NN. Patient Board. This display board is maintained at the hospital with a copy in the caretaker work area. It serves as a visual aid for use in caring for hospitalized cranes. On it, treatment instructions and other records pertinent to patient care are posted.

System Use

The 40 or so forms, files, notebooks, and display boards that comprise the Patuxent system provide a broad, detailed framework for the management of the most important data for a crane colony. Not all records need to be used by every institution. For example, if, at one institution, all eggs are mechanically incubated and all chicks are hand-reared, then

those records dealing with foster parent incubation (E, Q, LL, and MM), and foster parent rearing (C, F, P, and MM) are not used. If all propagation is through naturally fertile pairs, then all records dealing with artificial insemination (G, Z, and KK) can be eliminated.

The system can also be streamlined for use in very small crane colonies. Where few breeding pairs produce few eggs, many of the visual aids and production records become unnecessary. The most fundamental records are H, R, T, U, W, and AA. Little can be omitted from this nucleus of records without sacrificing the future usefulness of the colony. Because of the value of the records, we recommend that a duplicate copy be stored in a separate building.

Without actually using the system for a breeding season, it is difficult to understand the flow of information. Table 10.1, however, provides an overview of system use. Here, appropriate system responses are portrayed for some life history events of a foster parent-reared crane. When an egg hatches, for example, it is first recorded on the Breeding Pairs: Daily Check Sheet, then the Egg Card (A) is removed from the Egg Card Board (GG), and the hatching date and ID are placed on the Natality Sheet (O), Egg Card (A), Egg Chronology Board (II), Oviposition and Incubation Chronology (LL), Egg and Chick Board (MM), Dam Reproductive Record (H), and Egg Log (HH). A change is made on the chalk board, precursor of the Weekly Report (V), an entry is made in the Accession Book (T), and a Rearing Record (C) is created. Ultimately the Pair History Records (E and F), Specimen/Pen Inventory (U), Giant Table (W) and the Genealogical Summary (Y) must also be modified. All of these changes must be made by hand in a manual system. In a computer managed relational database, however, entry of the hatching event at one level will yield appropriate responses for all other records linked in the software matrix.

Patuxent's Automated Records System

At Patuxent, we have automated much of the records system presented here. We first experimented with the ISIS/ARKS program, but noted that at that time ARKS was unable to deal with the complex management of eggs. EGGS software, adapted from Patuxent's complex manual system, is being developed to operate with ARKS III. Another limitation of ARKS was that the details of life history events of the individual went into a general database named

"Special Data" with the result that no relational manipulation was possible. Fortunately, home grown programs can be incorporated with ARKS III to perform this function (L. Bingaman Lackey, Hendersonville, North Carolina, personal communication).

One final drawback of ARKS was that Patuxent's ID numbers were incompatible to use as accession numbers, so separate accession numbers had to be assigned. ICF and most institutions have been able to use their ID numbers as the ARKS accession numbers. Because of the early inflexibility and limitations of the ISIS/ARKS system (now somewhat alleviated by the availability of MedARKS, SPARKS, and EGGS), Patuxent redesigned the manual system (Ellis et al. 1991) and began automation. Today about one-third of the IC's in the Patuxent system have been automated. There is no need to automate visual aids and many records in Figure 10.1. The records that should be automated are: Egg Card (A), Pair History: Incubation and Rearing (E,F), Sire/Dam Reproductive Record (H), Individual Record (L), Individual Log (R), Natality Sheet (O), Accession Book (T), Specimen/Pen Inventory (U), Weekly Report (V), Annual Production Summary (X), Genealogical Summary (Y) Mortality Summary (AA), Shipment Report (CC), Egg Laying Interval Record (FF), and Egg Log (HH). The following reports can be generated from retrieving data from other records and without additional data entry: Natality Sheet (O), Accession Book (T), Species/Pen Inventory (U), Weekly Report (V), Giant Table (W), Annual Production Summary (X), Shipment Report (CC), Egg Laying Interval Record (FF), Pair History: Incubation (E), Pair History: Rearing (G), Individual Record (L), Genealogical Summary (V), Mortality Summary (AA), and Egg Log (HH).

Record Keeping

The Patuxent system consists of a complex array of records linked through limited redundancy to provide for the systematic retention and collation of the data of interest in managing a large crane collection. The manual and automated systems are so closely linked that it is possible to use all or part of either without inordinate duplication of effort. The automation process is continuing. Hard copies of the individual form sheets are available from Patuxent.

TABLE 10.1

Immediate Responses to Some Natural History Events for a Parent-reared Crane¹

	OVIPOSITION	HATCHING	SICKNESS BEFORE FLEDGING	DEATH BEFORE FLEDGING	POST FLEDGING PEN MOVES	PAIRING	POST FLEDGING SICKNESS	POST FLEDGING DEATH
A Egg Card	5	3						
C Parent-reared Chick: Daily Log	+	2	3					
D Pair History: Behavior					4			
G Sire/Dam AI Record	+							
H Sire/Dam Reproductive Record	+	+	+					
I Necropsy Record			+					+
J Medical Record			+				+	
L Individual Record					4	3		+
M Daily Log					1	1	1	1
O Nality Sheet		2						
P Parent-reared Chicks: Daily Check Sheet		1	1	2				
Q Breeding Pairs Daily Check Sheet	1	1		1				
R Individual Log					3	2	+	+
S Veterinary Logs			+	+			+	+
T Accession Book		+		+				+
U Specimen/Pen Inventory				+	2			+
V Weekly Report	+	+		+				+
AA Mortality Summary				+				+
HH Egg Log	+	+		+				
II Egg Chronology Board	3	4						
LL Oviposition and Incubation Chronology	2	5		4				
MM Egg and Chick Board	4	6		5				
NN Patient Board								+

¹ Numbers (e.g., 1, 2, 3, ...) indicate order of responses. A plus (+) indicates that responses can be in any order.

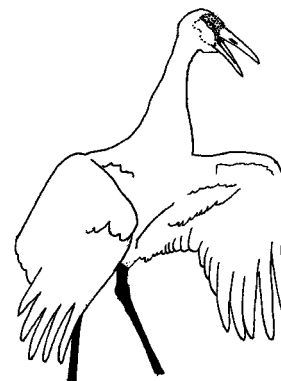
ICF's system is designed for relative simplicity and ability to integrate information with other institutions through ISIS/ARKS. Centralized individual files contain essentially all of an individual's records. ICF has developed a simple, computerized-text system for recording behavioral events. Important behavioral and reproductive milestones for the individual are also recorded in ARKS III. MedARKS is used for medical records, and copies are placed in the individual files. Annual summaries are produced and used for breeding objectives and reports.

Either system can be pared down for use with smaller collections or can be modified for use with other taxa. The use of such systems is essential to the long-term management of any crane colony intended for propagation or scientific research.

One final recommendation is of great importance. Although all animal care staff take and record information, each institution should designate one or two Record Keepers (or Registrars) who closely manage the entry of data into the automated system. At ICF, separate individuals handle ARKS and MedARKS records. Each Record Keeper should be fully familiar with the system, participate regularly in data entry, and serve as backup replacement in the event of loss of the other Record Keeper. However, a chief Record Keeper should oversee and control data entry to minimize errors in the data entry.

Literature Cited

- Archibald, G. W. 1976. The unison call of cranes as a useful taxonomic tool. Ph.D. dissertation, Cornell University, Ithaca, N. Y. 167 pp.
- Brown, R. A. 1975. A new records system for the New York Zoological Park. *International Zoo Yearbook* 15:229-235.
- Carpenter, J. W. 1977. Propagation and management of endangered species at the Patuxent Wildlife Research Center. Pages 23-33 *in* American Association of Zoo Veterinarians Annual Proceedings, Honolulu, Haw.
- Carpenter, J. W. 1986. Cranes (Order Gruiformes). Pages 316-326 *in* M. E. Fowler, editor. *Zoo and wild animal medicine*. W. B. Saunders Company, Philadelphia.
- Castleberry, M. W., E. D. Jenkins, and S. W. Thompson. 1966. Use of the electronic computer in retrieval of veterinary pathological data. *American Journal of Veterinary Research* 27(118):824-829.
- Committee on Laboratory Animal Records. 1979. Laboratory animal records. National Institutes of Health Administrative Document, Contract No. NO1-RR-5-2128, Washington, D.C. 67 pp.
- Ellis, D. H., S. R. Swengel, G. W. Archibald, and C. B. Kepler. 1996. A sociogram for the cranes of the world. *Under review*.
- Ellis, D. H., L. Hartman, S. G. Hereford, and J. L. McMillen. 1991. A records system for a captive crane flock. Pages 433-445 *in* J. Harris, editor. *Proceedings 1987 International Crane Workshop*. International Crane Foundation, Baraboo, Wis.
- Gee, G. F. 1983. Avian artificial insemination and semen preservation. Pages 375-398 *in* A. C. Risser, Jr. and F. S. Todd, editors. *Proceedings of the 1983 Jean Delacour/IFCB Symposium on Breeding Birds in Captivity*, Los Angeles, California. International Foundation for the Conservation of Birds, North Hollywood, Calif.
- Hartman, L., S. Duncan, and G. Archibald. 1987. The hatching process in cranes with recommendations for assisting abnormal chicks. Pages 387-397 *in* J. C. Lewis, editor. *Proceedings 1985 Crane Workshop*. Platte River Whooping Crane Habitat Maintenance Trust and U.S. Fish and Wildlife Service, Grand Island, Nebr.
- Sciabarrasi, S. A., and W. T. London. 1974. A computerized primate information retrieval system (PIRS). *Laboratory Animal Science* 24:773-782.
- Seal, U. S., and D. G. Makey. 1975. Computer usage for total animal and endangered species inventory systems: a specific proposal. Pages 178-188 *in* *Research in zoos and aquariums*. National Academy of Sciences, Washington, D.C.
- Seal, U. S., D. G. Makey, and L. E. Murtfeldt. 1976. ISIS: an animal census system. *International Zoo Yearbook* 16:180-184.
- Seal, U. S., D. G. Makey, D. Bridgwater, L. Simmons, and L. Murtfeldt. 1977. ISIS: a computerized record system for the management of wild animals in captivity. *International Zoo Yearbook* 17:68-70.
- Sheppard, C. 1985. Genealogy of the white-naped crane (*Grus vipio*). New York Zoological Society. 50 pp.
- Systematized Nomenclature of Medicine (SNOMED). 1976, 1977. Introduction, 22 pp., 1977; Disease, 137 pp., 1976; Etiology, 117 pp., 1976; Function, 155 pp., 1976; Morphology, 171 pp., 1976; Procedure, 141 pp., 1976; Topography, 199 pp., 1976. College of American Pathologists, Chicago, Ill.



Special Techniques, Part A: Crane Artificial Insemination



George F. Gee and Claire M. Mirande

Captive breeding of nondomestic birds has increased dramatically since 1950, and captive crane production often exceeds that of wild birds in their native habitat. Artificial insemination (AI) is one propagation technique (Gee and Temple 1978; Gee 1983) used extensively with cranes. With proper conditioning and management of the cranes, AI often produces fertility better than that achieved through natural matings.

The most obvious need for an AI program is to reduce **infertility** (Archibald 1974; Gee and Temple 1978; Lake 1978; Sexton 1979). In some mated pairs natural copulation can be difficult because of injury (including wing impairment to deter flight), deformity, differences in body size, or behavioral difficulties. Sometimes females are kept in separate pens because of mate aggression, pair incompatibility, or the lack of a mate. Occasionally, a productive female may be in a distant location separate from the male, where transfer of semen is the only way to avoid infertility. Fertility in a mated pair can be improved by AI using the same or a different male.

AI is useful in carrying out the goals of **special breeding** programs for captive propagation. The genetic influence of one male in a population can be increased by using his semen to sire young from several females each season. Conversely, semen from several males can be used to increase female fertility. (Such practices can, of course, lead to questions of paternity; techniques for resolving paternity are still expensive and results are dependent upon the availability of suitable genetic markers.) **Hybridization** between behaviorally incompatible species is possible with AI, and although it should be avoided for propagation purposes, some research questions require hybridization. Patuxent used Whooping Crane semen to produce four Whooper-Sandhill Cranes to study hybrid characteristics.

AI has other **special uses**. For example, a male's potential for producing progeny with specific traits (chicks that grow more rapidly or possess superior disease resistance) or his potential fertility can be

determined more quickly through AI than with natural matings. The collection of semen provides for other uses including laboratory studies evaluating reproductive potential (Sharlin et al. 1979), evaluating semen diluents (Sexton 1977), detecting disease (Thurston et al. 1975; Stipkovits et al. 1978; Ferrier et al. 1982), and separating species and subspecies through hybridization and sperm morphology (Sharlin et al. 1979; Russman and Harrison 1982).

Male Reproduction

Reproductive Anatomy

Crane anatomy (Fig. 7.2) is very different from mammal reproductive systems. For greater detail, also read Chapter 7. The paired testes lie deep in the body cavity of the crane, above the abdominal air sacs and below the anterior lobe of the kidneys. The vas deferens conduct the sperm from the testis to the cloaca. The vas deferens end in erectile papillae in the urodeum, the central chamber of the cloaca. Semen contains: (1) fluids secreted from the seminiferous tubules, (2) epithelial cells of the reproductive tract, (3) lymph from the lymph folds and erectile tissues in the cloaca, and (4) sperm (Mann 1964; Lake 1966; Buxton and Orcutt 1975; Nishiyama et al. 1976; Servouse et al. 1976; Burt and Chalovich 1978; Gasparska et al. 1981).

Semen Collection

Physiologists classify semen collection techniques according to levels of cooperation: **cooperative**, **massage**, and **electroejaculation**. Massage AI is more successful with cooperation, although stressed or aggressive cranes have also been stimulated to respond. Electroejaculation is successful with or without active cooperation from the bird.

Cooperative semen collection and insemination, pioneered with **sexually imprinted birds** of prey (Hamerstrom 1970; Temple 1972; Berry 1972; Grier 1973), requires careful timing to get an adequate number of samples and to get fertile eggs. Methods that intercept semen during natural copulation with other birds or dummy mounting devices are variations of the cooperative collection technique (Smyth 1968; Tan 1980).

The **massage** collection technique (Quinn and Burrows 1936) has been used for decades with domestic poultry and more recently with cranes (Archibald 1974; Gee and Temple 1978; Gee 1983). For this technique, the bird is restrained by an assistant while an operator collects the semen. This technique was first applied to cranes in 1969. The **basic technique** was altered to allow for the crane's long legs, sharp talons, and long beaks. The process generally takes 5 to 10 sec.

The following description will serve as a guide, but must be tailored to the unique behavioral and physical characteristics of each bird. It is important to reduce stress on the individual bird by capturing the desired bird quickly. It is also important for the AI team to attempt to stimulate cooperation when stroking.

An **assistant**, with help from two other members of the team, **captures** the male (if necessary guides him into the nearest corner) and holds him. Team capture reduces chase time and its resultant risk of injury. Adults which come to the gate and attack can often be safely and quickly captured by one person with less stress than if the bird flees. The assistant **cradles the bird** between the legs (Fig. 11A.1) with the bird's head protruding behind the assistant's back. The assistant



Fig. 11A.1. *Whooping Crane AI: Brian Clauss injects sample as Jane Nicolich massages.*

Photo David H. Ellis

rhythmically massages the bird's legs by grasping the shanks and stroking gently several times in a circular, inward and downward direction. The speed and pressure should be varied to coordinate with the calling and other responses from the bird.

The second person, the **operator**, kneeling behind the bird and facing the massage person, **strokes** the bird's back with the heel of the open hand toward the head and fingers directed toward the vent. Several strokes are given; each passes along the lower back to the base of the tail. With the other hand, another series of strokes passes from mid abdomen to vent. Both hands reach the tail region at the same time. The bird may respond to this stimulation by pushing forward against the assistant's thighs, emitting a low vocal growl or purr, and raising its tail. The operator then **pushes upon the tail** with the heel of the left hand (if right handed) and strokes the abdominal region with the right hand. Many birds respond to this stimulation by opening or even everting their cloacas. Next, the **cloaca is grasped** dorsally by the thumb and index finger and the semen is expressed.

The operator or a third person holds a small glass collection device (4-5 cm in diameter) in the right hand (if the person is right handed) for semen collection (Fig. 11A.2). Often, a spontaneous ejaculation occurs expelling the first drop of semen onto **the lip** of the glass (a sealed funnel or "shot glass"). Occasionally, the bird lifts its legs off the ground when it ejaculates and must be supported by the assistant's forearms. If the semen sample was small, sometimes the dorsal wall of the vent is again massaged to gently express the



Fig. 11A.2. *Collecting semen in a funnel. (Note protruding urodeum.)*

Photo David H. Ellis

remainder of the semen which is then scooped onto the lip of the glass. The entire process is normally finished in 5-10 sec. However, some Siberian Cranes require stroking for 2-3 min before obtaining a sample, and some individuals of each species require similarly long massage periods.

Special Semen Collection Tips

Semen collection is something of an art. To avoid excessive stress, perform the task quickly, be relaxed, and talk only to facilitate successful coordination and collection. Semen collection techniques must be modified to each crane's unique anatomical, physiological, or behavioral characteristics. Birds respond differently to individual people. Note responses to the assistant and operator and assign people accordingly.

Semen volume and sperm concentration vary greatly between birds (see Chapter 3), and some cranes produce samples too small for insemination. However, with special care in collection, as little as 0.01 cc of semen can be diluted, drawn up into a 1-cc (TB) syringe and inseminated directly into the female. At ICF and Patuxent, significant numbers of eggs have been fertilized by 0.01-0.02 cc of semen. When semen samples are this small, it is helpful to extend the sample such that the total volume at insemination is approximately 0.1 cc. About 16 million sperm should be provided for an effective insemination (Gee and Sexton 1979).

Small samples can be aspirated into a microliter pipette or collected on the edge of a slide (Howell and Bartholomew 1952; Smyth 1968; Lake 1978; Gee and Sexton 1990). Because small samples dehydrate rapidly, protect them immediately by adding a drop of diluent. Although ejaculates produced by some males contain insufficient sperm to fertilize an egg, a pooled sample from several small collections have been used successfully (McDaniel and Sexton 1977; Gee and Temple 1978). Patuxent has collected semen from Sandhill Cranes daily, Monday through Friday, for extended periods, but we now try to **restrict collections to two to three times per week**. Extended periods of daily collections or rough treatment may lead to cloacal tissue damage, stress, and reduced cooperation.

Blood in a semen sample may indicate cloacal injury. Occasionally, the damage is only a superficial scratch on the vent. Avoid collecting semen from a bird with an injury for 7-10 days while the vent heals.

Fecal contamination of semen is common, but can often be reduced by conditioning the birds to a schedule. Although contaminated semen is normally discarded, egg fertilization is still possible if the semen is cleaned of the coarse contaminants and used for insemination immediately. To clean a sample, let the contaminants settle, then draw the semen from the top or side of the sample with the syringe. When using contaminated semen, deposit the sample in the cloaca not the vagina to avoid infections of the oviduct (Perek et al. 1969).

Female Reproduction

Reproductive Anatomy

The one functional ovary lies deep in the body cavity above the abdominal air sacs and below the anterior lobe of the left kidney. The oviduct carries the ovum through the infundibulum, magnum, isthmus, uterus, and vagina (see Fig. 7.3) to the vent in 3 to 4 days (M. Putnam, University of Wisconsin, Madison, personal communication). The infundibular region receives the egg from the ovary and is the site of fertilization (Olsen 1942). The vagina is the passageway for the egg from the uterus to the cloaca and for the semen into the oviduct (Sturkie 1965). Sperm storage sites (sperm host glands) are present in the infundibulum and the uterovaginal (UV) juncture (Bobr et al. 1962). The UV-sperm host glands enable birds to lay several fertile eggs following a single copulation (Smyth 1968).

Investigators have identified **sperm host glands** in domestic and nondomestic birds including cranes (B. C. Wentworth, University of Wisconsin, Madison, personal communication). They may be common to all birds. The host glands are thought to release sperm on a continuous basis (Compton et al. 1977, 1978; Compton and Van Krey 1979a, 1979b; Bakst 1980, 1981). However, some believe the release of spermatozoa is greatest at ovulation or oviposition. Bakst (1980) reported fewer sperm in the oviduct following the passage of an egg suggesting that sperm are sequestered or removed during egg formation.

Near laying time, the ends of the pubic bones become more pliable and spread apart, and the cloacal tissues enlarge and soften. The dorsal lip of the vent expands more than the ventral lip and creates the appearance of an inverted smile. It is possible to

predict when the bird will lay by the size of the enlarging vent and spread of the pubic bones. A history for each bird from previous years helps because some birds expand more than others. We measure the spread at the ends of the pubic bones by passing the fingers between the pubic bones while stroking the abdomen. Hold the palm of the hand against the abdomen and stroke from the abdomen to the base of the tail. In most cranes in winter, the distance between the ends of the pubic bones is less than one finger width. This distance is two or more fingers wide just prior to laying. By palpating pubic spread you can forecast egg laying and choose the best time to inseminate.

Insemination

At Patuxent, female cranes are massaged just as for the males. In the ICF method, the female's back and sides (posterior to the wings) are stroked to simulate the male's abdomen on the female's back during copulation. Semen can be deposited into the cloaca or vagina.

With effective stimulation, the crane opens the cloaca. The vagina, the distal end of the oviduct, appears as a red rosette on the left wall of the urodeum. To expose the vagina, push aside the dorsal wall of the vent separating the urodeum from the proctodeum with the syringe or other device. To avoid injury to the soft cloacal and oviductal tissues, the probing or inseminating device should be smooth, without abrasive edges. With practice the inseminating device can be inserted into the vagina (Fig. 11A.3) during the few seconds it is visible. If the vagina cannot be seen, gently probe with the end of the syringe



Fig. 11A.3. *Injecting semen into female with syringe.*

Photo David H. Ellis

on the left side of the urodeum. Next, allow the inserted syringe to drop to a relaxed position. When the cloaca contracts around the syringe, stop stroking and gently push the plunger to deposit the semen. Although the female is still being supported, she will frequently relax when stroking stops. Resume stroking gently if she starts to struggle.

In properly trained cranes, much of the manipulation of the cloaca can be avoided. At ICF, 12 species of cranes have been successfully inseminated after they assumed copulation posture and everted their oviducts in response to massage stimulation and handling. It is even possible to deposit semen in the oviduct of uncooperative cranes. If you are unable to locate the oviduct by palpation, the distal end of the oviduct can often be everted by placing firm pressure on the female's abdomen and the walls of the cloaca. We do not recommend that an inexperienced person evert the cloaca because force can cause injury and undue stress to the bird.

Placing semen directly in the oviduct has been the preferred insemination technique with most nondomestic birds (Smyth 1968; Bird et al. 1976; Boyd et al. 1977), although satisfactory results derive from simply depositing semen in the cloaca (Gee 1969 unpubl.; Temple 1972; Berry 1972; Grier 1973; Archibald 1974). In the Sandhill Crane, fertility rates above 80% were achieved with cloacal insemination. In this program, insemination was begun two to three weeks before the first egg was laid and continued throughout the season with at least two inseminations (each containing about 16×10^6 sperm) each week and within a few hours after each oviposition (Gee and Sexton 1979; Gee et al. 1985).

However, with the same insemination schedule, we achieved **better fertility** when semen was placed in the vagina. Deep vaginal insemination is preferred for other species (Lorenz 1969; Ogasawara and Fuqua 1972) because the storage site (sperm host glands) is in the utero-vaginal juncture (Bobr et al. 1962). Moderate depth vaginal inseminations also give satisfactory results (Smyth 1968; Wentworth et al. 1975; Bird et al. 1976; Boyd et al. 1977) and reduce the possibility of injury (Ogasawara and Fuqua 1972; Wentworth et al. 1975) that can result from forcing the inseminating device to the utero-vaginal juncture. When semen is deposited in the cloaca instead of the oviduct, inseminations should be more frequent and timed to follow oviposition (Gee 1969 unpubl.; Temple 1972; Berry 1972; Grier 1973; Archibald 1974; Gee and Temple 1978).

The cloaca should not contain **feces** when inseminating. When the cloaca is full, the bird will defecate soon after insemination and fecal bacteria can kill large numbers of sperm and reduce fertility. For some females, it may be necessary to withhold feed and water for 6 to 8 hours before AI (Smyth 1968), but normally, herding the bird (male or female) around the pen for a few seconds before capture will induce defecation.

The **volume of semen** that is required to produce a fertile egg depends upon the sperm concentration and the capacity of the reproductive tract to retain the semen. Often, a single semen sample is adequate to inseminate two or more females with the result that a sample may be diluted. However, if the volume of diluted semen exceeds the capacity of the bird's vagina, fertility rates can be reduced because some sperm are, as a result, expelled from the lumen of the vagina. More frequent inseminations are advisable when the number of sperm per insemination is low (Meyer et al. 1980). For example, a single Sandhill Crane ejaculate (200-300 million sperm/mL, 0.05 mL/ejaculate; Gee and Temple 1978) may not contain enough sperm to produce a satisfactory fertility rate. Based on more than 20 years of experience with cranes, we **recommend repeated insemination** every other day for two weeks before the crane lays her first egg, 2-3 times each week after that, and within 1-2 h after every oviposition to get the best fertility.

To get the highest fertility rates, we generally inseminate the entire ejaculate after diluting it 1:1 with a poultry semen extender or an extender modified for use in cranes (Gee et al. 1985). The dilution provides greater volume, thereby reducing the loss of sperm on the sides of collection, handling, and inseminating devices. Most inseminations contain 0.05 to 0.15 mL of good quality semen (see the Semen Protection and Evaluation section that follows). If we determine that a poor semen sample was used, we often return with another sample later in the same day.

Fertility Management

AI is only one of several methods used to correct infertility. AI of cranes is labor-intensive. Because natural copulation in properly mated cranes generally results in fertility equal to that in artificially inseminated birds (Gee 1969 unpubl.), a **change of mates** may prove sufficient to raise fertility and may be more cost effective than AI. Most flighted birds will breed naturally if properly reared with their own species and

housed in large, net-covered pens (Ellis et al. 1991). For flight restricted birds, the number of males successfully copulating and the fertility rate are lower than for full-winged birds (Swengel and Archibald 1988; Ellis et al. 1991; Belterman and King 1993).

Other conditions that promote reproduction are treated in Chapters 6 and 7. Of the environmental conditions, light, temperature, and humidity are the three most important. In most cranes, semen production begins before, and continues until after, the end of egg production (see Chapter 3). However, the asynchronous production of semen and eggs does occur in cranes.

AI Training

When circumstances recommend AI, the **cranes should be trained** to accept the technique. Behavioral accommodations are of great importance in artificial insemination of cranes, especially those taken from the wild. **Stress** is difficult to control in crane AI, but can be reduced by using the same team, training birds to accept the procedures, and avoiding injury. Occasionally, the **training process** upsets rather than calms the bird and if continued, may interfere with the onset of egg production. In these cases, stop the training. Reinstate insemination only after the first egg is laid whereupon the bird may be more receptive. Another manipulation that may improve AI response is to place the bird or pair near other reproductively active birds. Visual and auditory displays by neighboring cranes sometimes stimulates reproductive activity (even in single birds).

Semen Protection and Evaluation

The following steps can be taken to **protect the semen** and to use it most effectively. Semen should not be exposed to direct sunlight. Store samples until use in a water bath or insulated container (ca 5° C) to reduce temperature fluctuations. A closed tube reduces dehydration and contamination. A diluent increases semen volume, reduces the risk of dehydration, and if sperm concentration is adequate, makes it possible to inseminate several birds from each ejaculate. A **diluent** was

developed especially for cranes (Gee et al. 1985), but most commercial poultry semen extenders (see Appendix) are adequate. Diluent reduces sperm concentration, bacterial contamination, provides energy, and controls pH and osmolality. All the tubes and **inseminating devices** that contact the semen should be clean and free of detergents. All equipment and supplies should be thoroughly rinsed with clean water before use. For reviews of factors harmful to sperm survival, see Mann (1964), Lake and Steward (1978), Lake (1969), and Smyth (1968).

Although the most **reliable semen test** is the production of **fertile eggs**, semen for use in AI can be evaluated immediately upon collection and later in the laboratory (see below). The **color** of good crane semen ranges from clear to milky white. **Fecal contamination** discolors the semen to shades of brown or green. Occasionally, flecks of blood may be present resulting from excessive force during collection or injury (Smyth 1968). Samples consistently contaminated with feces may need to be diluted with antibiotics to reduce the loss of sperm. The antibiotic, tobramycin, may even increase fertility when used as a diluent in "clean" semen (Sexton et al. 1980). Good crane semen is only **slightly thicker than water**. Samples that appear to be sticky or stringy are often contaminated with urates. Sometimes semen samples begin as a clear fluid in the collecting device and turn white as the urates precipitate out. **Watery semen** may result from collecting too much lymph in the sample because excessive force was used on the cloaca during collection. These watery fluids, like fecal and urate contaminants, adversely affect spermatozoa, especially if you hold the semen for some time before insemination (Smyth 1968; Lake 1971; Fujihara and Nishiyama 1976).

In the laboratory, samples of a crane's semen are examined for sperm number, motility, and morphology (Fig. 11A.4). A more extensive examination (i.e., an evaluation of metabolic rate and semen chemical composition) may be needed in special cases. The simplest **measures of semen quality** are sperm number and motility. Gross testing of semen quality is discussed later in this chapter.

Sperm number can be estimated from a semen score for density, a spermocrit, or by counting in a hemocytometer or in an automated counter. Sperm concentration can be evaluated on a hanging drop slide, under a cover slip on a slide, or in a capillary tube (Putnam 1982). The scores can be calibrated by comparing them to actual sperm counts. The sperma-

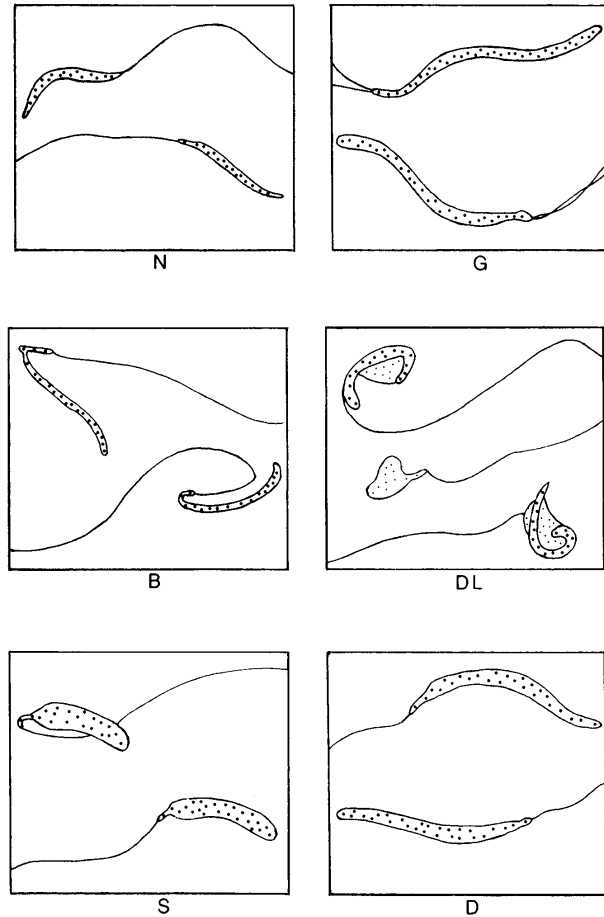


Fig. 11A.4. *Spermatozoa morphology, Greater Sandhill Crane: N, Normal; B, bent; S, swollen; G, giant; DL, droplet; D, dead. After Gee and Temple 1978.*

tocrit, a simple measure of sperm concentration, is useful in characterizing semen produced in quantity (>0.1 mL) and containing many sperm per mL ($>3 \times 10^9$) (Arscott and Kuhns 1969). The semen sample is loaded into the standard microhematocrit capillary and centrifuged. To count sperm, the semen is diluted (if necessary), fixed, and the sperm counted in a hemocytometer or in an automated counter (Jones and Wilson 1967). Optical density of a diluted semen sample can also be measured and sperm number determined from an established standard curve (Kosin and Wheeler 1956; Carson et al. 1955).

Sperm progressive motility, discussed in greater detail later, is an estimate of the percent of spermatozoa moving forward. Because some live cells are inactive, this measure is not an estimate of the percentage of live cells.

Sperm morphology can also provide information about the percentage of live cells in the semen as well as the frequency abnormalities and the size of cells.

One of the easiest determinations is a **live-dead count** from an eosin-nigrosin stained slide (Gee and Sexton 1979). Although this procedure is more time-consuming than progressive motility, it can be performed long after the insemination, usually without a loss in accuracy. However, excessive moisture in the atmosphere can make staining less definitive (Ogasawara et al. 1976).

Determine **abnormalities** from a variety of preparations including the eosin-nigrosin stained slide. Good slide staining techniques aid in delineating parts of the spermatozoa such as the head from the acrosomal cap and mid-piece (Sharlin et al. 1979; Russman and Harrison 1982). Abnormalities in sperm help in evaluating semen from the males and in determining effects of diluents and storage. Sperm head size as determined from properly stained slides can also help distinguish subspecies (Sharlin et al. 1979; Russman and Harrison 1982) and predict fecundity (Sharlin et al. 1979). Electron microscopy can also be useful in detecting membrane and fine structure abnormalities in spermatozoa.

Although laboratory tests are useful in evaluating semen, satisfactory fertility rates have come from semen that scored poorly in the laboratory, especially frozen-thawed semen (Sexton 1976). Cryoprotectants and freezing can affect sperm motility and morphology without destroying the ability of the frozen-thawed semen to produce fertile eggs.

Short-term Semen Storage

Although the best fertility rates come from using semen immediately following collection, crane semen can be stored for several hours without significantly reducing fertility. **Semen storage** for an hour or more calls for temperature control and protection from contamination and drying. Sperm of most species survive best at near freezing temperatures (Gee and Temple 1978; Sexton 1979). We use a wide-mouth thermos, ice water, and a submerged dry container for samples. Because bacterial contamination can rapidly destroy sperm cells, avoid contamination or use diluents to introduce antibacterial agents, stabilize pH and osmolality, and in other ways extend the life of a semen sample (Smyth 1968; Gee and Temple 1978; Sexton et al. 1980). Semen pH and osmolality vary from species to species. Semen pH ranges from 6.0 for a duck to 8.0 for a crane.

Any of the **semen extenders** used for domestic poultry are adequate for short-term storage (Lake and Steward 1978, Ogasawara and Ernst 1970, Sexton 1977, 1978). The Beltsville Poultry Semen Extender has been adapted for dilution of Sandhill and Whooping Crane semen (Gee et al. 1985, see Table 11A.1) by raising the pH to 7.8. You can use this extender for fresh storage and for freezing (see Chapter 11B). Semen from each crane species has a characteristic pH and osmolality (Gale 1987). Extender pH and osmolality should be tailored to the species for long term storage (fresh or frozen). By adapting **sperm preservation** techniques to a species, semen can be kept in the frozen state indefinitely (Sexton and Gee 1978; Watanabe and Terada 1980; Watanabe et al. 1981).

TABLE 11A.1

Crane Semen Extender

1000 mL distilled water
5.0 g D-fructose (MW = 180.16)
0.34 g magnesium chloride (MgCl ₂) (FW = 203.32)
0.65 g potassium phosphate (monobasic) (FW = 136.1)
12.7 g potassium phosphate (dibasic) (FW = 228.2)
0.64 g citric acid (MW = 306.4)
1.95 g N-[2-hydroxyl-1,1-bis(hydroxy-methyl)ethyl] taurine (MW = 229.25)
8.67 g L-glutamic acid (anhydrous) (MW = 169.1)
4.26 g sodium acetate (MW = 136.085)
Adjust pH to 7.8 with sodium hydroxide. Adjust to 310 mosM with distilled water.

Equipment, Facilities, and Supplies

The **basic equipment** used in AI is simple and inexpensive (Corten 1973, see Appendix). The record book is most important (see Chapter 10) and must be kept for comparisons during the year and from year to year. Devices used to collect semen include glass or plastic cups, "shot glasses," sealed funnels, syringes, test tubes, capillary tubes, and pipettes (Smyth 1968). A thermometer is needed for the semen storage case. Inseminating equipment includes syringes, needleless syringe caps, pipettes, straws, or eye droppers and

devices to hold or add diluents. Chaps should be worn by the massage person to help prevent injury from the crane's bill and talons. Goggles should be worn by all members of the AI team. In the AI kit (see Table 11A.2), put the AI supplies on one side of the kit and provide a separate area for first aid supplies to treat minor abrasions.

Crane facilities should be free from obstructions to reduce the chances of injury and to facilitate a quick, less stressful capture. A cloth or tennis netted corner (capture corner) in the pen reduces abrasion to the crane's wings during handling for AI. Also, clean facilities reduce the risk of semen contamination and soiling of the bird during capture.

Laboratory equipment, including a microscope, is needed for the routine examination of semen. Progressive motility estimates require only a clean slide and cover slip, but a hanging drop slide is useful for lengthy microscopic studies of living samples. It is desirable, but not necessary, to do additional evaluations of semen. A variety of stains are required if you are to do live-dead counts or special morphological studies. A balance with 0.01 g accuracy is needed to weigh out chemicals and to prepare stains and other supplies. Other supporting pieces of equipment and supplies include cell counters, slide trays, and photographic attachments. Also needed are pH paper or a pH meter, and to determine osmolality from small samples, a vapor point osmometer. Respirometers or spectrophotometers are needed to test semen metabolic activity.

TABLE 11A.2

AI Kit

AI funnels

1 cc syringes

semen extender

square gauze pads

roll gauze

adhesive tape

vet wrap

critoseal

small labels

capillary tubes

ruler

pencils

"sharpie" (indelible) marker

AI Program: Preparations for the AI Season

AI is labor intensive and calls for **planned and coordinated activities**. Form your **AI crew** a few weeks before the AI season to establish work schedules and to train personnel. Because birds respond to different people in different ways, choose people based on the bird's response to them. To avoid disturbance, maintain an established routine (same sequence each day, same time, with the same people) and move quietly with the least disturbance to the birds.

Write a short **reproductive history** for each bird that includes typical behavior, health, lay dates, semen characteristics, and fertility with AI. Also, prepare the proper **records** (AI book, species egg logs, dam reproductive records, and egg cards) before the season starts (see Chapter 10).

The AI Routine

Gradually introduce each pair to the collection and insemination routine. For the first two days (i.e., Monday and Wednesday), acclimate the birds to the crew by merely capturing, handling, and releasing them. On the third day (Friday), start stimulation and try to collect semen samples on every AI day thereafter. Begin insemination when the female is ready (i.e., responsive to handling, shows a widening in the distance between pubic bones, exhibits cloacal expansion, begins nest building, performs certain social displays such as Bill-down and Bill-down-growl, and becomes more aggressive). In a few birds the process may take longer. Do not proceed if the bird does not respond to stimulation. Use the crane's reproductive history as a guide and try to complete three inseminations before the first egg.

Males should be **rated** on a scale of 0 to 4 for their response to AI. We use the following score:

- 0 = No positive response to AI. Bird struggles and shows no sign of stimulation.
- 1 = Bird relaxes briefly but struggles most of the time.
- 2 = Neutral to slightly positive response. Doesn't struggle. Raises tail.
- 3 = Bird is relaxed. Raises tail. Everts cloaca.
- 4 = Responds strongly by raising tail, everting cloaca, vocalizes during massage. May climax.

The **female's** response to massage is **scored** as for the male. It is very important to measure the **pubic spread** prior to massage and insemination; otherwise semen could be expelled from the cloaca while taking the measurements. To measure the pubic spread, stroke the base of the tail with one hand, and with the other hand massage the lower abdomen below the vent in an upward motion. The spread of the pubic bones and the size of the cloaca is measured by finger widths. A pubic score of 2 means two fingers can fit between the pubic bones. Convert finger widths to mm after leaving the pen. A few days before laying, the pubic bones spread significantly (e.g., a female may change from 2 to 3.5 fingers) and the lips of the vent enlarge. Use the bird's previous records as a guide to condition and to help forecast egg laying.

Each AI day fill an adequate number of **syringes** before starting AI with 0.05 mL extender (or the average semen volume collected from your birds). Fill a few syringes with 0.02 mL extender to be ready for the few small ejaculates collected. In an alternative method, collect these semen, add an equal amount (a drop or two) of extender, draw the diluted semen into the insemination syringe, and then proceed with the insemination.

To **evaluate** a sample of **semen**, draw a small part of the semen (about 5 μ L) from the collecting funnel into a microcapillary tube (tube). Move the sample away from the tip of the tube by gently tapping the far end, then seal both ends with a putty ("Critoseal" or equivalent). Label the sample with pen number and place it in a small thermos (filled with ice and maintained between 0° and 5° C). Draw the rest of the semen into a 1 cc tuberculin syringe (Fig. 11A.5) already loaded with an equal volume of semen extender for intravaginal insemination. If the sample is very small, add enough extender to bring the volume up to 0.05–0.1 cc. An alternate method used by ICF is to use a microscope slide to examine the sample remaining in the syringe after insemination.

When semen is too contaminated to use, or the sample is too small, inseminate the female later in the day or the next day with the same donor. If paternity is of concern, it is better not to inseminate than to use another donor.

If a **contaminated semen sample** is to be used, draw the clean portion into the syringe, but use it only for cloacal, not vaginal, insemination. Large semen samples (0.08 mL and above) with good sperm concentration can be split and used to inseminate two females. Do not inseminate when a hard shelled egg is present in the oviduct.



Fig. 11A.5. Semen is drawn from the collecting funnel into a syringe.

Photo David H. Ellis.

Once you collect a female's first egg of the season, visually **check for eggs** at least twice daily, 0800 and 1600 h. During the laying season, check for eggs at 0700, 1200, and 1600 h, and during peak season, add another check later in the evening. When you expect a crane to lay an egg (calculated or felt in abdomen), make checks of that pen every few hours, so you can inseminate immediately after laying. **Insemination** soon after oviposition produces higher fertility of the next eggs. Schedule an AI team on holidays and odd hours to enable timely response. If the birds are multiple clutched (see Chapter 3), the interval between eggs increases, and checks twice each day are generally adequate.

Reproduction stresses birds, and AI compounds the problem. Some medical practices can help. Keep your breeding birds healthy. Put **padded protectors** on the leading edge of the wrist (see Chapter 8) for birds that have a tendency to scrape their wrists on the fence during capture. Report sick or injured birds to the

veterinary staff for evaluation and treatment. During handling for AI, inspect the cloaca for soreness, inflammation, and infection. Stop AI for a week or more when such conditions appear. When a sick bird has been handled, avoid spreading the infection to other birds.

Details of Semen Evaluation

At the lab, **examine the small semen** samples collected in capillary tubes or from the tips of the labeled TB syringes used for insemination earlier in the day. Remove the tube or syringe from the thermos and score the sample within two minutes after placing it under the 10x objective of the microscope. For a more detailed view, the contents from the tip of the syringe can be plunged onto a slide with a coverslip and viewed in the microscope field at 430x. Focus up and down at a point near the meniscus to find the sperm.

First, estimate **progressive motility** (percent of spermatozoa moving forward) and record in AI book. Score as follows:

- 0 = no motile sperm
- 1 = less than 25% motile
- 2 = 25% to 49% motile
- 3 = 50% to 74% motile
- 4 = more than 75% motile

Second, estimate **sperm concentration** (Fig. 11A.6) on a score of 0 to 4 and record in AI book. Score as follows:

- 0 = no sperm
- 1 = few sperm with large empty spaces
- 2 = many sperm with moderate spacing between them
- 3 = sperm numerous with little space between them
- 4 = packed sperm, hard to detect single sperm

Use the photographs of typical concentration scores (Fig. 11A.6) and display them above the microscope for comparison to help standardize scores between individuals and across years. In ICF's method, remember to **multiply** the sperm concentration seen in your field of view by your dilution factor. For example, if you extended the sperm sample 1:1, thus creating twice as much volume, you would multiply the sperm concentration by 2.

For males producing semen for the first time, in addition to the routine semen scores, make three complete **laboratory examinations** (early, mid, and late in the season) from an eosin-nigrosin stained slide. To prepare an **eosin-nigrosin (live-dead) slide**, place one

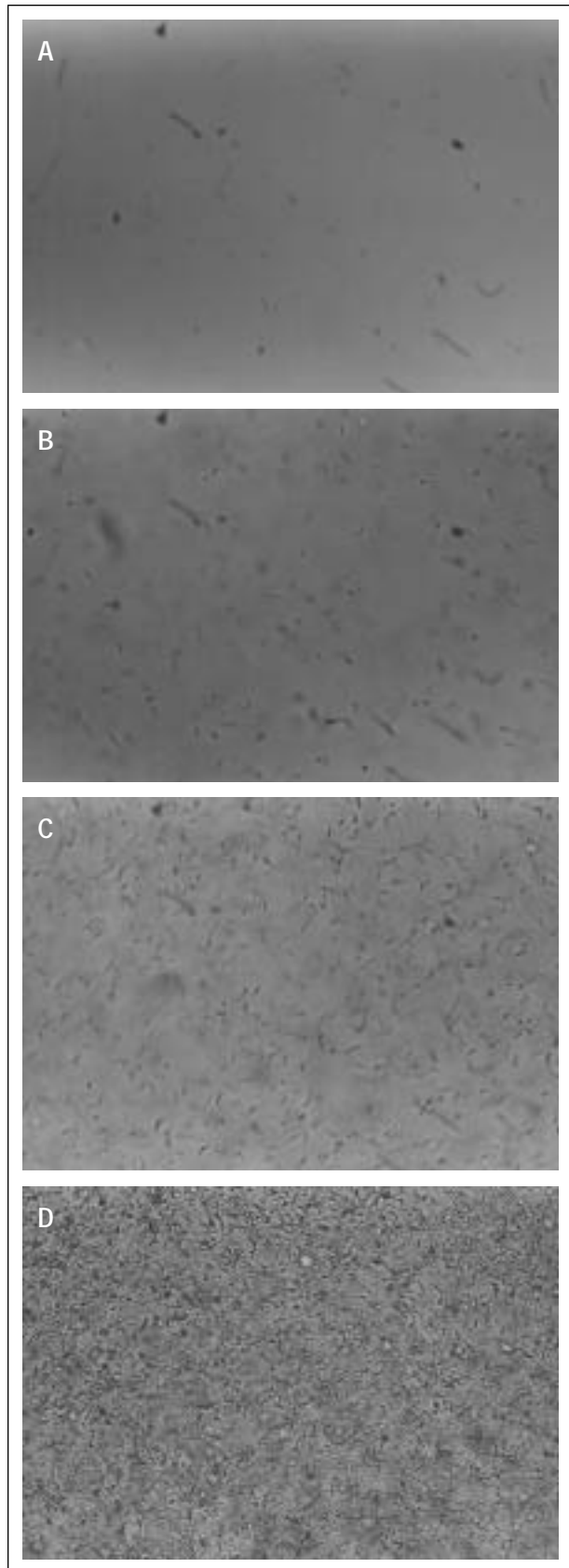


Fig. 11A.6. Crane sperm concentration scores: A (15x) = 1, B (15x) = 2, C (10x) = 3, D (5x) = 4.

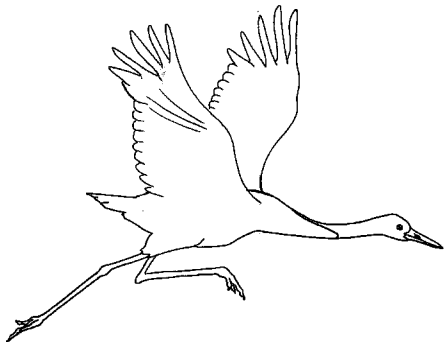
drop of 5% eosin (by weight) on the upper right hand corner of the slide. Place 3-4 drops of 10% nigrosin (by weight) directly below the eosin. Break off each end of the capillary tube and place one drop of semen from the tube on the slide close to but to the left of the eosin. Using a small glass rod, mix the eosin and semen, and then after 10 seconds, mix both with the nigrosin.

Place the end of another slide over the eosin-nigrosin stained semen sample at a 45° angle to the sample slide. Draw the upper slide toward the sample until the sample spreads along the underside of the upper slide. Next, push the upper slide away from the sample, along the surface of the lower slide, dragging out a thin uniform smear on the surface of the lower slide. Dry the slide quickly by waving in the air or, in damp weather, by exposing it to a warm air current.

For a permanent specimen, apply mounting medium when dry and mount a cover slip. You can view the slide under the microscope then or later when the mounting medium has dried. Count **clear cells as live** and **red or purple cells as dead**. Also, separate the live cells into five other categories: normal, bent, swollen, giant and droplet and compare them to normals (Fig. 11A.4) for other birds in your collection (Sharlin 1976; Gee and Temple 1978; Russman 1981; Sharlin et al. 1979).

AI As a Tool

First used in cranes in 1969, AI is now a practical propagation tool. The technique combines cooperative and massage methods to increase semen yield and egg fertility. Although semen characteristics are useful indicators of semen quality, the most reliable test is fertility rate.



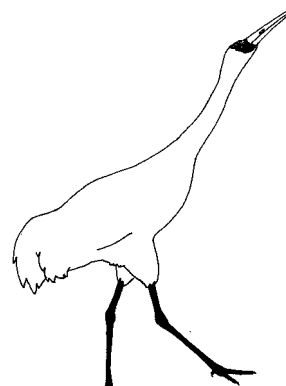
Literature Cited

- Archibald, G. S. 1974. Methods for breeding and rearing cranes in captivity. *International Zoo Yearbook* 14:147-155.
- Arcott, G. H., and R. V. Kuhns. 1969. Packed sperm volume versus optical density as a measure of semen concentrations. *Poultry Science* 48:1126-1127.
- Bakst, M. R. 1980. Sperm transport in the turkey. *Poultry Science* 59:1581.
- Bakst, M. R. 1981. Sperm recovery from oviducts of turkeys at known intervals after insemination and oviposition. *Journal of Reproduction and Fertility* 62:159-164.
- Berry, R. B. 1972. Reproduction by artificial insemination in captive American goshawks. *Journal of Wildlife Management* 36:1283-1288.
- Bird, D. M., P. C. Lague, and R. B. Buckland. 1976. Artificial insemination vs. natural mating in captive American kestrels. *Canadian Journal of Zoology* 54:1183-1191.
- Belterman, R., and C. E. King. 1993. Captive management and husbandry of Red-crowned Cranes in Europe. *International Zoo News* 40(1):6-20.
- Bobr, L. W., F. W. Lorenz, and F. X. Ogasawara. 1962. The role of the uterovaginal junction in storage of cock spermatozoa. *Poultry Science* 41:1628.
- Boyd, L. L., N. S. Boyd, and F. C. Dolber. 1977. Reproduction of prairie falcons by artificial insemination. *Journal of Wildlife Management* 41:266-271.
- Burt, C. T., and J. M. Chalovich. 1978. Serine ethanolamine phosphodiester a major component in chicken semen. *Biochimica et Biophysica ACTA* 529:186-188.
- Buxton, J. R., and F. S. Orcutt, Jr. 1975. Enzymes and electrolytes in the semen of Japanese Quail. *Poultry Science* 54:1556-1566.
- Carson, J. E., F. W. Lorenz, and V. S. Asmundson. 1955. Semen production in the turkey male. I. Seasonal variation. *Poultry Science* 34:336-343.
- Compton, M. M., and H. P. Van Krey. 1979a. Emptying of the uterovaginal sperm storage glands in the absence of ovulation and oviposition in the domestic hen. *Poultry Science* 58:187-190.
- Compton, M. M., and H. P. Van Krey. 1979b. A histological examination of the uterovaginal sperm storage glands in the domestic hen following an insemination with variable semen dosages. *Poultry Science* 58:478-480.
- Compton, M. M., H. P. Van Krey, and P. B. Siegel. 1977. The filling and emptying of uterovaginal sperm host glands in the domestic hen. *Poultry Science* 56:1705-1706.
- Compton, M. M., H. P. Van Krey, and P. B. Siegel. 1978. The filling and emptying of uterovaginal sperm host glands in the domestic hen. *Poultry Science* 57:1696-1700.
- Corten, P. J. M. 1973. Artificial insemination methods and equipment. *Captive Breed. Diurnal Birds of Prey* 1(4):3-5.
- Ellis, D. H., G. F. Gee, and K. O'Malley. 1991. First successful natural mating by captive-reared Whooping Crane. *Research Information Bulletin, U.S. Fish and Wildlife Service*. 91-80. 2 pp.

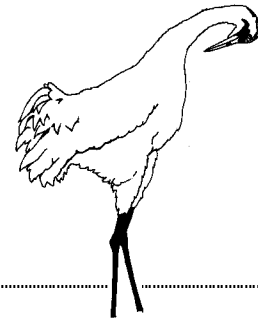
- Ferrier, W. T., H. B. Ortmyer, F. X. Ogasawara, and R. Zamanoto. 1982. The survivability of mycoplasma meleagridis in frozen-thawed turkey semen. *Poultry Science* 6:379-381.
- Fujihara, N., and H. Nishiyama. 1976. Studies on the accessory reproductive organs in the drake. Part 5. Effects of the fluid from the ejaculatory groove region on the spermatozoa of the drake. *Poultry Science* 55:2415-2420.
- Gale, J. A. 1987. Cryopreservation of semen from endangered crane species. University of Wisconsin, Madison. 58 pp.
- Gasparska, J., L. Ermenkova, Z. Jastrzberski, and G. Isaev. 1981. Activity, electrophoretic forms and morphologic characteristics of some enzymes in the blood and seminal plasma of cocks. *Prace i Materialy Zootechniczne* 0(26):9-18.
- Gee, G. F. 1969 unpubl. Reproductive physiology of the Greater Sandhill Crane. Pages 245-247 in Annual Progress Report Administrative Report, Patuxent Wildlife Research Center.
- Gee, G. F. 1983. Avian artificial insemination and semen preservation. Pages 375-398 in Jean Delacour/IFCB Symposium on Breeding Birds in Captivity. Hollywood, Calif.
- Gee, G. F., and T. J. Sexton. 1979. Artificial insemination of cranes with frozen semen. Pages 89-94 in J. C. Lewis, editor. Proceedings 1978 Crane Workshop. National Audubon Society and Colorado State Printing Service.
- Gee, G. F., and T. J. Sexton. 1990. Cryogenic preservation of semen from the Aleutian Canada Goose (*Branta canadensis leucopareia*). *Zoo Biology* 9:361-371.
- Gee, G. F., and S. A. Temple. 1978. Artificial insemination for breeding non-domestic birds. Symposia of the Zoological Society of London 43:51-72.
- Gee, G. F., M. R. Bakst, and T. J. Sexton. 1985. Cryogenic preservation of semen from the Greater Sandhill Crane. *Journal of Wildlife Management* 49(2):480-484.
- Grier, J. W. 1973. Techniques and results of artificial insemination with Golden Eagles. *Raptor Research* 7:1-2.
- Hamerstrom, F. 1970. An eagle to the sky. Iowa University Press, Ames. 142 pp.
- Howell, T. R., and G. A. Bartholomew, Jr. 1952. Experiments on the mating behavior of the Brewer Blackbird. *Condor* 54:140-141.
- Jones, J. E., and H. R. Wilson. 1967. Use of an electronic counter for sperm concentration determinations in chicken semen. *Poultry Science* 46:532-533.
- Kosin, I. L., and A. Wheeler. 1956. Methods for estimating spermatozoa numbers in turkey semen. *Northwest Science* 30:41-47.
- Lake, P. E. 1966. Physiology and biochemistry of poultry semen. Pages 93-123 in A. McLaren, editor. Advances in Reproductive Physiology. Logos Press, Ltd., London.
- Lake, P. E. 1969. Factors affecting fertility. Pages 3-29 in T. C. Carter and B. M. Freeman, editors. Fertility and hatchability of the hen's egg. British Egg Market Board Symposium No. 5. Oliver and Boyd, Edinburgh.
- Lake, P. E. 1971. The male in reproduction. Pages 1411-1447 in D. J. Ball and B. M. Freeman, editors. Physiology and biochemistry of the domestic fowl 3. Academic Press, New York.
- Lake, P. E. 1978. The principles and practice of semen collection and preservation in birds. Symposia of the Zoological Society of London 43:31-49.
- Lake, P. E. and J. M. Steward. 1978. Artificial insemination in poultry. Ministry of Agriculture, Fisheries, and Food [Great Britain], Bulletin No. 213.
- Lorenz, F. W. 1969. Reproduction in domestic fowl. Pages 569-608 in H. H. Cole and P. T. Cupps, editors. Reproduction in domestic animals. 2nd ed. Academic Press, New York.
- Mann, T. 1964. The biochemistry of semen and of the male reproductive tract. Methuen and Co., Ltd, London. 493 pp.
- McDaniel, G. R., and T. J. Sexton. 1977. Frequency of semen collection in relation to semen volume, sperm concentration, and fertility in the chicken. *Poultry Science* 56:1989-1993.
- Meyer, G. B., C. F. Props, A. T. Leighton, Jr., H. P. Van Krey, and L. M. Potter. 1980. Influence of dietary protein during the prebreeder period on subsequent reproductive performance of large white turkeys. 3. The effects of semen volume and frequency of insemination on fertility and hatchability. *Poultry Science* 59:363-368.
- Nishiyama, H., N. Nakashima, and N. Fujihara. 1976. Studies on the accessory reproductive organs in the drake. Part I. Addition to semen of the fluid from the ejaculatory groove region. *Poultry Science* 55:234-242.
- Ogasawara, F. X., and R. A. Ernst. 1970. Effects of three semen extenders on reproduction in turkeys. *California Agriculture*, August 1970:14-15.
- Ogasawara, F. X., and C. L. Fuqua. 1972. The vital importance of the uterovaginal sperm-host glands for the turkey hen. *Poultry Science* 51:1035-1039.
- Ogasawara, F. X., C. L. Fuqua, and K. C. Tripathi. 1976. Recommendations for using the live-dead stain with turkey spermatozoa. *Poultry Science* 55:2073.
- Olsen, M. W. 1942. Maturation, fertilization, and early cleavage in the hen's egg. *Journal of Morphology* 70:513.
- Perek, M., M. Elian, and E. D. Heller. 1969. Bacterial flora of semen and contamination of the reproductive organs of the hen following artificial insemination. *Research in Veterinary Science* 10:127-132.
- Putnam, M. S. 1982. Refined techniques in crane propagation at the International Crane Foundation. Pages 250-258 in J. C. Lewis, editor. Proceedings 1981 Crane Workshop. National Audubon Society, Tavernier, Fla.
- Quinn, J. P., and W. H. Burrows. 1936. Artificial insemination of fowls. *Journal of Heredity* 27:31-37.
- Russman, S. E. 1981. Sperm morphology in the crane. M.S. thesis, University of Illinois, Urbana. 87 pp.
- Russman, S. E., and P. C. Harrison. 1982. Sperm morphology in the crane. Pages 199-212 in J. C. Lewis, editor. Proceedings 1981 Crane Workshop. National Audubon Society, Tavernier, Fla.
- Servouse, M., M. J. Petitjean, and A. J. Rosenberg. 1976. A study of some seminal plasma enzymes of the cockerel. *Annales de Biologie Animale Biochimie Biophysique* 16:807-819.
- Sexton, T. J. 1976. Studies on the fertility of frozen fowl semen. Page 236 in Proceedings of the VIIIth International Congress on Animal Reproduction and Artificial Insemination, Krakow, Poland.

- Sexton, T. J. 1977. A new poultry semen extender. Part 1. Effect of extension on the fertility of chicken semen. *Poultry Science* 56:1443-1446.
- Sexton, T. J. 1978. A new poultry semen extender. 3. Effect of storage conditions on the fertilizing capacity of chicken semen stored at 5^o C. *Poultry Science* 57: 285-289.
- Sexton, T. J. 1979. Preservation of poultry semen: a review. Pages 159-170 *in* H. W. Hawk, editor. *Animal reproduction. Beltsville Agriculture Research Center Symposium* 3.
- Sexton, T. J., and G. F. Gee. 1978. A comparative study on the cryogenic preservation of semen from the Sandhill Crane and the domestic fowls. *Symposia of the Zoological Society of London* 43:89-95.
- Sexton, T. J., L. A. Jacobs, and G. R. McDaniel. 1980. A new poultry semen extender. 4. Effect of antibacterials in control of bacterial contamination in chicken semen. *Poultry Science* 59:274-281.
- Sharlin, J. S. 1976. Sperm head length as a predictor of fecundity in the Sandhill Crane. M.S. thesis, University of Maryland, College Park. 54 pp.
- Sharlin, J. S., C. S. Shaffner, and G. F. Gee. 1979. Sperm head length as a predictor of fecundity in the Sandhill Crane, *Grus canadensis*. *Journal of Reproduction and Fertility* 55:411-413.
- Smyth, J. R., Jr. 1968. Poultry. Pages 258-300 *in* E. J. Perry, editor. *The artificial insemination of farm animals*. Rutgers University Press, New Brunswick, N.J.
- Stipkovits, L., A. Rashwan, and M. Z. Sabry. 1978. Studies on pathogenicity of turkey ureaplasma. *Avian Pathology* 7:577-582.
- Sturkie, P. D. 1965. *Avian physiology*. Comstock/Cornell University Press, Ithaca, N.Y. 766 pp.
- Swengel, S. R., and G. W. Archibald. 1988. The status of cranes breeding in 1988. Pages 571-582 *in* 5th World Conference on Breeding Endangered Species in Captivity, Cincinnati, Ohio.
- Tan, N. S. 1980. The frequency of collection and semen production in Muscovy Ducks. *British Poultry Science* 21:265-272.
- Temple, S. A. 1972. Artificial insemination with imprinted birds of prey. *Nature (London)* 237:287-288.
- Thurston, R.J., R. A. Hess, H. V. Biellier, H. K. Aldinger, and R. F. Solorzano. 1975. Ultrastructural studies of semen abnormalities and herpesvirus associated with cultured testicular cells from domestic turkeys. *Journal of Reproduction and Fertility* 45:235-241.
- Watanabe, M., and T. Terada. 1976. A new diluent for deep freezing preservation of fowl spermatozoa. *Proceedings of the VIIIth International Congress of Animal Reproduction and Artificial Insemination* 274.
- Watanabe, M., and T. Terada. 1980. Fertility of frozen fowl semen stored for long-term, nine years. *Journal of the Faculty of Applied Biological Science Hiroshima University* 19:155-160.
- Watanabe, M., Y. Matsumoto, N. Takeshita, and T. Terada. 1981. Fertility of muscovy semen frozen for about three years. *Journal of the Faculty of Applied Biological Science Hiroshima University* 20:81-86.
- Wentworth, B. C., M. J. Wineland, and G. D. Paton. 1975. Fertility of turkey hens correlated with depth of insemination. *Poultry Science* 54:682-687.





Special Techniques, Part B: Cryopreservation



Timothy L. Hargrove and George F. Gee

Chicken semen was first frozen in 1941 (Shaffner et al. 1941). Since that time, frozen semen has produced fertile eggs from various species of raptors (Brock 1986; Parks et al. 1986; Gee et al. 1993), cranes (Gee et al. 1985; Gale 1987; Hargrove et al. 1993 unpubl.), geese (Gee and Sexton 1990), and psittacines (Hargrove 1986; Samour et al. 1988).

Maximizing fertility and genetic diversity are important for breeding endangered species in captivity, and cryogenic preservation of semen can accomplish both. If gamete production is asynchronous, the female may be artificially inseminated with frozen-thawed semen samples (Fig. 11b.1). Furthermore, frozen semen banks can protect the founder gene pool for generations.

Collection and Dilution

Avian semen was first collected by the massage technique in chickens (Burrows and Quinn 1939). This same technique has been applied to semen collection in other avian species including cranes (Archibald 1974; Gee and Temple 1978; Chapter 11A). Some research projects may require collecting semen by narrow-mouthed devices (e.g., a 0.25-mL caraway or a 0.35-mL natelson capillary tube), but normally, samples are collected in wide-mouthed devices such as a close-ended funnel (35-mm diameter cup with 50-mm stem; Fig. 11a.2) or some other similar glass container. Any surface which comes into contact with the semen should be clean, dry, sterile, and never contain soap residue. Immediately after collection, draw a tiny (ca 0.01 mL) sample into a capillary tube (as described in Chapter 11A). This tube is later examined for sperm concentration, motility, and for live-dead counts. Few cells in the tube die in the hour or less needed to return to the laboratory if the weather is cool or if the tube is stored in a cool container (ice bath or thermos). Transfer the ejaculate to a Pasteur pipet sealed at



Fig. 11b.1 Bruce Williams holding the first crane chick, a Greater Sandhill Crane, ever produced from frozen semen, 1978.

Photo George F. Gee

the small end. Determine the volume with an open-ended tom cat catheter (14 cm) attached to a 1-cc syringe. Dilute the ejaculate with one part crane semen extender (1:1), which is a modified version of the Beltsville Poultry Semen Extender (BPSE; Sexton 1977; Gee et al. 1985). Label the Pasteur pipet for identification and cover it with parafilm to prevent evaporation and contamination. These procedures are

performed at ambient temperature.

Place the Pasteur pipet inside a larger test tube and transfer the two into an insulated ice bath (0-4°C). The water level in the ice bath should be sufficiently high to cover at least the lower portion of the tube containing the diluted ejaculate. The tube-within-a-tube arrangement slows the rate at which the ejaculate cools, thereby promoting survival of the spermatozoa. The top of the insulated container is closed between samples to maintain the desired temperature and to decrease the effects of airborne contamination and sunlight. Uncontaminated semen may be held in this manner up to 3 hours prior to freezing (Gee 1991).

Sample Preparation

Upon arrival at the lab, the 0.01-mL sample in the capillary tube is examined for motility, concentration, and urate contamination. Samples showing good concentration with little or no urate contamination,

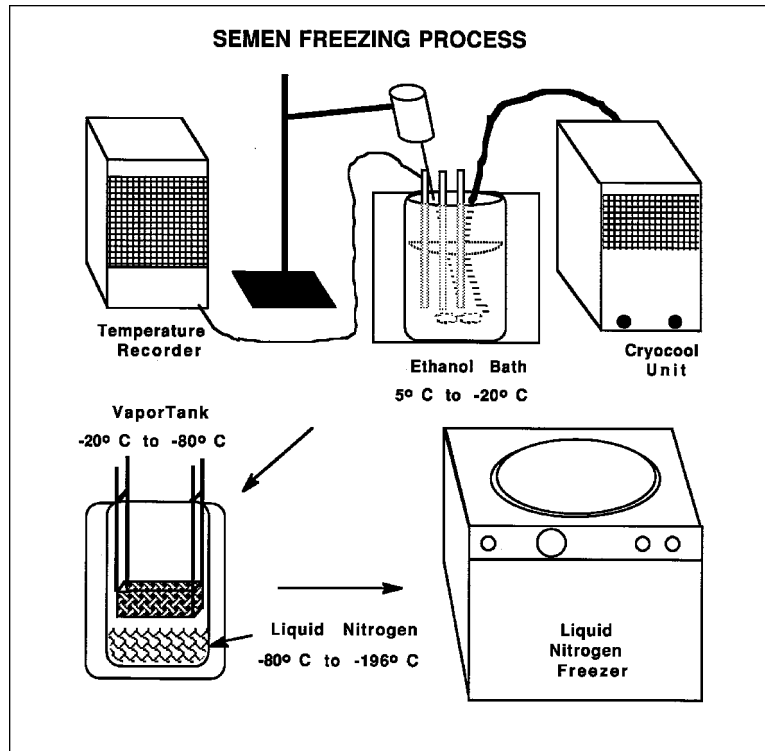


Fig. 11b.2 Semen freezing process.

irrespective of the degree of motility, are prepared for freezing.

As mentioned earlier, semen is diluted when collected with an equal volume of crane extender. Samples that will be frozen are diluted by one-half the volume of the diluted ejaculate with 24% dimethylsulfoxide (DMSO) in crane extender to get a final 8% DMSO concentration (one part semen, one part extender, and one part DMSO extender). When adding the DMSO, the 24% DMSO in crane extender and diluted ejaculate should be at the same temperature (0–4° C). DMSO helps protect the sperm from damage during freezing and thawing. For the maximum cryoprotectant effect with the least amount of cell toxicity, the ejaculate should equilibrate with DMSO for 15 minutes in the ice bath (Gee 1991).

While the semen sample is equilibrating, the ethanol bath (in which the straws containing the semen are placed for freezing) is cooled to 5° C. For the ethanol bath, we use a Neslab Agitanor (an insulated multipurpose bath with stirrer; see Appendix) which is cooled by a Neslab Cryocool Immersion Cooler CC-100 (a refrigeration compressor immersion probe; see Appendix). The stirrer in the ethanol bath maintains a constant temperature around each cane. (Canes are 29.3 cm long, semicircular with a flat top

and bottom and are constructed of aluminum). A temperature probe is placed in the ethanol bath on a semen cane. The probe consists of a Type T bimetal thermocouple in a semen straw containing 8% DMSO in crane extender and is sealed on top with vinyl plastic putty (e.g., Critoseal, see Appendix). The thermocouple is attached to a temperature-recording unit (Honeywell Electronik III, Type T; see Appendix), a single pen strip chart recorder with a temperature range of 5° to –200° C.

Near the end of the equilibration period, heat-seal the straws on one end. These straws are either 0.2- or 0.5-mL and are labeled by male ID and colony. First, reserve a small residual semen sample (0.01 mL) for density and live-dead counts (these counts are performed after the samples are frozen). Then, transfer the remaining semen sample to the straws (using a tom cat catheter and a 1-cc

syringe) and heat-seal the other end. The sealed straws are transferred to a consecutively labeled cane (e.g., cane 88-64 is the 64th sample frozen in 1988) and placed in the ethanol bath.

The first step of the freezing process (Fig. 11b.2) is cooling the samples in the ethanol bath from +5° to –20° C at a rate of –1° C/min. Second, cool the samples from –20° to –80° C at a rate of –50° C/min by placing them in liquid nitrogen vapor. This may be accomplished by holding the samples in the neck of the storage tank or by placing them in a vapor tank filled with 5 cm of liquid nitrogen. Third, plunge the samples into liquid nitrogen (–196° C; a freezing rate of –160° C/min; Gee et al. 1985). If a vapor tank is used in steps two and three, the samples should remain in the vapor tank for 5 min before transferring to the liquid nitrogen storage tank.

Determine sperm density using a hemacytometer. First, draw up a 0.01-mL subsample of ejaculate you prepared for freezing. Then dilute this 50:1 by adding 0.5 mL neutral formalin. You can purchase neutral formalin or prepare your own by adding 5 g sodium bicarbonate and 1 mL commercial formalin to 100 mL distilled water. Thoroughly mix the semen and formalin and allow to sit for 5 min. Mix the sample and place a small portion of this solution on a hemacytometer (Fig. 8.7). Wait 10–15 min for the sperm cells to settle into one focal plane (if more time is allowed

the sample may dehydrate). At 100X magnification, count the sperm cells within 5 of the 25 small squares in the 1-mm square. Then calculate density with the following formula:

$$\text{(No. Sperm Cells} \times \text{Dilution} \times 4000) / \text{No. Squares Counted} = \text{No. Sperm Cells} / 0.001 \text{ mL Ejaculate.}$$

The number of smallest squares counted is 80 (5 × 16) if only one grid of the hemacytometer is counted and 160 if both sides are counted. To get the number of sperm cells/mL, multiply the calculated value by 1000.

For example, if both sides of the hemacytometer add up to 145 sperm cells and the dilution is 200X, then the results are as follows:

$$(145 \times 200 \times 4000) / 160 = 725,000 \text{ sperm} / 0.001 \text{ mL} = 725 \text{ million/mL.}$$

Knowing the original undiluted frozen volume, the percent live, and the density, one can calculate the number of live sperm cells in the freshly frozen sample. For example, if

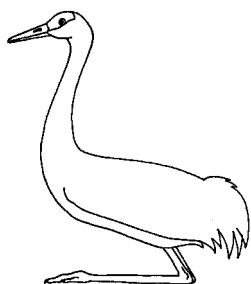
0.05mL = original ejaculate volume,

725 million/mL = density, and

90% of the sperm cells were alive,

then $0.05 \times 725,000,000 \times .900 = 32,625,000$ live sperm cells in the original sample.

Live-dead counts are made with 5% eosin and 10% nigrosin stain (Burrows and Quinn 1939; Hackett and Macpherson 1965). Place one drop of semen on a clean dry glass slide, one drop of 5% eosin next to the drop of semen, and 3 drops of 10% nigrosin next to the eosin. Mix the eosin and sperm and let sit for 5-10 seconds. Then mix with the 3 drops of 10% nigrosin, spread the sample like a blood smear, and air dry it quickly. Do *not* dry over a flame, but a flow of warm air (e.g., from a hair dryer) may be used. Glue a 24 × 50 mm coverslip to the slide with Permount (see Appendix), balsam, or other mounting medium. The slide can be examined now or later for live and dead sperm. Live sperm appear white against the blue (nigrosin) background; dead sperm appear red (eosin) or pink against the blue background. Determine percentage live by counting 300 sperm on each of three slides or 500 on each of two slides.



Use of Frozen Semen

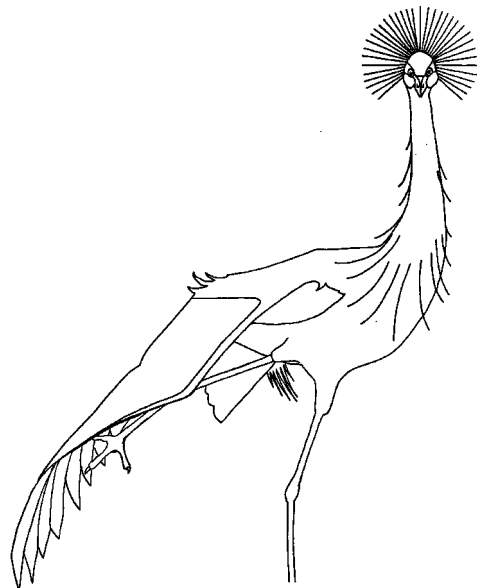
To thaw frozen samples for insemination, transfer the canes from the storage tank to an ice bath (crushed ice saturated with water, 0.5° C) for 3-5 min. Remove the straw containing the thawed semen, dry the surface, and cut off one end. Transfer the thawed semen with a tom cat catheter attached to a 1-cc syringe from the straw to a closed end Pasteur pipet in an ice bath or keep in the syringe for insemination. Examine the samples for motility and live cells as described earlier.

Use the equivalent of two to four ejaculates for each insemination to compensate for the loss of cells during freezing (50-60%) and on the sides of tubes, straws, and transfer devices. To achieve good fertility, use 15-20 million live cells per insemination (Gee et al. 1985). Samples should be inseminated immediately after thawing and in the same manner as fresh semen.

Literature Cited

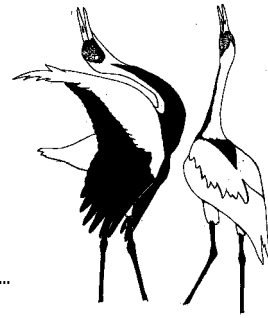
- Archibald, G. S. 1974. Methods for breeding and rearing cranes in captivity. *International Zoo Yearbook* 14:147-155.
- Brock, M. K. 1986. Cryopreservation of semen of the American Kestrel (*Falco sparverius*). M.S. thesis, MacDonald College of McGill University, Montreal, Canada. 85 pp.
- Burrows, W. H., and J. P. Quinn. 1939. Artificial insemination of chickens and turkeys. U.S. Department of Agriculture Circular No. 525:1-12.
- Gale, J. R. 1987. Cryopreservation of semen from endangered crane species. M.S. thesis, University of Wisconsin, Madison. 58 pp.
- Gee, G. F. 1991. Cryopreservation of crane semen. Pages 453-456 *in* J. Harris, editor. Proceedings 1987 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Gee, G. F., M. R. Bakst, and T. J. Sexton. 1985. Cryopreservation of semen from the Greater Sandhill Crane. *Journal of Wildlife Management* 49:480-484.
- Gee, G. F., C. A. Morrell, J. C. Franson, and O. H. Pattee. 1993. Cryopreservation of American Kestrel semen with dimethylsulfoxide. *Journal of Raptor Research* 27:21-25.
- Gee, G. F., and T. J. Sexton. 1979. Artificial insemination of cranes with frozen semen. Pages 89-94 *in* J. C. Lewis, editor. Proceedings 1978 Crane Workshop. National Audubon Society and Colorado State University Printing Service.
- Gee, G. F., and T. J. Sexton. 1990. Cryogenic preservation of semen from the Aleutian Canada Goose. *Zoo Biology* 9:361-371.

- Gee, G. F., and S. A. Temple. 1978. Artificial insemination for breeding non-domestic birds. *Symposia of the Zoological Society of London* 43:51-72.
- Hackett, A. J., and J. W. Macpherson. 1965. Some staining procedures for spermatozoa. A review. *Canadian Veterinary Journal* 6:55-62.
- Hargrove, T. L. 1986. Cryogenic preservation of Budgerigar (*Melopsittacus undulatus*) semen. M.S. thesis, Florida Atlantic University, Boca Raton. 40 pp.
- Hargrove, T. L., G. F. Gee, and C. Mirande. 1993 unpubl. Comparison of two DMSO levels on the fertilizing ability of semen in three species of cranes.
- Parks, J. E., W. R. Heck, and V. Hardaswick. 1986. Cryopreservation of Peregrine Falcon semen and post-thaw dialysis to remove glycerol. *Raptor Research* 20:16-20.
- Samour, J. H., J. A. Markham, and H. D. M. Moore. 1988. Semen cryopreservation and artificial insemination in Budgerigars (*Melopsittacus undulatus*). *Journal of Zoology (London)* 216:169-176.
- Sexton, T. J. 1977. A new poultry semen extender. I. Effect of extension on the fertility of chicken semen. *Poultry Science* 56:1443-1446.
- Shaffner, C. S., E. W. Henderson, and C. G. Card. 1941. Viability of spermatozoa of the chicken under various environmental conditions. *Poultry Science* 20:259-265.



Special Techniques, C: Sex Determination

Scott R. Swengel



Cranes are considered **monomorphic**. Although males average slightly larger, size is not always reliable as an indicator of sex. Subadult and adult cranes can normally be sexed by vocalizations, and there is potential for sexing chicks by sonagram analysis (Carlson 1991). General behavior can also indicate, but not diagnose, sex. **Fecal steroid analysis** can be used to sex nearly all subadults, adults, and many young birds, and cranes of all ages can be **genetically sexed** by DNA probe, by microscopic examination of their chromosomes (karyotyping), or by **total DNA** measurements. Cranes that are 3 months old or older can be gonadally sexed via **laparoscopy**.

Vocalizations

Cranes approach “vocal maturity” when they are 8 to 12 months old (Walkinshaw 1973; Nesbitt 1975; personal observation). Vocally mature cranes have sex-specific differences in their **Unison-calls** and, in many species, their **Guard-calls** (Archibald 1976a, 1976b; Carlson and Trost 1992). Sonagram analysis of vocalizations is an accurate way to sex adult and subadult cranes (Archibald 1976a, 1976b; Carlson 1991; Carlson and Trost 1992), and perhaps most Whooping Crane chicks (G. Carlson, Idaho State University, Pocatello, Idaho, personal communication).

Guard-call

Cranes Guard-call by giving one loud burst, pausing one or more seconds, giving another call, and so on. Mates often give synchronous Guard-calls. Male cranes have lower pitched voices than females in all species except the African Crowned Cranes. These differences are most obvious when cranes of both sexes are calling, but experienced persons can sex birds without reference to another crane.

Unison-call

Archibald (1976a, 1976b) studied the Unison-call display in detail and found ways to differentiate the sexes of all but the African Crowned Cranes. In most species, sexes have different **stances** (Fig. 11C.1), and/or different **number of notes** given, during the Unison-call. The principal differences in Unison-calls that indicate sex fall into five groups: (1) in some species, the male raises his elbow and/or lowers his primaries during the display, while the female does not; (2) in some species, the female usually, or always, begins the display; (3) in some species, the female gives two to three notes for each male note of the display; (4) in some species, the male holds his bill more vertically, or further over his back beyond the vertical, than the female; and (5) in most species, the female’s voice is higher pitched than the male’s.

Black Crowned and Gray Crowned Cranes. Crowned Cranes usually incorporate a series of **ka-wonk** like Guard-calls into their Unison-calls. The slight differences in pitch between the sexes are not sufficient to diagnose a crane’s sex by ear. The Black Crowned Crane usually uses Guard-calls exclusively. The only sexual difference in this species is the female’s higher pitched ka-wonk calls. Gray Crowned Cranes, on the other hand, use low-pitched **booms** as their Unison-calls, and employ ka-wonks as their Guard-calls. The female’s ka-wonk calls average slightly longer than the male’s in this species. Although both sexes have similarly pitched ka-wonk calls, the female’s booming calls are lower pitched than the male’s.

Demoiselle Crane. The female usually begins the Unison-call by throwing her head back beyond the vertical, while the male follows her first note by giving lower pitched, longer, and more broken notes. He holds his neck vertical with the bill elevated 45° above the horizontal. The female either holds her initial position or gradually returns her head to a horizontal position. The female’s voice is higher pitched than the male’s.

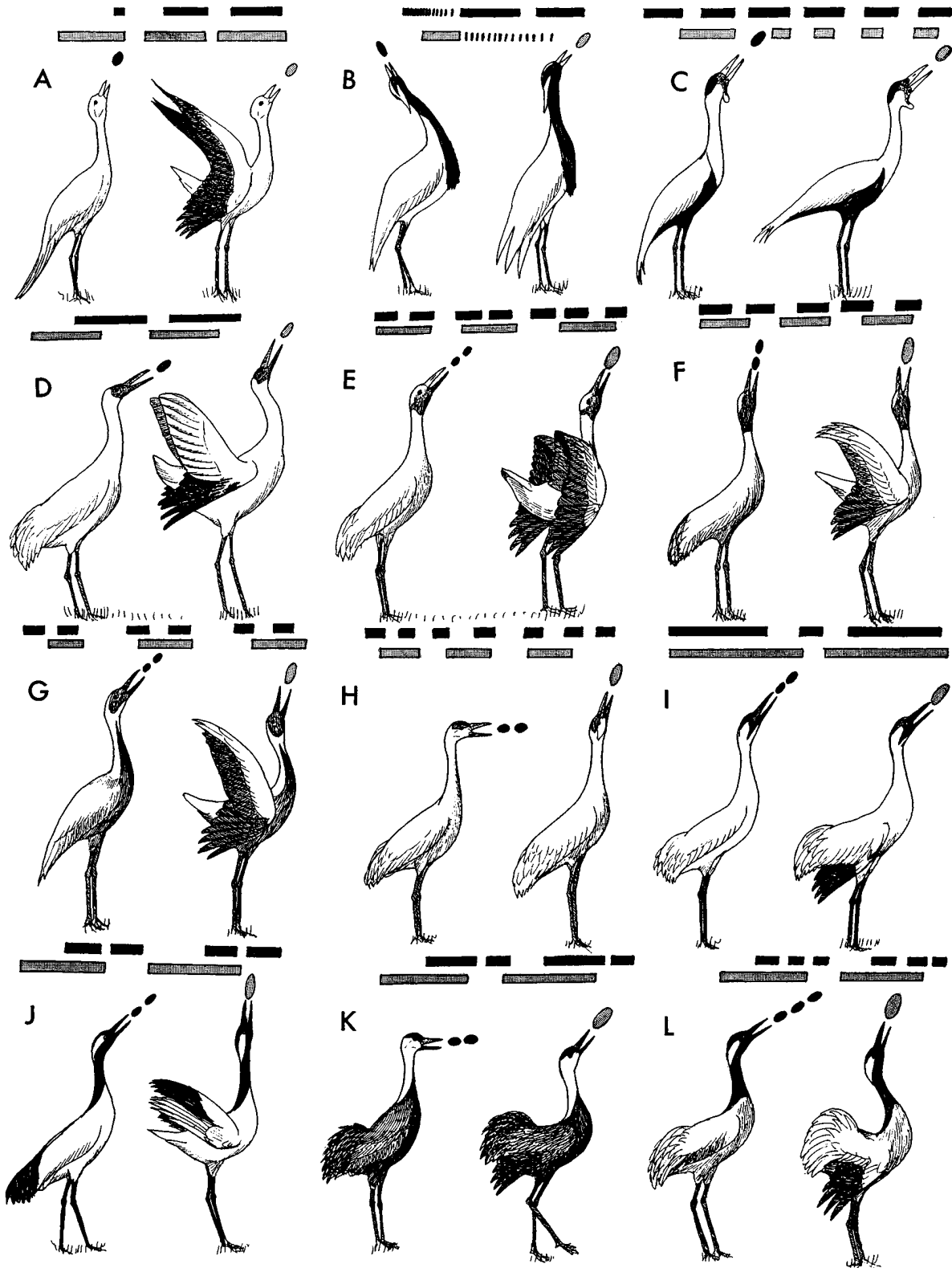


Fig. 11C.1. Male and female Unison-call postures for the following crane species: (A) Blue, (B) Demoiselle, (C) Wattled, (D) Siberian, (E) Brolga, (F) Sarus, (G) White-naped, (H) Sandhill, (I) Whooping, (J) Red-crowned, (K) Hooded, and (L) Eurasian. Durations of vocalizations are indicated by shaded bars (male) and black bars (female). "Balloons" indicate typical number of female calls per male call. Based on Archibald (1976b).

Art Paul A. Johnsgard. Used by permission from Johnsgard (1983).

Blue Crane. The female usually throws her neck back 20° beyond the vertical at the start of the call. The male throws his head and neck back even further, about 40° beyond the vertical. The male raises his humeri above the back during the display, while the female does not.

Wattled Crane. The female begins the call by rapidly lowering her head to shoulder level, then rapidly raising her neck with the head about 30° in front of the vertical. She maintains this posture throughout the call. The male quickly follows the female's introduction with a long and partly broken call, then gives longer, fewer, and lower pitched notes than the female as in the Indian Sarus Crane. With his last note, which is long and broken like the first, he raises his humeri 20° above the back. The female does not elevate her wings.

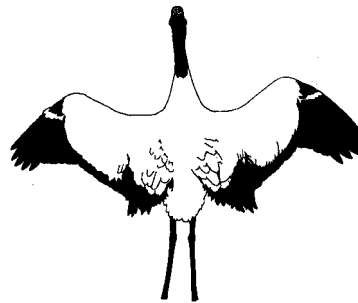
Siberian Crane. The female's voice is higher pitched than the male's, but because some males are higher pitched than others, this male-female difference is sometimes apparent only when the two sexes can be compared. Either sex may lower its primaries or walk during the display. One sex-specific difference is that when the male begins the call, he swings his head up quickly and then throws it down near his chest with a long preliminary note. Sometimes, however, the female begins the call.

Other Grus Cranes. In most of the remaining Grus cranes the female gives two to three notes for each male note of the Unison-call. There are sexual differences in the stance during the display in all of these species.

In the **Sandhill Crane**, the female usually initiates the call with an explosion of rapid notes. The male sometimes begins the call. The female gives two or three notes for every male note. The male holds his head further back and with the bill more vertical than the female. The female holds her bill nearly level, but quickly flips her bill up with each note. The female's voice is higher pitched than the male's in both the Unison- and Guard-calls. The female's Guard-call notes are more broken than the male's.

In the **Sarus, Brolga, and White-naped Cranes**, the male raises his humeri high above the back and completely lowers his primaries during the Unison-call, creating an impressive visual display (Fig. 6.10). The female does not move her wings during the display, and her voice is higher pitched. The female usually begins the Unison-call in two of these species, and always does so in the White-naped Crane.

In the **Common, Hooded, Whooping, Black-necked, and Red-crowned Cranes**, the male usually raises his humeri during the display, often raising and lowering them with each note. The female does not raise her humeri, except occasionally in the Whooping Crane and rarely in the Common Crane. In these two species, the male raises his humeri more often than the female and extends his head far over his body, while the female has a more vertical stance. In each of these five species the female gives two or three notes for each male note of the Unison-call, and her voice is higher pitched.



General Differences in Behavior Between the Sexes

In wild cranes, the male nearly always leads the female when a pair moves from place to place (Tacha 1987). The male is the principal defender of the pair and tends to adopt more erect and aggressive postures with his head held higher than the female's. The male often spends more time watching for intruders than the female. The female more often shows neck-retracted submissive postures when an intruder approaches. By contrast, the female in captivity very often, and perhaps most of the time, initiates dance and calling activities.

Captive, hand-reared males often attack people and may become more aggressive after being paired. Hand-reared females may also approach people but are usually more submissive than males. Once paired, the female may become aggressive like the male. The male of a pair usually displays more intensely, and approaches an intruder closer, than the female.

Bare-skin- or Wattle-expansion reflects increased aggression in cranes. Males usually expand their crowns or wattles more than females. In the Wattled Crane, an aggressive male may have larger wattles than his mate during conflicts.

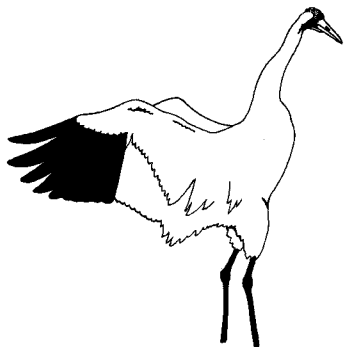
Size

Male cranes are usually heavier and taller than females, but there is considerable overlap between the sexes (Walkinshaw 1973; Johnsgard 1983). It is possible to sex larger-than-average males and smaller-than-average females by this method. Captive males of six species averaged 14.5-28.5% heavier, had 2.5-11.7% longer culmens, and 3.3-11.1% longer tarsi than conspecific females (Swengel 1992). Because cranes are usually heavier in captivity, their weights cannot be compared with wild birds.

There is also considerable **seasonal variation in weight**. Winter weights may be greater than summer body weights by 35% or more. For temperate and arctic-nesting cranes, weight is much more variable, hence much less useful as an indicator of sex (Song 1991; Swengel 1992).

Multi-parameter discriminant functions are now available to sex adults of two species. Murata et al. (1988) found that a discriminant function combining tail, wing chord, tarsus, and culmen measurements could safely sex captive Red-crowned Cranes. Markin and Krever (1992) developed a similar function using culmen, tarsus, and middle toe to sex wild Common Cranes. They have also created discriminant functions for sexing Demoiselle, Siberian, Sandhill, White-naped, and Red-crowned Cranes (V. Krever, Central Laboratory of Game Management, Moscow, Russia, unpublished data [on file at ICF]). Because of feather wear, wing chord and tail measurements are not very useful for sexing cranes.

In 20 wild Brolga pairs, Blackman (1971) found that males were always heavier and had longer heads, tarsi, and bodies than their mates. This suggests that female Brolgas, and possibly other cranes, choose mates larger than themselves.



Genetic Sexing

Karyotyping

Male cranes have two Z sex chromosomes, while females have one Z and one W chromosome (Fig. 11C.2). A crane's sex can be determined by the number of large chromosomes (**macrochromosomes**) (Rasch and Kurtin 1976; Sasaki and Takagi 1981; Goodpasture et al. 1992); cranes have four to five pairs of macrochromosomes besides the sex chromosomes. The Z chromosome is about as large as the fourth or fifth largest pair of chromosomes, while the W chromosome is much smaller. In the female, dividing mitotic cells in metaphase (chromosome spread) contain four or five paired and one unpaired macrochromosome.

Chromosomes can be obtained either from a blood sample or from the pulp of a growing feather. Some investigators karyotype dividing white blood cells (Takagi et al. 1972; Biederman and Lin 1982) or feather pulp cells (Sasaki et al. 1968; Goodpasture et al. 1992). In birds, the feather pulp culture method is usually more successful in creating good chromosome spreads.

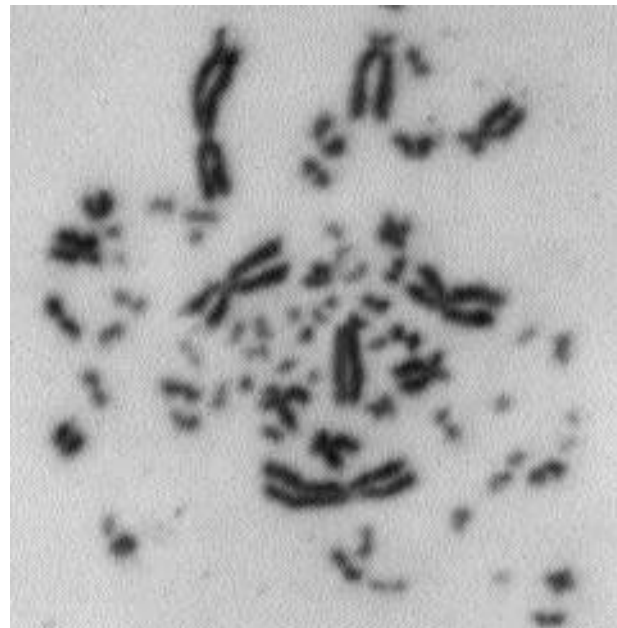


Fig. 11C.2. Chromosome spread of a female Whooping Crane, with the Z chromosome identified. The W chromosome is one of the very small chromosomes located through a process of elimination. Males have two W chromosomes.

Photo George F. Gee

Feather Pulp Sexing. For this method, a **large emerging feather** is pulled from the wing or tail. The feather can be obtained from a chick or an actively molting crane. If the bird to be sexed has no growing feathers, one or two large feathers can be pulled three to four weeks prior to the sampling date to stimulate regrowth. We pull two feathers to be sure at least one is growing (in the event one does not immediately grow back).

Gee (1982) found that pulling fully grown primaries from immature Sandhill Cranes often resulted in replacements that were deformed, were incapable of supporting flight, and were replaced several times in a year. To avoid this problem, we recommend using tail feathers or tertiaries. Pulling growing feathers seems to cause less harm to the feather follicle than pulling a fully grown feather.

When pulling growing feathers, be certain to remove the entire quill from the feather follicle. A large pair of pliers or hemostats provides a good purchase. Attach them as close to the base of the feather as possible, and always pull straight out. If a portion of the broken shaft is left in the follicle, bleeding may be prolonged. Although the condition is very rare, cranes have died from excessive blood loss through a broken blood quill of a large feather (G. F. Gee, Patuxent, personal communication). Most replacement secondaries are morphologically normal (personal observation), although their future loss and replacement rates have not been studied.

Pull the growing feather, wipe the shaft with alcohol, and cut it 2-3 cm from its insertion with sterile scissors. Immediately place the basal part of the feather in media provided by a cytological sexing lab. Contact the lab in advance to obtain shipping media and instructions for shipment. Samples should be shipped as soon as possible and should be refrigerated until shipping. Commercial feather pulp sexing labs in the United States include Avian Genetics Sexing Lab and Avigen (see Appendix).

Blood Culture. Remove the crane's food five hours before drawing blood. Place 0.2 mL of sodium heparin (10,000 units/mL) into a sterile 3-cc syringe and work the plunger to coat the barrel of the syringe. Draw 2-3 cc of blood from the jugular or brachial vein, and invert the syringe several times to mix the blood with the heparin. Keep the blood refrigerated, and send it to a lab as soon as possible. In the United States and Canada, some research labs at major universities and zoos karyotype birds using blood cells

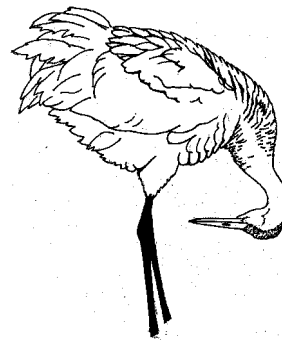
(Takagi and Sasaki 1974; Biederman et al. 1982; Kumamoto 1984; see Appendix).

DNA Probe

Zoogen, Inc. (see Appendix) has developed a commercially available DNA probe technique (RFLP, Restriction Fragment Length Polymorphism) to differentiate male and female chromosomes. A 0.02-mL sample of a bird's blood (Halverson 1990), mixed with 70% ethanol, is sent by regular mail. This method has correctly sexed over 100 cranes at ICF and can be used on chicks.

Genome Size

A rarely used cytological sexing method is a measurement of the **total DNA content** of crane erythrocytes. Because the Z chromosome is larger than the W, males have larger genomes than females. Rasch (1976, 1977) found that males had 4-6% larger DNA Feulgen staining levels than females in their erythrocyte nuclei as determined by cytophotometry. This method requires only a few drops of blood.



Surgical Sexing

This method uses a laparoscope or otoscope (fiberoptic or otherwise) to view the sex organs through a small incision in the crane's left side (McDonald 1982; see description in Chapter 8). Cranes should be **anesthetized** before surgery to decrease both stress and the risk of accidental injury to vital organs if the bird struggles during laparoscopy. Because of the risk of injury, a veterinarian or trained technician should perform this operation. For birds in general, about 1 bird in 250 dies during laparoscopy (McDonald 1986).

On inspection, the **testes** are white to tan, rather cylindrical, and smooth on the surface. In young

cranes, they are small (0.1-0.2 mm by 0.5-1.21 mm) and usually avascular, while in mature cranes they have vascularized surfaces, are much larger (2-3 cm by 3-5 cm), and vary in size seasonally. The ovary is often not found in laparoscopy of young females. When visible, it may be flat, is pink to tan, and looks like "pebbled" fat. In subadult females, the ovary acquires a fine granular surface, and when the bird is mature, the follicles appear like a cluster of grapes. Nearly all female birds have only one functioning ovary (the left).

Vent Sexing

Most adult cranes, and some subadults or even yearlings, can be vent-sexed by methods described by Blackman (1971) and Tacha and Lewis (1979). While one person uses the massage technique to relax the bird as in artificial insemination (see Chapter 11A), a second person examines the cloaca, using the fingers to get a better view of critical features. Male cranes have two raised papillae side by side in the middle of the unmanipulated cloaca; these average 1 mm across and are usually lighter in color than the surrounding tissue. The presence of cloacal papillae in cranes older than one year is indicative of a male; their absence indicates a female. Most females greater than a year old have one or more corroborative cloacal features: an oviduct opening on the lower left of the cloaca when viewed from behind, and a small spot near the top of the oviduct, the bursa of Fabricius. Both are easiest to see during the breeding season and in reproductive females. Blackman (1971) illustrates these features, but the photos are upside down with respect to the view described above.

Fecal Steroid Sexing

The feces of female birds have higher immunoreactive estrogen/testosterone (E/T) ratios than males (Czekala and Lasley 1977). One fresh fecal sample is enough to determine sex. This method is about 90% accurate (Stavy et al. 1979), but seasonal and age variation can cause occasional overlap in the hormone ratios of the two sexes, especially during the nonbreeding season (Bercovitz 1983). Stavy et al. (1979) found that low testosterone values in adult birds was indicative of a

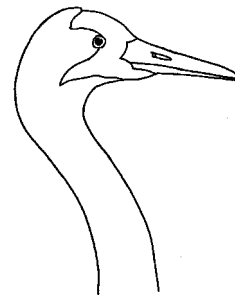
female, regardless of the E/T ratio. Fecal samples should be analyzed immediately or frozen and stored at temperatures below -40°C .

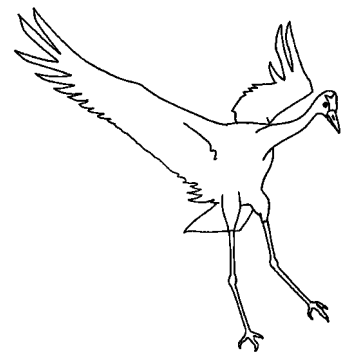
Adult birds during the breeding season give the best results. However, waste material left in the egg when a bird hatches has been used for sexing hatchlings. The technique works even for tiny chicks because sex hormones are important in the sexual differentiation of bird embryos (T. Gross, Biotechnologies for the Ecological, Evolutionary, and Conservation Sciences, Gainesville, Florida, and A. Bercovitz, San Diego Zoo, San Diego, California, personal communications) and egg wastes sample a relatively large time period.

Literature Cited

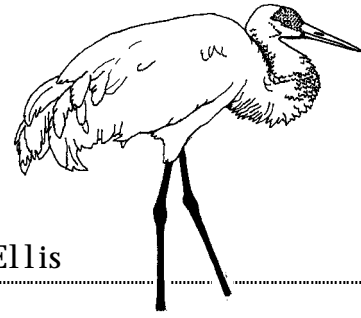
- Archibald, G. W. 1976a. Crane taxonomy as revealed by the unison call. Pages 225-251 in J. C. Lewis, editor. Proceedings of the International Crane Workshop. Oklahoma State University Publishing and Printing, Stillwater.
- Archibald, G. W. 1976b. The unison call of cranes as a useful taxonomic tool. Ph.D. dissertation, Cornell University, Ithaca, N.Y. 167 pp.
- Bercovitz, A. 1983. Applications for fecal steroid analysis (waste not want not). Pages 513-523 in Proceedings IFCB/Jean Delacour Symposium on Breeding Birds in Captivity. International Foundation for the Conservation of Birds, North Hollywood, Calif.
- Biederman, B. M., and C. C. Lin. 1982. A leukocyte culture and chromosome preparation technique for avian species. *In Vitro* 18:415-418.
- Biederman, B. M., C. C. Lin, E. Kuyt, and R. C. Drewien. 1982. Genome of the Whooping Crane. *Journal of Heredity* 73:145-148.
- Blackman, J. G. 1971. Sex determination of Australian Cranes (Gruidae). *Queensland Journal of Agriculture and Animal Science* 28:281-286.
- Carlson, G. 1991. The feasibility of individual identification and sex determination of Whooping Cranes (*Grus americana*) by analysis of vocalizations. M.S. thesis, Idaho State University, Pocatello. 60 pp.
- Carlson, G., and C. H. Trost. 1992. Sex determination of the Whooping Crane by analysis of vocalizations. *Condor* 94:532-536.
- Czekala, N. M., and B. L. Lasley. 1977. A technical note on sex determination in monomorphic birds. *International Zoo Yearbook* 17:209-211.
- Gee, G. F. 1982. Hormonal treatment and flight feather molt in immature Sandhill Cranes. Pages 237-243 in J. C. Lewis, editor. Proceedings 1981 Crane Workshop. National Audubon Society, Tavernier, Fla.

- Goodpasture, C., G. Seluja, and G. Gee. 1992. Karyotype and identification of sex in two endangered cranes species. Pages 219-224 in D. A. Wood, editor. Proceedings 1988 North American Crane Workshop. Florida Nongame Wildlife Program Technical Report #12.
- Halverson, J. 1990. Avian sex identification by recombinant DNA technology. Pages 257-262 in 1990 Proceedings of the Association of Avian Veterinarians.
- Johnsgard, P. A. 1983. Cranes of the world. Indiana University Press, Bloomington. 257 pp.
- Kumamoto, A. 1984. Avian lymphocyte culture protocol. San Diego Zoo Research Department. 6 pp.
- Markin, Y., and V. Krever. 1992. On morphometric parameters of Common Crane sexual dimethyl. Paper presented at European Crane Working Group Meeting, Tallinn, Russia. 9 pp.
- McDonald, S. 1982. Surgical sexing of birds by laparoscopy. California Veterinarian 5/1982:16-22.
- McDonald, S. 1986. Surgical sexing. Bird Talk, July:26-32.
- Murata, K., T. Suzuki, M. Yosufuku, and W. Yoshitake. 1988. Sex determination in Manchurian Crane *Grus japonensis* using discriminant analysis. Journal of the Yamashina Institute of Ornithology 20:101-106.
- Nesbitt, S. A. 1975. Voice maturity in Florida Sandhill Cranes. Florida Field Naturalist 3:19.
- Rasch, E. M. 1976. Use of deoxyribonucleic acid-Feulgen cytophotometry for sex identification in juvenile cranes (Aves: Gruidae). Journal of Histochemistry and Cytochemistry 24:607.
- Rasch, E. M. 1977. Further studies of genome size and sex identification in endangered species of cranes (Aves: Gruidae). Journal of Histochemistry and Cytochemistry 25:242.
- Rasch, E. M., and P. J. Kurtin. 1976. Sex identification of Sandhill Cranes from karyotype analysis. Pages 309-316 in J. C. Lewis, editor. Proceedings International Crane Workshop. Oklahoma State University Publishing and Printing, Stillwater.
- Sasaki, M., T. Ikeuchi, and S. Makino. 1968. A feather pulp culture technique for avian chromosomes, with notes on the chromosomes of the Peafowl and the Ostrich. Experientia 24:1292-1293.
- Sasaki, M., and N. Takagi. 1981. Chromosomes in Gruiformes, with notes on the chromosomal diagnosis of avian sex. Pages 19-23 in J. C. Lewis and H. Masatomi, editors. Crane research around the world. International Crane Foundation, Baraboo, Wis.
- Song, J. 1991. A study on the amount of food intake and its main nutritional components for the White-naped Crane. Pages 161-163 in J. T. Harris, editor. Proceedings 1987 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Stavy, M., D. Gilbert, and R. D. Martin. 1979. Routine determination of sex in monomorphic bird species using faecal steroid analysis. International Zoo Yearbook 14:209-214.
- Swengel, S. 1992. Sexual size dimorphism and size indices of six species of captive cranes at the International Crane Foundation. Proceedings North American Crane Workshop 6:151-158.
- Tacha, T. C. 1987. Foraging and maintenance behaviors of Sandhill Cranes. Pages 93-105 in J. C. Lewis, editor. Proceedings 1985 Crane Workshop. Platte River Whooping Crane Habitat Maintenance Trust and U.S. Fish and Wildlife Service, Grand Island, Nebr.
- Tacha, T. C., and J. C. Lewis. 1979. Sex determination of Sandhill Cranes by cloacal examination. Pages 81-83 in J. C. Lewis, editor. Proceedings 1978 Crane Workshop. National Audubon Society. Colorado State University Printing Service, Fort Collins.
- Takagi, N., M. Itoh, and M. Sasaki. 1972. Chromosome studies in four species of Ratitae (Aves). Chromosoma (Berlin) 36:281-291.
- Takagi, N., and M. Sasaki. 1974. A phylogenetic study of bird karyotypes. Chromosoma 46:91-120.
- Walkinshaw, L. H. 1973. Cranes of the world. Winchester Press, New York. 370 pp.





Special Techniques, Part D: Reintroduction Techniques

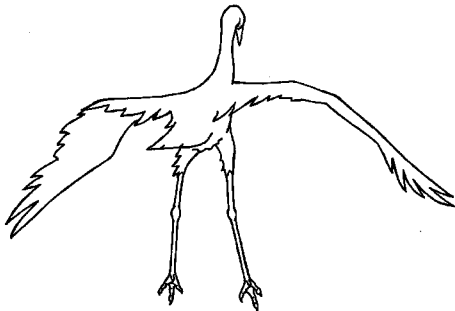


Meenakshi Nagendran, Richard P. Urbanek and David H. Ellis

The reintroduction of animals to augment remnant populations or establish new populations is sometimes essential to the preservation of threatened or endangered species. Release stock may be translocated (wild) adults or young, animals reared in captivity, or eggs from wild or captive flocks. Reintroduction techniques for fledged cranes were described by Konrad (1976), Derrickson and Carpenter (1983), Horwich (1986, 1989), Bizeau et al. (1987), Ellis et al. (1992a), Urbanek and Bookhout (1992), and Horwich et al. (1993). The only sizable use of eggs to start a wild crane population involved placing Whooping Crane eggs in Sandhill Crane nests at Grays Lake National Wildlife Refuge (NWR) (Drewien et al. 1989 unpubl).

Captive-reared cranes must be conditioned for survival in the wild. Herein, we discuss variations of **two basic rearing methods** that have been used to rear cranes in captivity for reintroduction. In **parent-rearing**, or surrogate parent-rearing, an egg or chick is placed in a pen with a pair of cranes. **Hand-rearing** involves either costumed humans aided by mounted crane heads or puppets, crane vocalizations, and other imprinting aids, or uncostumed humans using some imprinting aids.

Husbandry aspects of both hand-rearing and parent-rearing are detailed in Chapter 5. Adaptations of these techniques to rear birds for release are discussed here. A summary of previous release attempts is tabulated in Table 11D.1.



Release Methods

Abrupt Releases

Release of captive-reared cranes without acclimating them to the release site (herein termed abrupt releases; Table 11D.1) has consistently resulted in high mortality. The first release of sizable numbers of captive-reared cranes occurred in 1971, when 14 hand-reared Florida Sandhill Cranes were released in south-central Florida without acclimation (Nesbitt 1979). None of the 14 integrated into the wild flock, and within a few months all had died of exposure, starvation, or accident. A single parent-reared crane released in northern Florida, however, survived 3 years. Following the experiment with hand-reared cranes in Florida, abrupt releases of parent-reared Greater Sandhill Cranes were attempted at Grays Lake NWR in 1976 (n=1) and 1980 (n=11) (Drewien et al. 1982). Of seven young that survived to migrate south in 1980, none reappeared at Grays Lake the following spring. In 1984, 21 Greater Sandhill Cranes were released at Grays Lake after being held in a small pen on site for 4-6 days; only 9 (43%) survived to migrate (Bizeau et al. 1987).

Gentle Releases

In a gentle release, the cranes are held at the release site for two or more weeks, fed at the release site following release, and allowed to slowly acclimate to the release environment. Since 1981, more than a dozen gentle releases have been made using parent-reared cranes from Patuxent. In a nonmigratory situation, 15 of 27 (56%) Florida Sandhill Cranes survived their first winter (Nesbitt 1988). Annual survival rates around 70% have been achieved in Patuxent's extensive release program with Mississippi Sandhill Cranes (Ellis et al. 1992a).

TABLE 11D.1

A comparison of survival of captive-reared Sandhill Cranes after release to the wild.

Rearing		Release				Survival ¹		Reference
Method ²	Location	No. ³	Location	Year	Method ⁴	No.	%	
Parent	Patuxent	12 ⁵	Idaho	1976, 1980	Abrupt	1	8	Drewien et al. 1982
Parent	Patuxent	1	Florida	1976	Abrupt	1	100	Nesbitt 1979
Parent	Patuxent	21 ⁶	Idaho	1984	Abrupt	4	19	Bizeau et al. 1987
Parent	Patuxent	27 ⁷	Florida	1986, 1987	Gentle	15	56	Nesbitt 1988
Parent	Patuxent	10	Florida	1991	Gentle	3	30	Nesbitt ⁸
Parent	Patuxent	103 ⁹	Mississippi	1980-1993	Gentle	69	67	Ellis et al. 1992a
Hand	Patuxent	17 ¹⁰	Florida	1971-1977	Abrupt	0	0	Nesbitt 1979
Hand-isolation	On site	1	Oregon	n.d. ¹¹	Acclimation	1	100	Hyde 1968:165-168
Hand-isolation	On site	17	British Columbia	1981	Acclimation	7-9	41	Leach 1987
Hand-isolation	On site	1	Michigan	1981	Acclimation	1	100	Isham ¹²
Hand-isolation	Release Site	2	Wisconsin	1982	Gentle or Acclimation	1	50	Archibald and Archibald 1993
Costume	ICF or On site	5	Wisconsin	1985	Gentle	4	80	Horwich 1989
Costume	On site	7	Texas	1988	Gentle or Acclimation	2	29	Nagendran 1992a
Costume	On site	16	Michigan	1988	Acclimation	15	94	Urbanek and Bookhout 1992
Costume	Patuxent	28	Mississippi	1989-1991	Gentle	26	93	Ellis et al. 1992a
Costume	On site	12	Michigan	1989	Acclimation	9	75	Urbanek and Bookhout 1992
Costume	On site	9	Michigan	1990	Acclimation	8	89	Urbanek and Bookhout 1992

¹ Survival is credited if alive and free after migrating south (British Columbia, Oregon, Idaho) or after one year (all other studies).

² Hand refers to conventional hand-rearing with little effort to control exposure to humans. Hand-isolation refers to controlled exposure to humans, but no use of a costume and some exposure to puppets or other crane imprinting birds or models. Costume refers to rearing by costumed caretakers with puppets, tape recorded brood calls, live crane imprinting models, etc.

³ All cranes released were juveniles (less than 1 year old) except as indicated in footnotes.

⁴ Abrupt releases involve rearing elsewhere and releasing the birds less than two weeks after arrival at the release site. Gentle releases involve rearing at a propagation site, but holding the birds at the release site two or more weeks and providing food for them after release. Acclimated releases involve rearing colts all, or in part, at the release site.

⁵ One yearling released abruptly in August 1976; five yearlings, two 2-year-olds, and four 3-year-olds released abruptly 18 June 1980.

⁶ Nineteen yearlings and two 2-year-olds held on site in small, roofed pen for 4-6 days before release, 19 June to 3 July 1984.

⁷ Eleven juveniles and 4 yearlings released 4 April 1986 and 12 juveniles released 2 January 1987 after being brailed and held for 4-6 weeks in large, open release pen.

⁸ Shipped to Gainesville February 1991. Held there until 28 October when moved to release pen at Three Lakes Wildlife Management Area. Brails removed 25 November 1991 (S. A. Nesbitt, Florida Game and Fresh Water Fish Commission, Gainesville, Florida, personal communication).⁹

⁹ Included one yearling released winter 1980-1981.

¹⁰ In addition to the 14 juveniles released in 1971, 3 older cranes (some as old as 4 years) were released 1974-1977.

¹¹ Probably 1950's. Hyde does not give a date.

¹² M. Isham, Bellevue, Michigan, personal communication.

Captive Rearing by Crane Pairs

Chicks reared by their own parents, or by surrogates of the same species, show proper sexual imprinting. Parent-reared cranes learn some foraging skills from their parents. If their parents are shy, the chicks are also naturally wary of humans and do not need human avoidance conditioning during rearing. Because some pairs of captive cranes prove unsuitable for both incubating eggs and rearing chicks, approximately two pairs are needed for each endangered chick.

Parent-reared chicks are formed into cohorts within a few days of fledging. This socialization process is best done in large flight netted pens (15- to 30-m long). After 4-6 weeks in these pens, colts are moved to their release site. Release pens are unnetted and usually at least a hectare in size so the cranes remain wing brailed during the month-long acclimation period. This lengthy process has been very successful in nonmigratory situations, but is too protracted to be used with migratory birds.

Two-thirds (41 of 66) of the parent-reared birds released at the Mississippi Sandhill Crane NWR from 1981 through 1989 survived for at least one year (McMillen et al. 1987; Zwank and Wilson 1987; Ellis et al. 1992a). All Mississippi birds surviving more than a few months have successfully integrated into the wild flock. Parent-reared cranes have bred after release with conspecific wild cranes and raised chicks successfully (Zwank and Wilson 1987; Ellis et al. 1992a).

Rearing by Wild and Tame Crane Parents

In Hokkaido, Japan, flightless male Red-crowned Cranes have lured wild females into their enclosures (Konrad 1976). The resulting pairs produced chicks that fledged into the wild flock. Occasionally, captive Sandhill Cranes have lured in wild mates (Hyde 1957; G. W. Archibald, ICF, personal communication).

A variation of this technique was tried twice with cross-fostered Whooping Cranes at Grays Lake (Drewien et al. 1989 unpubl.). Because adult male Whooping Cranes in this experimental flock failed to return to the marsh with female mates, several attempts were made to capture and translocate adult females that had dispersed in the surrounding states. When these attempts failed to produce viable pairs,

two hand-reared females (one each in 1981 and 1989) were sent from Patuxent and introduced to the adult males. Both females were courted and while it appeared that bonds were forming, neither attempt resulted in eggs or in pairs that migrated together (Derrickson and Carpenter 1983; Drewien et al. 1989 unpubl.).

Another variation of pairing captive-reared and wild cranes occurred in northern China: White-naped and Red-crowned Crane chicks were hand-reared and gently released in the marshes at Zhalong (Jie et al. 1989 unpubl.). The birds were then returned to captivity to prevent their loss in the coming winter. In subsequent years, these semi-domestic birds paired with each other or with wild mates and nested in the marshes near their natal area. Young were generally kept in captivity with their parents the first winter. These birds joined the wild birds during the following year. Offspring resulting from these tame-wild matings were reportedly much more tolerant of human approach and consequently better able to live in a human-dominated environment.

A similar experiment is being conducted with Red-crowned and White-naped Cranes at the Khinganski Nature Reserve in northeastern Russia (Y. Andronov, Arkhara 676740, Amurfkaja Region, Russia, personal communication). The purpose of this experiment is to modify the behavior of the wild population to make them more tolerant of the rapidly increasing human population. Suitable nesting and rearing habitat is still available, but the wild populations are avoiding areas with human activity. To date, semi-wild birds from captivity have successfully attracted wild mates, and wild pairs are also increasing their usage of disturbed areas in response to the presence of semi-wild birds.

Release of Cross-fostered Cranes

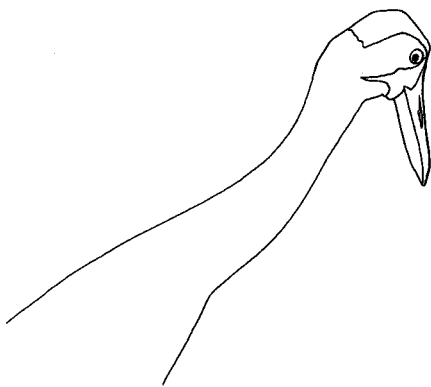
Between 1975 and 1988, an experiment was conducted to create a migratory flock of Whooping Cranes at Grays Lake NWR. Sandhill Crane eggs were exchanged for Whooping Crane eggs (289) from Patuxent and Wood Buffalo National Park in Canada. Because of high mortality and because none of these cross-fostered Whooping Cranes (reared by Sandhill Cranes) paired and bred successfully, the experiment was discontinued.

Advantages of this technique are that chicks reared by wild cranes are imprinted on cranes, fear humans, and display near normal crane behavior. Likewise, these chicks learn survival skills from their parents.

The primary disadvantage is that cross-fostered young appear to be sexually imprinted on their foster species. A cross-fostering study conducted in Japan (Nakayama 1970) and a more recent pilot study at ICF (Mahan and Simmers 1992) indicate that cross-fostered chicks prefer their cross-fostered parent species socially and sexually. Although cross-fostered young have been bred through intensive management in captivity, this appears to be impractical in the wild.

Hand-rearing by Uncostumed Humans

Hand-rearing is a more efficient way to rear large numbers of crane chicks for captivity (see Chapter 5). Survival rates are generally high for hand-rearing because disease can be more closely managed than for parent-rearing. Hand-reared chicks are usually more comfortable in captivity and will breed well. Conventional hand-rearing has not, however, proven successful for reintroduction purposes. Crane chicks reared in close contact with humans lack fear of humans, some prefer humans to cranes when under stress, and they adapt poorly in the wild (Nesbitt 1979; Table 11D.1). A modification of human-contact hand-rearing can be called **screen-rearing**. In this method the chicks are fed by humans who hide behind a screen so as to not be visible to the chick. However, the chicks have frequent exposure to humans during weighing, pen cleaning, and when medication is administered. Although these chicks are somewhat more wary of humans than chicks reared without the screen, they should probably not be considered release candidates. Like conventional hand-rearing, screen-rearing is suitable for birds to be retained in captivity.



Hand-rearing with Costumes

Advantages and Disadvantages

Using costume-rearing, it is possible to hand rear a crane chick yet imprint it on its own species. This technique increases the number of young cranes available for release each season, whereas with parent-rearing, each pair of cranes can only successfully rear one or rarely two young each season. Furthermore, costume-rearing can be accomplished at the release site thus promoting imprinting of chicks on their natal area, a process that is probably essential for migratory flocks.

Costume-reared cranes have experienced high post-release survival both in migratory and nonmigratory situations (Horwich 1986, 1989; Ellis et al. 1992a; Urbanek and Bookhout 1992; Archibald and Archibald 1993; Horwich et al. 1993; Table 11D.1). Early in the development of this rearing and release method, some of the cranes required assistance (they were moved to staging areas) to initiate proper migration. This problem was important when the cohort size was large, but by reducing the release cohort size to 4-5 birds, this problem has been solved. Overall, more than 80% of costume-reared cranes that were released using the gentle release technique successfully migrated and returned to the general release area. In a nonmigratory situation, costume-reared Mississippi Sandhill Cranes have experienced ca 90% survival to one year.

Costume-rearing requires a greater time investment than standard hand-rearing. Routine chores proceed more slowly and in less comfort. Exercising chicks while in costume demands more caution. The discomfort of the costume increases on hot summer days. The caretaker must also exercise greater care when tending chicks because the costume restricts vision.

Techniques

In the costume-rearing technique, the caretakers dress in an amorphous, **hooded costume**. The main purpose of the costume (Fig. 11D.1) is to conceal the human form so the chick will not become habituated to humans and seek human company after release. The costume covers a person from head to knee. The hood hides the human face behind a mask of cheese cloth, nylon window screen, or dark camouflage netting.



Fig. 11D.1. *Costumed caretaker with Siberian Crane chicks, northwestern Siberia, 1991.* Photo David H. Ellis

Hands are hidden within the arm pieces of the costume. The color of the costume should be the color of the adult crane. Horwich (1986) attached crane feathers to the arm pieces, although in later studies no feathers were used.

Although the crane costume bears little resemblance to a crane, the **crane puppet head**, which is manipulated by hand, should be realistic (Fig. 5.8). The puppet head and neck can be made from soft cloth (e.g., terry cloth or “fake fur”). Glass or plastic eyes can be obtained from a taxidermy supply house or craft store. Metal or wooden mandibles glued to a spring-loaded clothespin or a scissor base form a functional beak (Fig. 11D.2). The costumed parent feeds chicks insects as would an adult crane. The puppet can also be used to dig up worms, roots, and tubers. Alternately, **taxidermic crane heads** (Fig. 11D.3) or cast plastic heads may be used. These heads are more realistic and may help with imprinting cues. Because we have never made the beak on these heads to open and close, food items must be pierced, scooped up, or otherwise secured to the bill and offered to the chick. Chicks are very interactive with the head, be it puppet, mount, or casting.

Because parents brood call to their eggs during incubation, to promote imprinting, we play tape-recorded vocalizations of conspecific cranes to each egg for 5-15 min periods about 4 times per day beginning around 3 weeks, but at least as soon as the chick breaks into the air cell (ca day 27 of incubation). During the latter part of incubation, the brood call stimulates chick vocalization and movement, and may



Fig. 11D.2. *Rob Horwich, who developed costume rearing as an operational technique, demonstrates the puppet head. Advantages of this design are the ability to grasp with the bill and to wash the terry cloth cape.* Photo David H. Ellis



Fig. 11D.3. *Taxidermic heads made from Sandhill Cranes. The upper head is cast in plastic and will be fitted with a “fake fur” covering. The lower head has been altered (i.e., facial stripe area plucked and dyed, and elsewhere gray feathers have been bleached with human hair dye [Ellis and Ellis 1975]) to resemble a Whooping Crane.* Photo David H. Ellis

encourage the chick through the hatching process. The costumed parent can also use a portable audio tape recorder (suspended beneath the costume) to simulate the crane brood call. At Patuxent, the costumed caretaker vocally mimics the cranes' brood call so the tape recorders are used only before hatching.

The tape recorder can be used to produce not only the easily imitated Brood-call, but also Unison-calls, Guard-calls, and Alarm-calls for use in each appropriate context. For example, an Alarm-call can be played when a raptor flies overhead or when a chick is "attacked" by a non-costumed human. By far the most important call for rearing purposes is the Brood-call. Crane parents Brood-call to the eggs, and once the chick hatches its response to this call is remarkable. The Brood-call encourages the chick to approach its parent or the costumed human and feed, drink, or follow as indicated by the behavior of the parent.

Hand-rearing, especially costume-rearing, requires strict adherence to a rearing protocol. When the chicks hatch, the costumed parent feeds them with the puppet and teaches them how to feed on prepared food from a bowl. Teaching them to feed normally requires 10 to 15 min hourly for the first 3 to 5 days, but sessions may require up to 30 min and may continue for 2 weeks before the chicks are feeding independently.

Exposing chicks to an adult, conspecific crane used as an imprinting model promotes proper sexual imprinting (see Chapter 5) and may teach other behavior patterns. Imprinting probably involves several stages in cranes. The presence of a conspecific, live adult during these critical times increases the chances of proper imprinting. When possible, we also provide a taxidermic crane in brooding posture to each chick at hatching. The brooder model can contain a heat pad or be positioned beneath the heat lamp to provide warmth (Fig. 11D.4).

Because the chick's interactions with its live imprinting model are limited by the barrier, it may be important for chicks to interact with other members of their release cohort. Exercising chicks together from an early age under careful supervision may reinforce imprinting on their own species and it does allow formation of a release cohort in time for migration. In migratory situations, we also recommend that release cohorts contain no more than 2-5 birds (Urbanek and Bookhout 1992). Release cohorts larger than 5 colts are less prone to integrate into the wild flock. Cohorts can exceed 15 colts in nonmigratory situations.



Fig. 11D.4. *Taxidermic brooder model with heat lamp overhead used for costume rearing Mississippi Sandhill Cranes.*

Photo David H. Ellis

To make good release candidates, hand-reared chicks must fear and avoid humans. Chicks reared for release should be handled as little as possible. Handling is a uniquely human-like rather than crane-like activity, and excessive handling, especially of older chicks, may result in a bird that is less wary of humans. As a chick grows older, the necessity for handling decreases, and handling becomes an increasingly stressful experience.

Because the costume impairs vision, chicks must be handled with extreme caution and in a well-illuminated area. Routine health checks such as daily general physical examination, including eyes, nares, vent, weights, and preventive medication, should be done in costume, but health checks involving more negative experiences, such as drawing blood, should be done without a costume. At Patuxent, chicks are sometimes hooded during physical examination so that costumed caretakers or veterinarians may remove their own hoods and perform the examination without visual obstruction.

Chicks instinctively feed on insects and other small moving organisms soon after hatching. By 2 months of age, they also probe and feed on roots and tubers. Using the costume and head to introduce them to foods naturally occurring in the wild should help them find natural foods after release. If more than 5 young chicks (<6 weeks old) are taken afield at any given time, a second costumed parent will be necessary to prevent aggression related injuries. Most cranes adapt well to new food crops. Corn and sorghum can

be added to the pelleted diet when the chick is about 2 months old. Chicks trained to eat food grains and other foods found in their environment may adapt more quickly to release conditions.

Daily cleaning and feeding chores should be done in costume and in silence unless the rearing facility allows for chicks to be locked outside. If adequate costumed parents are available to lead and walk the chicks away from the facility, indoor chores may also be performed without a costume. Cleaning activities go much faster without a costume.

Human and predator avoidance conditioning is used to teach costume-reared chicks to develop an aversion to humans and mammalian predators. At ICF, chicks are at least 45 days old before they are subjected to their first “mock” attack by non-costumed humans. Younger chicks might injure themselves during such an “attack.” Under field rearing conditions (i.e., where chicks are reared and released in the same remote area), it may be advisable to wait until the chicks can fly before they are intentionally frightened. Flighted chicks fly away after being frightened, but return a short time later to search for the costumed parent. Prefledged chicks, however, run and hide where the costumed parents might not be able to readily find and protect them.

At Patuxent, chicks are subjected to human avoidance conditioning beginning at about 20 days of age. Chicks and imprinting model adults are locked outdoors while an uncostumed human runs through the alley next to the chick pens and produces loud noises (e.g., raking a stick along the chain link pen wall, banging pots, and yelling). A hidden human simultaneously plays a tape recording of a crane Guard-call. Imprinting models (adults) often Guard-call. Chicks that show little or no reaction are physically jostled, then released.

This training is scheduled at 2-week intervals, but only certain “non-wary” chicks are repeatedly exposed. After the chicks are pooled in flight pens, mock attacks are staged wherein one or two humans chase after the chicks in their pen. Attack bouts cease after all chicks are wary.

Crane chicks sometimes appear to be instinctively wary of avian predators, but it may be helpful to use tape recorded adult Guard- (or Alarm-) Calls to instill or reinforce this fear. Under some conditions, it may be advisable to teach a fear of canids by using a trained dog to chase fledged birds. After release, young cranes quickly learn from wild cranes to avoid predators.

Facilities

For costume-rearing, the rearing site must be temporally or geographically isolated from human activity. Rearing crane chicks under **field conditions** near their future release site may help imprint the chicks on their natal area and enables the chicks to learn foraging and survival skills. Males tend to be more philopatric than females, returning to their natal area after the first winter (Drewien et al. 1989 unpubl.). If frequent contact is maintained with the release birds, the costumed human can approach the birds several months after integration into the wild and can readily recapture cranes if it is necessary to transfer them to another area, to replace a transmitter, or for some other purpose.

Cranes raised at the release site are returned to predator-proof pens at night. Solar powered electric fences can be used, and the constant presence of the costumed parent also provides good protection. Cranes reared in the field have demonstrated high survival rates after release into a migratory situation (Archibald and Archibald 1993; Urbanek and Bookhout 1992; Table 11D.1). With care, high chick survival rates can be achieved even in a wilderness environment.

In the absence of parents to brood chicks, shelters must protect chicks from predators and inclement weather. Urbanek (1990 unpubl.) described an economical design for a costume-rearing facility in the field (Fig. 11D.5). Heat lamps provide heat for chicks until they are feathered (see Chapter 5). Hot water bottles can be used in locations lacking power supply (Nagendran 1991 unpubl., 1992b unpubl.).



Fig. 11D.5. *The costume rearing facility at Seney National Wildlife Refuge, Michigan.*

Photo Richard P. Urbanek

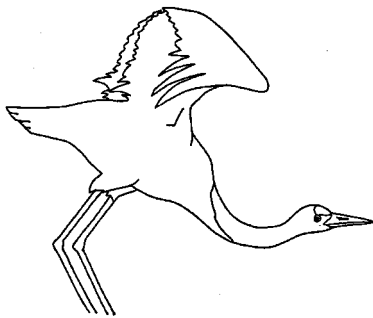
Release Procedures

Cohort Formation

When costume-reared birds are not reared at the release site and are introduced in a nonmigratory situation as in Patuxent's program for the Mississippi Sandhill Crane, the chicks are kept at the chick rearing facility until they are 55–60 days old. Temporary juvenile cohorts are then formed in netted community pens. Slower maturing cranes may require 70–100 days before being formed into release cohorts. Once formed, each cohort should be penned adjacent to a small group of parent-reared (i.e., wild-acting), conspecific, adult "socialization models" to encourage fear and avoidance of humans.

Parent-reared chicks remain with their foster parents somewhat longer. If the parent-rearing pens are without nets, the flight capability of parent-reared chicks should be closely monitored after about 55 days. When capable of flight, the chicks should be brailed (Ellis and Dein 1991; see Chapter 11E) until about 140 days of age. When the colts are removed from their foster parent's pen, they are assigned to a release cohort, and released in a flight netted community pen.

When costume-reared chicks will be released into a migratory flock, cohort members may be assigned early in rearing, and reared together. By contrast, at Seney NWR, where the entire rearing and release process takes place on site, the cohorts are intentionally broken up and rearranged at the time of release to decrease familiarity among cohort members and to encourage association with wild birds. When rearing is carried out at an established rearing center, but where chicks must migrate, they are moved to the release area two or more weeks prior to migration to promote imprinting on the release site. These chicks are not brailed.



Release Site Selections

When chicks are ready for release to the wild they are moved to a release pen. The release pens at Mississippi Sandhill Crane NWR are 1.2 to 2.0 ha in size with a fence 2.4 m in height. Fences are buried 0.3 m below ground and an electrical wire placed near the top to safeguard against predators. Feeders provide supplemental food within the pens. Natural foraging habitat in the immediate area further encourages the wild cranes to remain in the release area and encourages the chicks to feed on natural foods during release. Release birds typically learn wariness of predators from wild cranes. Providing a pool in the release pen encourages the chicks to roost in water and reduces their risk of predation. When possible, roosting and foraging habitat should be provided within the release pen. Cranes need to remain protected in the pen until they acclimate to the area, associate with wild cranes, and establish site tenacity.

Whenever possible, the release site should be in an area occupied by wild cranes. Survival of released chicks improves when they feed and roost with wild cranes. In a migratory situation, the released chicks must learn the migration route through their association with these wild birds. Baiting wild cranes into the release area will encourage mingling. When the objective is to augment or establish a migratory population, studies so far suggest that release should occur on the northern breeding grounds (Urbanek and Bookhout 1992; Horwich et al. 1993) rather than on the wintering grounds (Nagendran 1992a).

Pre-release Procedures

Birds should be transported during cool weather and as quickly and efficiently as possible. We use ventilated boxes with a floor covered with wood shavings underlaid with carpet to absorb wastes and prevent slipping. To transport a Sandhill Crane, we use a box 60 x 90 cm and 120 cm high. Box size should be adjusted for each species. The chick should be able to turn around and stand upright. Commercial airline carriers may require boxes constructed of wood, but cardboard wardrobe boxes are excellent.

Before release, chicks should be **color-banded** and may be equipped with **radio transmitters** (Fig. 11D.6). Radio transmitters less than 50 g can be mounted on leg bands and fitted above the hock. Those greater than 60 g are mounted as a backpack. A good material for the backpack harness is Teflon ribbon (Ellis et al.



Fig. 11D.6. *Radio transmitter on leg band.*

Photo Stephen A. Nesbitt

1992b; Olsen et al. 1992). Plastic color bands should be placed above the hock (Fig. 2.11) and can range from 1.9-7.5 cm in height. A monel or aluminum band may be placed above the hock or just above the foot.

Release Techniques

Nonmigratory cranes at the Mississippi Sandhill Crane NWR are brailled on one wing (Ellis and Dein 1991) for 4 weeks. Within a few days of removing the brails, the cranes begin to fly in and out of their pen. Chicks normally roost in the pen every evening for the first 10 days. This practice decreases their risk of predation. Within 2-4 months, the released cranes wear themselves from the food provided in the release pens.

Migratory cranes must be ready to leave a few weeks after fledging. Brailing, which sometimes results in temporary wrist stiffness and a degree of flight impairment, should not be used for migrants. At Seney NWR and in Siberia, we trained our chicks to return to a scarecrow-like costumed dummy (Fig. 11D.7). The costumed dummy was then used to introduce our chicks to wild cranes. At Seney, wild cranes, previously baited into the pen, quickly acclimated to the costumed dummy, and the dummy attracted the release cranes to the pen until they began following, feeding with, and roosting with wild birds outside the pen, after which time the dummy was removed. Providing corn, an important food of migrating cranes, attracts large numbers of wild cranes into the pen, facilitating integration into the wild. Because Seney cranes have been reared on-site, they adapt quickly to wild conditions and integrate with wild birds within a few weeks of release.

During 1988-90, 38 Sandhill Crane chicks were reared and released on Seney NWR. All survived the 18 to 60 days intervening between release and departure on their first migration (Urbanek and Bookhout 1992). The major problem at Seney NWR appears when too many costume-reared chicks are released in one area with too few wild cranes. Chicks in a release cohort prefer to remain in the company of each other after release. They create their own flock identity and are less likely to follow wild birds. This problem can be solved by dividing the release cohort into groups of no more than four or five birds and then releasing each group at a different site. Ideally, a release pen should be available at each site. However, success is possible by first releasing all the subgroups in a single pen and then translocating the small subgroups to other sites after these birds have completed their acclimation to wild conditions. This procedure was used to induce all members of a cohort of nine Sandhill Crane chicks to begin migration correctly from Seney NWR in 1990 (Urbanek and Bookhout 1992). Through the years, a few birds failed to leave the release areas in Wisconsin and Michigan with the wild cranes. These birds were retrieved by the costumed parent, boxed, and transported to another crane stopover area for re-release (Urbanek and Bookhout 1992).

For releases that must occur in the subarctic, the earliest captive-produced eggs of the season should be used. The chicks that result will be older than the wild chicks, so the released chicks have an extra week or two to integrate with wild cranes before departure.



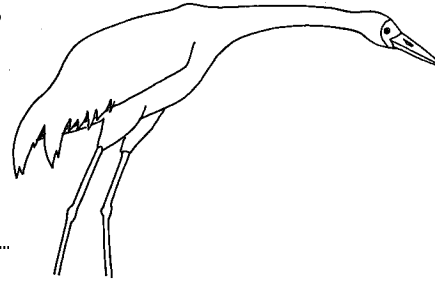
Fig. 11D.7. *A scarecrow-like costumed dummy is used to lure costume-reared chicks (here a Sandhill Crane) into a feeding area where they may be recaptured or encouraged to mix with wild cranes, Seney National Wildlife Refuge, Michigan.*

Photo Ted and Jean Reuther

Literature Cited

- Archibald, K., and G. Archibald. 1993. Releasing puppet-reared Sandhill Cranes into the wild: a progress report. Pages 251-254 *in* D. A. Wood, editor. Proceedings 1988 North American Crane Workshop. Florida Nongame Wildlife Program Technical Report #12.
- Bizeau, E. G., T. V. Schumacher, R. C. Drewien, and W. M. Brown. 1987. An experimental release of captive-reared Greater Sandhill Cranes. Pages 78-88 *in* J. C. Lewis, editor. Proceedings 1985 Crane Workshop. Platte River Whooping Crane Maintenance Trust, Grand Island, Nebr.
- Derrickson, S. R., and J. W. Carpenter. 1983. Behavioral management of captive cranes—factors influencing propagation and reintroduction. Pages 493-511 *in* G. W. Archibald and R. F. Pasquier, editors. Proceedings of the 1983 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Drewien, R. C., W. M. Brown, and E. G. Bizeau. 1989 unpubl. Whooping Crane cross-fostering experiment. Report to Whooping Crane Recovery Team by Wildlife Research Institute, University of Idaho, Moscow. 10 pp.
- Drewien, R. C., S. R. Derrickson, and E. G. Bizeau. 1982. Experimental release of captive parent-reared Greater Sandhill Cranes at Grays Lake Refuge, Idaho. Pages 99-111 *in* J. C. Lewis, editor. Proceedings 1981 Crane Workshop. National Audubon Society, Tavernier, Fla.
- Ellis, D. H., and F. J. Dein. 1991. Flight restraint techniques for captive cranes. Pages 447-451 *in* J. Harris, editor. Proceedings 1987 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Ellis, D. H., and C. H. Ellis. 1975. Color marking Golden Eagles with human hair dyes. *Journal of Wildlife Management* 39:445-447.
- Ellis, D. H., G. H. Olsen, G. F. Gee, J. M. Nicolich, K. E. O'Malley, M. Nagendran, S. G. Hereford, P. Range, W. T. Harper, R. P. Ingram, and D. G. Smith. 1992a. Techniques for rearing and releasing nonmigratory cranes: lessons from the Mississippi Sandhill Crane program. Proceedings North American Crane Workshop 6:135-141.
- Ellis, D. H., D. G. Smith, G. H. Olsen, M. R. Fuller, S. E. Landfried, H. Higuchi, and C. H. Vermillion. 1992b. Progress in satellite tracking cranes. Proceedings North American Crane Workshop 6:57-61.
- Horwich, R. H. 1986. Reintroduction of Sandhill Cranes to the wild. *ICF Bugle* 12:1, 4-5.
- Horwich, R. H. 1989. Use of surrogate parental models and age periods in a successful release of hand-reared Sandhill Cranes. *Zoo Biology* 8:379-390.
- Horwich, R.H., J. Wood, and R. Anderson. 1993. Release of Sandhill Crane chicks hand-reared with artificial stimuli. Pages *in* D. A. Wood, editor. Proceedings 1988 North American Crane Workshop. Florida Nongame Wildlife Program Technical Report #12.
- Hyde, D. O. 1957. Crane notes. *Blue Jay* 15:19-21.
- Hyde, D. O. 1968. *Sandy*. Dial Press, New York. 214 pp.
- Jie, X., L. Su, and X. Jiang. 1989 unpubl. A way of expediting the recovery of the population of the Red-crowned Crane. Report, Zhalong Nature Reserve, Qiqihar, China. 8 pp.
- Konrad, P. M. 1976. Potential for reintroduction of cranes into areas of former habitation. Pages 317-325 *in* Proceedings International Crane Workshop. Oklahoma State University, Stillwater.
- Leach, B. 1987. The Sandhill Crane recovery project in the Lower Fraser Valley, British Columbia. Pages 415-423 *in* G. W. Archibald and R. F. Pasquier, editors. Proceedings of the 1983 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Mahan, T. A., and B. S. Simmers. 1992. Social preference of four cross-foster reared Sandhill Cranes. Proceedings North American Crane Workshop 6:43-49.
- McMillen, J. L., D. H. Ellis, and D. G. Smith. 1987. The role of captive propagation in the recovery of the Mississippi Sandhill Crane. *Endangered Species Technical Bulletin* 12(5-6):6-8.
- Nagendran, M. 1991 unpubl. Isolation-rearing and release of Siberian Cranes. WWF Project: 4588-USSR Siberian Cranes. Final Project Report. 11 pp.
- Nagendran, M. 1992a. Winter release of isolation-reared greater Sandhill Cranes in south Texas. Proceedings North American Crane Workshop 6:131-134.
- Nagendran, M. 1992b unpubl. Isolation-rearing and release of Siberian Cranes. Project Report. 9 pp.
- Nakayama, T. 1970. Japonensis raised by foster parents. *Animals and Zoos* 22(2):42-43.
- Nesbitt, S. A. 1979. Notes on the suitability of captive-reared Sandhill Cranes for release into the wild. Pages 85-88 *in* J. C. Lewis, editor. Proceedings 1978 Crane Workshop. Colorado State University Printing Service, Fort Collins.
- Nesbitt, S. A. 1988. An evaluation of the Florida Sandhill Crane population of peninsular Florida and its potential to support a population of non-migratory Whooping Cranes. Florida Game and Fresh Water Fish Commission, Gainesville. 109 pp.
- Olsen, G. H., D. H. Ellis, S. E. Landfried, L. J. Miller, S. S. Klugman, M. R. Fuller, and C. H. Vermillion. 1992. Behavior of Sandhill Cranes harnessed with different satellite transmitters. Proceedings North American Crane Workshop 6:50-56.
- Urbanek, R. P. 1990 unpubl. Behavior and survival of captive-reared juvenile Sandhill Cranes introduced by gentle release into a migratory flock of Sandhill Cranes. Final Report., Ohio Cooperative Fish Wildlife Research Unit, Columbus. 98 pp.
- Urbanek, R. P., and T. A. Bookhout. 1992. Development of an isolation-rearing/gentle release procedure for reintroducing migratory cranes. Proceedings North American Crane Workshop 6:120-130.
- Zwank, P. J. and C. D. Wilson. 1987. Survival of captive, parent-reared Mississippi Sandhill Cranes released on a refuge. *Conservation Biology* 1:165-168.

Special Techniques, Part E: Flight Restraint



David H. Ellis and F. Joshua Dein

Many techniques are available for preventing escape of captive cranes. These include tenotomy, tenectomy, wing clipping, confinement under nets, amputation, brailing, and vane trimming (Ellis and Dein 1991). The advantages and limitations of each technique are presented.

Flight Restraint Methods

Techniques for birds include: (1) **limited amputation** (removal of a portion of the wing; the most common form is pinioning which involves removal of the hand) (Young 1948; Schwarte 1965; Sedgewick 1967; Williamson and Russell 1971; Robinson and Buzikowski 1975; Osinskij and Taran 1978; Madill 1981; Wallach and Boever 1983; Amand 1986); (2) **tenotomy** (severing the extensors of the hand) (Schroeder and Koch 1940; Miller 1973); (3) **tenectomy** (removal of a portion of the extensors of the hand) (Schwarte 1965; Sedgewick 1967; Miller 1973; Amand 1986); (4) **patagiotomy** (removal of the patagial membrane and apposition of the radius and humerus) (Sedgewick 1967; Mangili 1971; Robinson 1975; Madill 1981); (5) **functional ankylosis** (fixing the ulna, carpal, and metacarpal bones with stainless steel wire) (Sedgewick 1967); (6) **wing (feather) clipping** (cutting the distal portions of the primary and secondary feathers) (Young 1948; Schwarte 1965; Sedgewick 1967; Gandaland and Amand 1982; Amand 1986; Harrison and Harrison 1986); (7) **brailing** (binding on wing) (Schwarte 1965; Zwank and Derrickson 1981; Amand 1986); (8) **vane trimming**, which renders young cranes flightless from fledging until they can be safely wing clipped (when their quills are fully grown); and (9) confinement under **nets**.

Radical amputation of the wing also renders birds flightless, but is seldom used because captive birds are usually confined for propagation or display purposes, uses which presumably would be impaired by extensive mutilation. One less radical form of amputation,

pinioning (removal of all or a portion of the hand, wing below the wrists) of neonatal chicks, is routinely performed at the New York Zoological Society (Sheppard and Bruning 1983).

Recommended Methods

Patuxent and ICF do not use or recommend radical amputation, tenectomy, patagiotomy, or ankylosis. We use each of the techniques discussed below.

Netted Pens

Nets are used for birds that are designated for release or for full-winged captive breeders. We recommend using nylon coverings for chain link pens. These pens are constructed typically of 2.4-m (8-ft) tall chain link. Nets are supported by 1-cm (0.375-in) plastic-coated steel cables crossing the pens at approximately 6.1-m (20-ft) intervals. In some pens, interior poles are used to support netting.

We use 5.1-cm (2-in) mesh woven-nylon nets, and recommend this **mesh size** as a maximum. Birds held experimentally under nets with larger mesh have been occasionally snared in the net and held suspended by one or both wrists (G. W. Archibald and S. R. Swengel, ICF, personal communication). With 5.1-cm (2-in) mesh, birds which spring up against the nets occasionally pass their heads through the mesh and are momentarily held suspended. Sandhill Cranes pull free under their own weight as do most Whooping Cranes. Rarely, however, a Whooping Crane is held suspended until pulled free by a caretaker. We have incurred no known injuries from these incidents, but believe that this problem can be avoided using a slightly smaller mesh for large cranes.

Netted pens allow birds to be full-winged and therefore presumably better able to balance during copulation. Chances of reproducing naturally (without artificial insemination) are thereby increased.

In cooler environments where snow or ice storms are likely, netted pens, unless heavily braced, can collapse. We recommend either permanent interior support posts or a sufficiently large work force with sufficient temporary vertical support posts to maintain netted pens during snow or ice.

Tenotomy

A veterinarian or other trained person uses a thermocautery instrument (Fig. 11E.1) to sever the *tendo longa* and destroy the synovial capsule of the wrist (*junctura carpi*). The operation should be performed with a local anesthetic. We infiltrate the site with 2-3 mL of 2% lidocaine HCl, wait 5 min, and freeze the skin surface with an ethyl chloride spray immediately before surgery. Young birds are typically tenotomized in the fall of their first year. After tenotomy, the wing is taped tightly folded for six weeks to promote ankylosis. A successful tenotomy allows for only limited wing extension capability (Fig. 11E.2).

Some tenotomized cranes are, in a strong wind, still capable of limited flight. To prevent the escape of such birds, we clip the primaries of the tenotomized wing after each molt.

Wing Clipping

Wing clipping is used for birds that will be held flightless for at least three years (the normal maximum amount of time required between molts), but may be designated for flight thereafter. Two variations of clipping are available. Either all 10 primaries and most or all of the distal secondaries from one wing are cut with scissors (Fig. 11E.3) or all primaries except the three



Fig. 11E.1. A tenotomy is performed on the folded wing with a thermocautery instrument.

Photo David H. Ellis



Fig. 11E.2. Wing extension capability in a properly tenotomized bird (Jane Nicolich with Greater Sandhill Crane).

See also Fig. 3.6.

Photo David H. Ellis

most distal and all of the secondaries are cut. Birds with exceptional escape capabilities are wing clipped more extensively. Typically, each rachis is cut about 2.5 cm (1 in) from its point of emergence from the integument.

When clipping, **special care** is taken to avoid cutting any feather that is still growing. Profuse and prolonged bleeding from the quill occurs if this precaution is not taken. To stop bleeding, the feather should be pulled from the follicle. To prevent cutting the rachis too soon, the wing is spread and the underside of the wing is inspected to identify blood quills. Feathers that are still growing are temporarily vane trimmed, as described below, and later clipped when feathers are hard-penned (i.e., fully grown and free of blood in the calamus).

Vane Trimming

Vane trimming temporarily grounds birds while their flight feathers (primaries and secondaries) are growing. Once the flight feathers are hard-penned, the rachises are clipped (Fig. 11E.3).

In this process (Fig. 11E.4), a portion of the vanes of the primaries and the distal three to six secondaries of



Fig. 11E.3. *A wing-clipped wing.*

Photo David H. Ellis



Fig. 11E.4. *A vane-trimmed wing (see also Fig. 1.16).*

Photo David H. Ellis

one wing are trimmed with scissors. The rachis and the feather tip is left untrimmed to prevent birds in social groups from striking pen mates with sharp rachis tips. As illustrated, the outer vane of the five most distal primaries is left intact to prevent breakage of the rachises. Vanes are usually trimmed when birds are 60-70 days old.

Brailing

This technique is used for temporarily restraining fledglings, flighted adults, and birds during shipment. Shipped birds are usually brailed on one wing. For birds brailed long term, brails are changed to the opposite wing at regular intervals (usually every two weeks) to prevent stiffening of the immobilized wing. Epperson (1982 *unpubl.*) found significant but reversible impairment in wing extension capability in birds brailed only two weeks. Birds typically regained full flight capability within 1 to 2 weeks of brail removal.

The procedure requires a brail and a riveting device. The brail is a narrow band of flexible plastic 2 cm x 37.8 cm (0.75 in x 15 in) and about 1 mm thick. It

is pre-drilled with holes about 1 cm apart. We use a commercially available rivet gun to secure the brail. Leather straps and other riveting or sewing devices can be readily substituted as long as the conditions for proper fit, described next, are met.

When brailing a crane, one caretaker holds the bird immobile while a second inspects the wing to be certain that less than 4 cm of the rachis of each primary is still filled with blood. If the blood-filled zone in some of the quills is more extensive than 4 cm, brailing is postponed to avoid damage to growing feathers. If the bird is ready to be brailed, the brail is inserted between the bases of the third and fourth most distal primaries (numbers 7 and 8), and the strap is formed into a loose loop over the patagium. With the wing folded, the rivet, with one washer on the shaft, is placed through two of the pre-drilled holes in the brail. By probing upward with the free hand, a path is opened for the rivet to pass between the feathers and through a third hole in the brail on the underside of the wing. The loops above and below the rivet should be about equal size. The washer is placed on the rivet and fastened. The trailing end of the brail should point downward and be trimmed to within 3 cm of the rivet. To remove a brail, cut the upper loop (Fig. 11E.5) and slide the brail off the primaries. The final positions of the brail, rivet, and body parts are illustrated in Fig. 11E.6.



Fig. 11E.5. *Removing a properly fitting brail.* Photo David H. Ellis

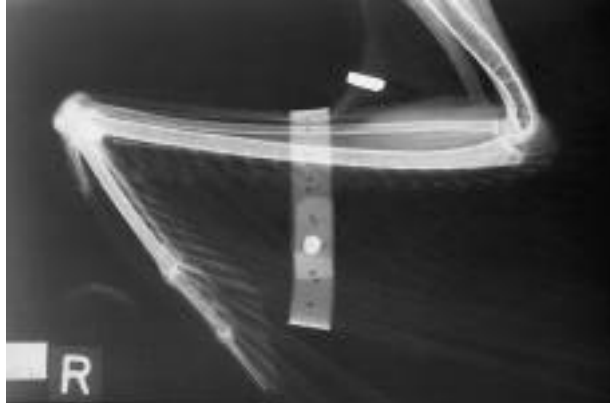


Fig. 11E.6. *Radiograph of a brailed wing.* Photo F. Joshua Dein

Proper fit is important. If the brail is too loose, it will slide toward the humerus until the primaries are free; if too tight, it may restrict circulation in the wrist and hand or cut into the skin. The fit is checked by placing two fingers in the upper loop on the dorsal surface of the wing. If the fingers slide under easily but snugly, the fit is good. If the fingers do not slide easily under the upper loop, the brail should be removed.

Upon release, brailed birds (especially those brailed for the first time) will stumble or even fall when they fail to spread the now brailed wing for balance. Also, during the first 10 minutes or so after release, birds strain at the brail and preen vigorously at the site of the brail. After a few hours, however, the birds typically pay little attention to the brail.

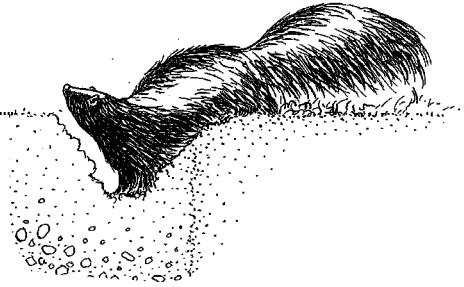
Over 300 previously brailed cranes have been released to the wild (see Chapter 11D). Many of these have survived several years after release. Occasionally, a bird shows slight (but long-term) **impairment** in its ability to extend the hand of one wing following a lengthy brailing period. Such individuals do well in non-migratory flocks, but we suspect that they would be significantly impaired if migratory. Sometimes during brailing, the patagium or the integument between the primaries is damaged by a brail. Such wounds are rare: only once has a bird been so incapacitated by this kind of injury that it was not released.

Literature Cited

- Amand, W. B. 1986. Surgical problems (Galliformes). Pages 473-475 in M. E. Fowler, editor. *Zoo and wild animal medicine*. 2nd ed. W. B. Saunders, Philadelphia, Pa.
- Ellis, D. H., and F. J. Dein. 1991. Flight restraint techniques for captive cranes. Pages 447-451 in J. Harris, editor. *Proceedings 1987 Crane Workshop*. International Crane Foundation, Baraboo, Wis.
- Epperson, R. G., Jr. 1982 unpubl. Evaluation of the effects of restraining flight in young cranes using wing brails. U.S. Fish and Wildlife Service, Patuxent Wildlife Research Center. 13 pp.
- Gandal, L. P., and W. B. Amand. 1982. Anesthetic and surgical techniques. Pages 304-328 in M. L. Petrak, editor. *Diseases of cage and aviary birds*. 2nd ed. Lea and Febiger, Philadelphia, Pa.
- Harrison, G. J., and L. R. Harrison. 1986. Management procedures. Pages 85-100 in G. L. Harrison and L. R. Harrison, editors. *Clinical avian medicine and surgery*. W. B. Saunders, Philadelphia, Pa.
- Madill, D. N. 1981. Surgical procedures. Pages 211-212 in T. G. Hungerford, editor. *Refresher course on aviary and caged birds*. Proceedings No. 55, University of Sydney, Post Graduate Committee in Veterinary Science, Sydney, N.S.W., Australia.
- Mangili, G. 1971. Unilateral patagiectomy: a new method of preventing flight in captive birds. *International Zoo Yearbook* 11:252-254.
- Miller, J. C. 1973. The importance of immobilizing wings after tenotomy and tenotomy. *Veterinary Medicine and Small Animal Clinician* 68:35-38.
- Osinskij, L. P., and V. S. Taran. 1978. On methods of partial amputation of hand in birds. [In Russian.] *Vestnik Zoologii* 1978(4):69-72.
- Robinson, P. T. 1975. Unilateral patagiectomy: a technique for deflighting large birds. *Veterinary Medicine and Small Animal Clinician* 70:143-145.
- Robinson, P. T., and R. B. Buzikowski. 1975. Pinioning young birds with hemostatic clips. *Veterinary Medicine and Small Animal Clinician* 70:1415-1417.
- Schroeder, C. R., and K. Koch. 1940. Preventing flight in birds by tenotomy. *Journal of the American Veterinary Medical Association* 97:169-170.
- Schwarte, L. H. 1965. Poultry surgery. Pages 1149-1161 in H. E. Biester and L. H. Schwarte, editors. *Diseases of poultry*. 5th ed. Iowa State University Press, Ames.
- Sedgewick, C. J. 1967. Deflighting pet birds. *Modern Veterinary Practice* 48(3):38-40.
- Sheppard, C. and D. Bruning. 1983. Development of techniques to aid the long-term survival of White-naped and other rare crane species. Pages 203-207 in *Proceedings of the Annual Conference of the American Association of Zoological Parks and Aquariums*.
- Wallach, J. D., and W. Boever. 1983. *Diseases of exotic animals: medical and surgical management*. W. B. Saunders, Philadelphia, Pa. 1,159 pp.
- Williamson, W. M., and W. C. Russell. 1971. Prevention of flight in older captive birds. *Journal of the American Veterinary Medical Association* 159:596-598.
- Young, W. A. 1948. Wing amputation of birds in lieu of pinioning. *Journal of the American Veterinary Medical Association* 112:224-226.
- Zwank, P. J., and S. R. Derrickson. 1981. Gentle release of captive, parent-reared Sandhill Cranes into the wild. Pages 112-116 in J. C. Lewis, editor. *Proceedings 1981 Crane Workshop*. National Audubon Society, Tavernier, Fla.

Special Techniques, Part F: Predator and Pest Management

Thomas E. Lewis



A pest is any unwanted organism that directly interferes with human activity (Miller 1988). Insect pests can transmit diseases to captive cranes (Carpenter et al. 1987). Mammal and bird pests consume and contaminate food and can also transmit diseases and parasites (Carpenter and Derrickson 1987). Plant pests may injure cranes, cause illness, or hamper normal husbandry practices. The University of Maryland (1986-87) publication on pest management is a primary source for this chapter, and references to it have been abbreviated UM.

Depredation by birds and mammals accounted for the loss of 51% of 456 wild Sandhill Crane nests in Oregon (Littlefield 1976). Hartman (1987) reported that both raptors and mammals kill captive cranes. Eggs and chicks are, of course, the most vulnerable.

An effective predator and pest management program should: (1) minimize the negative impacts of predators and pests; (2) provide for the safety of personnel, captive cranes, and the environment; (3) operate in accordance with appropriate laws and regulations; and (4) provide training to personnel involved in predator and pest management.

Laws and Regulations

In the United States, the Environmental Protection Agency (EPA) regulates pesticides. The Federal Insecticide, Fungicide, and Rodenticide Act (FIFRA), enacted in 1947 and amended in 1972 and 1978, has established requirements for the registration, manufacture, transportation, and use of pesticides (UM; Miller 1988). State and local governments regulate predator control unless the target species is migratory, threatened, or endangered. In these cases, the following federal regulations must also be met: the Code of Federal Regulations Title 50 (50 C.F.R.) Parts 10

and 21 (Migratory Bird Treaty Act), Part 22 (Eagle Protection Act), and Parts 17 and 23 (Endangered Species Act) (Millsap 1987). A person operating a predator or pest management program must comply with all local and federal laws and regulations, and often must be certified. Contact local or state governments to determine applicable regulations. Failure to comply could result in a fine and/or imprisonment (UM).

Training and Safety

At least one employee in a crane husbandry program should be trained and certified in predator and pest management. Once certified, a person can often train other personnel.

The most important safety rule is to follow pesticide labels and instructions for operating equipment. Material safety data sheets (MSDS) should be obtained from the manufacturer and retained. Because many pesticides can be absorbed through the skin, inhaled, swallowed, or enter the body through the eyes and ears, use protective clothing and equipment and follow proper cleanup procedures to reduce exposure (UM). Protective clothing and equipment should be washed after every use. Pesticide contaminated clothing should be laundered separately from other clothing (University of Maryland 1984). Persons routinely using pesticides should have regular medical checkups and should inform their physician so he can watch for symptoms of overexposure and check medications for adverse interactions with pesticides (UM).

Integrated Pest Management (IPM)

The IPM approach involves many tactics to keep predators and pests below acceptable levels while minimizing harmful effects to the environment (UM; Miller 1988). Each program should include (1) cultural control, (2) sanitation, (3) mechanical and physical control, and (4) chemical control.

Cultural control includes planting, harvesting, and tillage practices that are unfavorable to predators and pests (UM). For example, mowing regularly reduces undesirable woody vegetation in and around crane pens and facilities, and planting densely branched shade trees, such as the Bradford pear (*Pyrus calleryana*), discourages raptors from perching (Fig. 11F.1).

Sanitation reduces food, water, or shelter for pests (UM). Keep food preparation and food storage areas free of spilled food. Providing cranes with the proper amount of food reduces spillage, and removal of spilled food helps prevent disease (Larue 1981; Carpenter and Derrickson 1981, 1987; Carpenter 1986).

Mechanical and physical control devices separate cranes from predators and pests. Electric fences (Fig. 11F.2) exclude mammals (Putnam and Archibald 1987), and spikes or pointed wires on top of utility poles or other likely raptor perches discourage avian predation. Trapping and removal should be employed when other methods have failed.



Fig. 11F.1. *The densely branched Bradford pear tree provides shade but does not provide good perches for raptors.*

Photo Jonathan P. Male

Chemical control agents kill, repel, attract, sterilize, or otherwise interfere with the normal behavior of predators or pests (UM). These include pesticides, herbicides, avicides, and frightening agents. Choose the agent that is least disruptive to the crane colony and to the environment and least toxic to the pest's natural enemies.



Fig. 11F.2. *Electric fence and solar charge units.*

Photo David H. Ellis

Specific Management Techniques

Mammalian Predators

Mortality of captive cranes has been caused by raccoons (*Procyon lotor*) (Hartman 1987; Doughty 1989) and probably foxes (*Vulpes* sp. or *Urocyon* sp.) (Carpenter and Derrickson 1981). Feral cats (*Felis catus*) and dogs (*Canis familiaris*), opossums (*Didelphis marsupialis*), and mustelids (Mustelidae) are also potential crane predators. Even rodents (Rodentia) may prey upon crane eggs. At Patuxent, rats (*Rattus* sp.) were once (1966-68) an important mortality factor for eggs (B. I. Williams, Patuxent, personal communication).

Physical barriers (e.g., fences) limit access and prevent losses to mammalian predators. Burying the perimeter fence and extending the wire fabric outward in the trench (Fig. 11F.3) discourages mammalian predators from digging under fences (see Chapter 12; Putnam and Archibald 1987). Archibald and Viess (1979) recommend burying fencing 0.3 m (1 ft) in gravel. Nylon flight netting (Fig. 12.8) discourages avian and to a degree mammalian predators, but both Hartman (1987) and Doughty (1989) reported that raccoons killed cranes despite flight netting. Electric fencing (Figs. 11F.2 and 11F.3) also discourages mammalian predators (see Chapter 12; Putnam and Archibald 1987). We recommend a single, electrical bottom wire (0.5 m [1½ ft] above ground level on brackets holding it 6 cm from the fence) and double

electrical top wires (at least 2 m above ground level and on brackets holding the inner wire 6 cm from the fence and the outer wire 10 cm from the fence). Wire mesh (2.5 cm, 1 in) should extend into the ground and extend above the ground at least to the height of the lower electrical wire. Fence lines and netting should be checked weekly, and the electric current should be tested daily.

Even when outside the pen, mammalian predators can also cause crane injuries or mortality. When frightened, cranes collide with fences or other obstacles. To avoid this problem, crane holding facilities should be surrounded by a perimeter fence (see Chapter 12) providing a “buffer zone” 10 m wide or

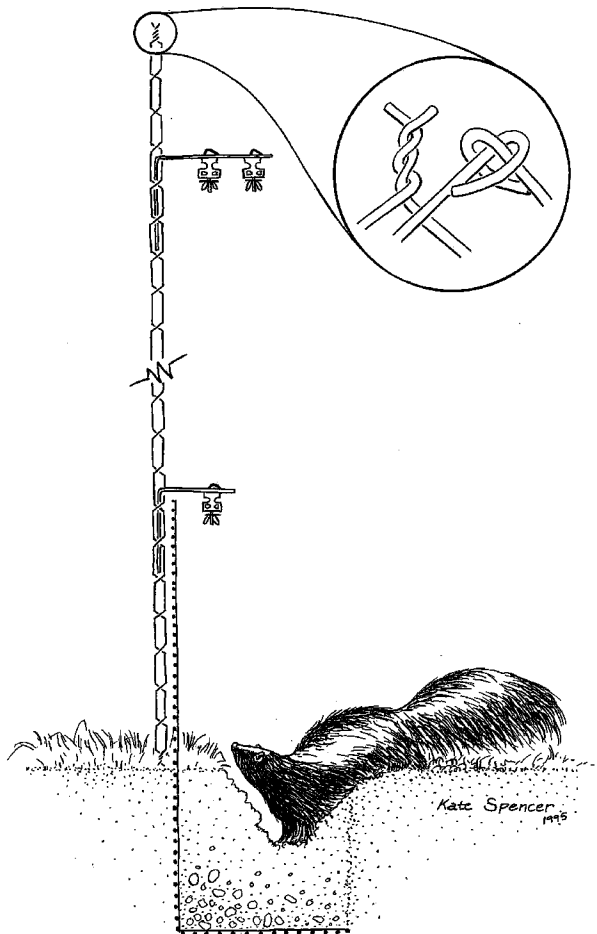


Fig. 11F.3. Crane breeding pen showing predator exclusion modifications. Inset shows two types of chain link ends. Barbed ends are used only on perimeter fences. Knuckled ends are used on crane pens. Knuckles at the base protect chicks. Those at the top facilitate net attachment or, if the pen is unnetted, prevent injuries if cranes contact the fence top in high winds. Many configurations are possible for electrical wires. In extreme cases, alternate electrical and ground wires for up to 30 cm from the fence. Art Kate Spencer

wider. Visual barriers (e.g., tennis netting, Fig. 12.12) attached to a perimeter fence or the pen wall also help reduce disturbance.

Problem predators can be trapped and translocated, or killed. We recommend using live traps when practical; humane treatment should be provided regardless of which removal technique is used. Trapping success will be greatest when the trap is brought to the animal rather than trying to lure the animal to a new location.

Live traps (Fig. 11F.4) are effective for raccoons, skunks (Mustelidae), opossums, and domestic dogs and cats (Bogges et al. 1990). At Patuxent, we have used Tomahawk Model 108 for raccoons and opossums and model 110A, B, or C for medium-sized canids (see Appendix). Manufacturers can suggest the best trap for specific needs. These traps must be baited. Canned sardines or cat food attract raccoons and opossums while meat baits work well for canids.

A variety of commercial steel traps are available to capture animals (Day et al. 1980). Conibear traps (Fig. 11F.4) (see Appendix) are effective over burrows and where predators are travelling through narrow corridors, while foot-hold traps (Fig. 11F.4) are more effective on raccoons (Bogges et al. 1990). Hawthorne (1980) recommends No. 1-1½ foot-hold traps for raccoons. Day et al. (1980) states that No. 1 and No. 2 steel traps are effective on foxes, and No. 3 and No. 4 steel traps are effective on larger canids. Henderson (1985) recommends using two Victor No. 3N coyote traps per set for coyotes (*Canis latrans*). Foot-hold traps with offset and padded jaws are used to capture carnivores more humanely (Day et al. 1980). Red wolves (*Canis rufus*) are captured with Woodstream



Fig. 11F.4. Three kinds of traps: live (top), Conibear (left), and foot-hold (right; note padded, rubber jaws).

Photo Thomas E. Lewis

“Soft Catch” No. 3 traps (see Appendix) without serious harm to the animals (personal observation).

The most popular ways to set foot-hold traps are scent posts, dirt holes, and trail sets. Scent post sets use a natural scent such as urine or feces as an attractant. Dirt hole sets use a pungent meat-based attractant in a hole that is 15–25 cm behind the trap. Sets should be placed so that prevailing winds carry the odor across the path of the predator. Trail sets are placed along travel routes where natural features funnel predator movements. All sets should be placed on level ground when possible.

Carefully bury foot-hold traps 1 cm below the surface and chain them to a drag or stake. Place a canvas, plastic, cloth, wire mesh, or wax paper cover over the bait pan and under the jaws, then sift dry fine soil over the trap, chain, and drag or stake. The sets should be camouflaged to blend into the surrounding area. Traps must be checked daily and be set only in areas inaccessible to cranes.

Mammalian Pests

Carpenter and Derrickson (1987) report rodents spreading disease and parasites between areas. Rodents can also consume feed, contaminate feed with their feces and urine, and damage facilities by gnawing and burrowing. Raccoons, opossums, skunks, and marmots (*Marmota* sp.) can consume large quantities of crane feed in a relatively short time. When marmots dig under fences, they provide access for other mammals. Cranes and caretakers can also be injured in marmot burrows.

Fumigants are effective toxicants against burrowing mammals. At Patuxent, we use Giant Destroyer cartridges (see Appendix) with active ingredients of sodium nitrate (46.2%), sulfur (34.8%), and charcoal (8.7%). Place the cartridges directly into the burrow and cover all openings with soil. Filling the holes also eliminates the possibility of cranes succumbing to the fumigant or injuring themselves in the burrow.

For excluding small rodents, crane feed sheds at Patuxent have metal shields (Fig. 11F.5) above the feeder. Good sanitation in feed storage and feeding areas also reduces rodent activity.

Rodenticides are also effective in controlling rodents. Most are stomach poisons. Unless you use two or more different baits in rotation, rodents may develop resistance to or learn to avoid the baits. Many modern baits pose a very low risk of secondary poisoning. For example, Quintox (see Appendix), with

the active ingredient cholecalciferol poses low risk to non-target animals. Place these baits in tamperproof boxes (Fig. 11F.6) that facilitate use by rodents but exclude non-target animals. Place bait stations along walls and in known rodent runways, but far enough from pens so that cranes cannot reach them and within perimeter fences so contact with children is unlikely. At Patuxent, we place a bait station in front of every second occupied crane pen (i.e., at ca 60 m intervals). Bait stations should be checked weekly. Bait consumption rates indicate the level of infestation.

Problem rodents can be captured individually in live traps. At Patuxent, we bait marmots into a large live trap using apple slices.

Avian Predators

Raptors, both diurnal and nocturnal, may kill adult cranes or chicks. Great-horned Owls (*Bubo virginianus*) have killed captive cranes (Archibald and Viess 1979; Hartman 1987). At Patuxent, Great-horned Owls have killed both adult and juvenile cranes. In addition, crows (*Corvus* sp.) at Patuxent have harassed incubating cranes until the cranes left their nest long enough for the crows to break or steal the eggs (B. I. Williams, Patuxent, personal communication).



Fig. 11F.5. An overhead rodent shield protects the crane food.

Photo Jonathan P. Male



Fig. 11F.6. Rodent bait stations.

Photo Jonathan P. Male

In the United States, the capture, possession, or transport of raptors for depredation control requires a depredation permit (Millsap 1987). The most effective technique to reduce avian predation is to use netted pens (Fig. 12.8). Another effective method is to release target species (such as the domestic Pekin duck, *Anas platyrhynchos*) and allow them to roam free in the colony. When a duck is killed, the carcass is thereafter used to trap the raptor.

Several methods are available to capture raptorial birds while preventing injury to the raptor. Padded foot-hold traps have been useful for owls and eagles elsewhere (Bloom 1987) and at Patuxent. Padded foot-hold traps are most effective when at least four traps are placed around a bait. Use 60-cm chains to attach each trap to a wooden drag (ca 50 cm x 10 cm x 10 cm). The drag prevents escape and absorbs the shock as the bird attempts to flee. Partially cover the traps and chains with soil, grass, leaves, etc. Too much debris over the trap will reduce holding efficiency.

After a raptor kills a crane, it will often return on subsequent days to feed on the carcass. For example, at Patuxent, one Great-horned Owl returned to the same carcass on three consecutive nights allowing us to capture it. Raptors can either be baited to the kill site with the carcass of the crane they killed or another large bird carcass can be substituted. The trap site can also be somewhat removed from the kill site if the pen is still occupied by cranes.

A verbal trap (Bloom 1987) or padded foot-hold trap (Fig. 11F.7) set atop a nearby pole is effective alone or in conjunction with traps around a carcass. If necessary, a temporary perch pole can be installed near the kill to support the trap. The verbal consists of one large (10-cm diameter or larger) nylon noose and trigger mounted atop a post. A spring closes the noose around the bird's leg or legs when it lands on the trigger. The spring is tied to the perch with a nylon line that allows the bird to flutter safely to the ground.

Once a bird is captured, it should be examined for injuries, and then released at least 20 km away from the trap site. At Patuxent, we commonly transport owls at least 100 km from the kill site.

Avian Pests

Avian and mammalian pests pose similar problems. Wild birds, especially other cranes, should be excluded from a crane colony to avoid introducing parasites and diseases (Carpenter and Derrickson 1987). Flocks of small birds sometimes consume large



Fig. 11F.7. Padded foot-hold pole trap (note jaw with rubber to protect the legs of a bird).

Photo Jonathan P. Male

quantities of feed and contaminate food with feces. Other pest birds, such as House Sparrows (*Passer domesticus*), may nest in crane shelter areas and could thereby create parasite and disease problems.

Alternatives to killing pest birds are available. Although flight netting (Fig. 12.8; mesh size 5.1 cm [2 in]) will discourage predators, many species of small birds can pass through this barrier. Providing food indoors can reduce the attraction to certain pest bird species. A pest guard (Fig. 11F.8) has been designed and tested at Patuxent to protect crane feeders. It



Fig. 11F.8. Pest guard on crane feeder at right allows the crane's bill to pass, but excludes even very small pest birds and mammals. Normally feeders are suspended 0.5 m from the ground.

Photo David H. Ellis

consists of parallel bars 11 mm apart. This design allows the bill of the crane access to food but excludes small birds and mammals. To discourage House Sparrows, remove their nests, and, when possible, cover the opening to the nest site with 1.2 cm (0.5 in) wire hardware cloth to prevent reentry and subsequent nesting attempts. If the nest sites cannot be covered, nests should be removed bi-weekly.

If pest birds cannot be excluded from crane holding facilities, trapping and chemical control may be necessary. Although Starlings (*Sturnus vulgaris*), House Sparrows, and feral Pigeons (*Columba livia*) do not have protection under federal law in some countries, they may have local protection (MDA 1984). Native birds may also be affected by trapping procedures so federal and local permits are required. The U.S. Fish and Wildlife Service (1977), Hawthorne (1980), and Bub (1991) discuss methods of trapping pest birds. Mist nets can be used (Day et al. 1980), but birds must then be released far (at least 10 km) from the crane colony.

When trapping is ineffective, toxic or aversive chemicals may be required. Employ caution to avoid contact with the cranes. Avitrol (see Appendix) is a frightening agent with an active ingredient of 4-aminopyridine. When ingested, it causes the bird to emit distress cries while flying erratically. This behavior frightens the rest of the flock. The product is relatively safe, affects only 1% of the target species, and has no secondary hazards (Hawthorne 1980). Avitrol is registered for control of feral Pigeons, gulls (*Larus* sp.), House Sparrows, Starlings, and blackbirds (Icterinae).

Starlicide Complete (DRC-1339, 3-chloro-4-methylbenzamine HCl; see Appendix) is a chemical toxin used to control birds. At Patuxent, we control Starlings, crows, and Grackles (*Quiscalus* sp.) with Starlicide in crane feeders in unoccupied pens. During extreme infestations, we briefly (no longer than 8 hours) remove the feeders from occupied pens during application so that the offending birds will concentrate on the Starlicide. After use, contaminated feeders are thoroughly cleaned. Any spilled Starlicide must be removed before cranes are introduced into these pens. Cranes that have accidentally ingested Starlicide pellets at Patuxent have been treated with activated charcoal and have recovered with no permanent effects (G. F. Gee, Patuxent, personal communication). Starlicide is highly toxic to most pest birds, but less toxic to predatory birds and mammals (DeCino et al. 1966; Schafer 1981).

Avicides can be placed on elevated boxes (25-30 cm) in the area where pest birds are feeding or, during periods of snow cover, placed on a plastic sheet on the snow. Cover avicides overnight to prevent them from becoming wet from dew or precipitation or from being eaten by non-target species. Unused avicide should be destroyed by incineration or, if in good condition, placed back in the original container.

Reptilian Predators and Pests

To reduce the threat large snakes may pose to chicks, use one of the two types of snake proofing discussed by Hawthorne (1980): (1) a 0.64 cm (0.25 in) heavy galvanized mesh screen 90 cm wide which is buried a few cm in the ground and slanted 30 degrees outward from bottom to top, or (2) a 10 to 15 cm wide and 5 cm deep strip of concrete around the perimeter with an electric wire 12 cm above the concrete. Mowing close to the ground eliminates food and cover for reptiles. Moats may be used, and reptile traps are also available (Day et al. 1980).

Arthropod Pests

Problems from arthropods include: (1) ectoparasites, (2) irritations or mortality caused by insect bites and stings, and (3) mortality caused by insect-borne diseases. In 1984, the eastern equine encephalitis virus killed 7 of 39 Whooping Cranes at Patuxent (Derrickson 1985; Carpenter et al. 1987). The principle vector was the mosquito *Culiseta melanura*. Other arthropod-borne diseases are listed in Chapter 8. Some of these (e.g., Lyme disease) also threaten caretakers.

Spot application of insecticides or repellents is the most common method of insect control. Bees (Apidae) and wasps (Vespidae) in crane holding facilities must be controlled to avoid potentially dangerous stings to cranes and caretakers. At Patuxent, small chicks have developed swollen areas from suspected stings. When wasp nests are found, spray them immediately. Wasp spray (Wasp Freeze; see Appendix), when applied to the corners of feed sheds and other shelters, appears to repel for about 30 days. Insect attacks are also dangerous to humans especially when operating machinery. To avoid injury, destroy ground nests by first flagging them upon discovery, and then spraying them during the evening or early morning when the insects are inactive.

When flies (Order Diptera) threaten chicks or injured birds, bait with attractants and fly paper (but keep these out of reach of cranes), or treat afflicted birds with insect repellent. Fire ants (*Solenopsis* sp.) threaten ground nesting birds in some areas (Vinson and Sorenson 1986). If ants are a problem, seek help from governmental agencies and private exterminators that control insects. Many tick-borne (Ixodidae) diseases threaten humans and wildlife (Davidson and Nettles 1988). Personnel should wear repellent, tuck pant legs into socks, and examine themselves for ticks after each visit to infested areas. Permethrin (active ingredient 0.5% Permethrin; see Appendix) repels ticks, but should only be applied to clothing. Regular pen mowing also reduces contact with ticks.

Plant Pests

Pest plants can cause illness and injury to cranes, damage facilities, and interfere with normal husbandry practices. In general, cranes do not eat noxious plants; however, if a problem is suspected, obtain information through local agricultural extension agents. Plants with thorns or sharp projections can cut or puncture cranes (especially chicks) and cause secondary infections.

Except for shade trees, grasses should predominate in outdoor crane holding facilities. Vines and woody vegetation can cause injuries to cranes, damage fences and facilities, interfere with the proper functioning of electric fences, interfere with mowing, and damage equipment. Large plants can also hamper visual inspection of cranes and facilities.

Mowing reduces woody vegetation and makes conditions less suitable for hosts of diseases (Carpenter and Derrickson 1987). To reduce disturbance to the cranes, vegetation control can be coordinated around the pen rotation schedule (see Chapter 2). At Patuxent, we mow empty pens first, then move birds into the empty pens, and then mow the formerly occupied pens. At Patuxent, we mow about three times each season. The pens in the breeding colonies are not mowed until egg production is completed and there are no chicks under 30 days of age in the immediate area. We mow these pens again in late summer, 5-8 weeks after the first mowing.

A plant-free zone should extend 0.25 m from each side of perimeter fences and can be maintained with non-selective herbicides like Roundup (N-phosphonomethyl-glycine 41%; see Appendix). An additional 1 m strip on either side of perimeter fence should be

mowed to keep the vegetation short. A vegetation-free area around water sources is maintained by placing a 10 cm deep bed of gravel on top of a 1 m diameter sheet of plastic (Fig. 12.13). The interior of feed sheds can be kept vegetation free with a 10 cm deep layer of sand that also facilitates removal of spilled food (Fig. 11F.9). Using non-selective herbicides and renewing gravel or sand beds when needed will maintain these areas vegetation free but should be coordinated with mowing to reduce disturbance. Avoid using herbicides where they are accessible to cranes.

Combine various methods of vegetation control to maximize their effectiveness. For example, if mowing is scheduled a few days after herbicide use, regrowth is reduced (i.e., weakened by the herbicides).

If pest plant problems arise during the breeding season, postpone control measures unless there is an immediate threat to the cranes. For example, it may be advisable to remove thistles from a pen with a small chick.



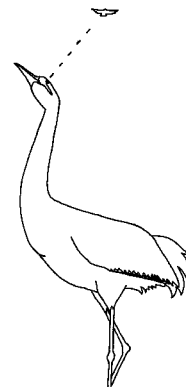
Fig. 11F.9. Sand floor in the feed shed facilitates removal of spilled food and other debris.

Photo Jonathan P. Male

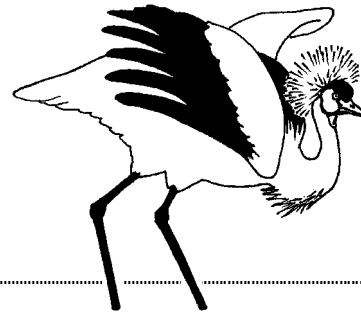
Literature Cited

- Archibald, G. W., and D. L. Viess. 1979. Captive propagation at the International Crane Foundation, 1973-78. Pages 51-73 in J. C. Lewis, editor. Proceedings 1978 Crane Workshop. Colorado State University Printing Service, Ft. Collins.
- Bloom, P. H. 1987. Capturing and handling raptors. Pages 99-123 in B. A. Giron, B. A. Millsap, K. W. Kline, and D. W. Bird, editors. Raptor management techniques manual. National Wildlife Federation, Washington, D.C.
- Boggess, E. K., et al. 1990. Traps, trapping, and furbearer management: a review. *Wildlife Society Technical Review* 90-1. 31 pp.
- Bub, H. 1991. Bird trapping and bird banding: a handbook for trapping methods all over the world. [English translation: F. Hamerstrom and K. Wuertz-Schaefer.] Cornell University Press, Ithaca, N.Y. 330 pp.
- Carpenter, J. W. 1986. Cranes (Order Gruiformes). Pages 315-326 in M. E. Fowler, editor. *Zoo and wild animal medicine*. W. B. Saunders, Philadelphia, Pa.

- Carpenter, J. W., F. J. Dein, and G. W. Clark. 1987. An outbreak of Eastern Equine Encephalitis virus in captive whooping cranes. Pages 123-127 *in* J. C. Lewis, editor. Proceedings 1985 Crane Workshop. Platte River Whooping Crane Maintenance Trust, Grand Island, Nebr.
- Carpenter, J. W., and S. R. Derrickson. 1981. Whooping Crane mortality at the Patuxent Wildlife Research Center, 1966-1981. Pages 175-179 *in* J. C. Lewis, editor. Proceedings 1981 Crane Workshop. National Audubon Society, Tavernier, Fla.
- Carpenter, J. W., and S. R. Derrickson. 1987. Infections and parasitic diseases of cranes: principles of treatment and prevention. Pages 539-553 *in* G. W. Archibald and R. F. Pasquier, editors. Proceedings of the 1983 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Davidson, W. R., and V. F. Nettles. 1988. Field manual of wildlife diseases in the Southeastern United States. Southeastern Cooperative Wildlife Disease Study, College of Veterinary Medicine, University of Georgia, Athens. 309 pp.
- Day, G. I., S. D. Schemnitz, and R. D. Taber. 1980. Capturing and marking wild animals. Pages 61-88 *in* S. D. Schemnitz, editor. Wildlife management techniques manual. 4th ed. The Wildlife Society, Bethesda, Md.
- DeCino, T. J., D. J. Cunningham, and E. W. Schafer. 1966. Toxicity of DRC-1339 to Starlings. *Journal of Wildlife Management* 30:249-253.
- Derrickson, S. R. 1985. Captive propagation of Whooping Cranes, 1982-1984. Pages 377-386 *in* J. C. Lewis, editor. Proceedings of the 1985 Crane Workshop. Platte River Whooping Crane Maintenance Trust, Grand Island, Nebr.
- Doughty, R. W. 1989. Return of the Whooping Crane. University of Texas Press, Austin. 182 pp.
- Hartman, L. M. 1987. Summary of mortality of 14 species of cranes at The International Crane Foundation, 1972-1982. Pages 555-570 *in* G. W. Archibald and R. F. Pasquier, editors. Proceedings of the 1983 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Hawthorne, D. W. 1980. Wildlife damage and control techniques. Pages 411-439 *in* S. D. Schemnitz, editor. Wildlife management techniques manual. 4th ed. The Wildlife Society, Bethesda, Md.
- Henderson, R. F. 1985. How to trap a coyote. KSU Extension Bulletin C-660, Kansas State University Cooperative Extension Service, Manhattan, Kansas. 10 pp.
- Larue, C. 1981. Techniques for breeding cranes in captivity. Pages 15-18 *in* J. C. Lewis and H. Masatomi, editors. Crane research around the world. International Crane Foundation, Baraboo, Wis.
- Littlefield, C. D. 1976. Productivity of Greater Sandhill Cranes on Malheur National Wildlife Refuge, Oregon. Pages 86-92 *in* J. C. Lewis, editor. Proceedings of the International Crane Workshop. Oklahoma State University Publishing and Printing, Stillwater.
- Maryland Department of Agriculture (MDA). [1984]. Pesticide training manual on bird control. MDA 200-84, Maryland Department of Agriculture, Annapolis. 27 pp.
- Miller, G. T., Jr., editor. 1988. Living in the environment: an introduction to environmental science. 5th ed. Wadsworth Publishing, Belmont, Calif. 603 pp.
- Millsap, B. A. 1987. Introduction to federal laws and raptor management. Pages 23-33 *in* B. A. Giron, B. A. Millsap, K. W. Kline, and D. W. Bird, editors. Raptor management techniques manual. National Wildlife Federation, Washington, D.C.
- Putnam, M. S., and G. W. Archibald. 1987. The Siberian Crane: its history and biology in captivity. Pages 173-195 *in* Proceedings of the 1983 International Crane Workshop. G. W. Archibald and R. F. Pasquier, editors. International Crane Foundation, Baraboo, Wis.
- Schafer, E. W., Jr. 1981. Bird control chemicals—nature, modes of action, and toxicity. Pages 129-139 *in* A. A. Hanson, editor. Handbook of pest management in agriculture. Vol. III. CRC Press, West Palm Beach, Fla.
- University of Maryland. 1984. Laundering pesticide contaminated clothing. University of Maryland Cooperative Extension Service, College Park. 2 pp.
- University of Maryland (UM). 1986-87. Maryland pesticide applicator training series: core manual. University of Maryland, Cooperative Extension Service, College Park. 42 pp.
- U.S. Fish and Wildlife Service. 1977. North American bird banding manual. Vol. 2. Bird banding techniques. Washington, D.C.
- Vinson, S. B., and A. A. Sorenson. 1986. Imported fire ants: life history and impact. Department of Entomology, Texas A&M University, College Station, TX and Agriculture and Environmental Services, Texas Department of Agriculture, Austin. 28 pp.



Facilities



Scott R. Swengel and Richard W. Besser

Incubation and Hatching Facilities

Incubation and hatching areas should be in different rooms with **separate air supplies**. Positive air pressure in both rooms is important to insure that air flows out of the room, rather than in, when a door is opened. Air conditioning helps control relative humidity, thereby allowing the incubators and hatchers to maintain a more constant environment. Air conditioning becomes especially important on hot summer days.

These rooms should also have surfaces that are easy to disinfect (e.g., formica counters and smooth walls) and a central floor drain. **Emergency power** sources are imperative. Gasoline or propane generators may serve this purpose.

Chick Rearing Facilities

Hand-rearing Facilities

Service Area. The service room for the chick-rearing facilities should be an insulated and heated room with hot and cold running water. In torrid environments, air conditioning is advisable. The walls and floors should also be readily cleaned, and a floor drain in the center of a tiled or concrete floor is important. An exhaust fan may be used to reduce humidity in the room after cleaning.

Two **sinks** are recommended: one should be mounted on a counter, and a second one should be a 1 m² floor basin with 15-cm high sides. The small sink is used for washing food and water containers, while the floor basin is used for washing large items such as brooder box carpets. Splash zones on walls near the floor basin should be sealed to prevent water damage and to avoid septic conditions.

The service room should also include **shelves and cabinets** (for storing equipment and medical supplies) and a small **refrigerator/freezer** (for storing vitamins and medicine). Another nearby room should house a one-month **food supply** (less in warm, humid weather) in paper bags on wooden pallets. Opened food bags may be emptied into a sealable steel or plastic container. In some areas, air conditioning will be required to keep food dry.

Brooder Room. The brooder room, like the service room, should be insulated, heated, and easily disinfected. The floors should be tile or sealed concrete. The walls should be painted with enamel or epoxy-based paint, or otherwise sealed. Electrical outlets and light fixtures should be able to withstand periodic hoseings. Counter space should be provided for record keeping, preparation of medicines, and weighing chicks.

Brooder boxes, if portable, should be elevated to avoid the cold floor. Floor brooders should have insulated substrates. Electrical outlets should be placed where they cannot be contacted by the chicks. Brooder boxes (Fig. 12.1) should be at least 0.5 to 1 m² and have fine mesh screen or plexiglass sides to enable chicks to see their neighbors but to prevent them from fighting and injuring one another. The inside com-



Fig. 12.1. Brooder box with a hatchling chick.

Photo ICF

partment should be at least 35-cm high for small chicks and 50-cm high for larger ones.

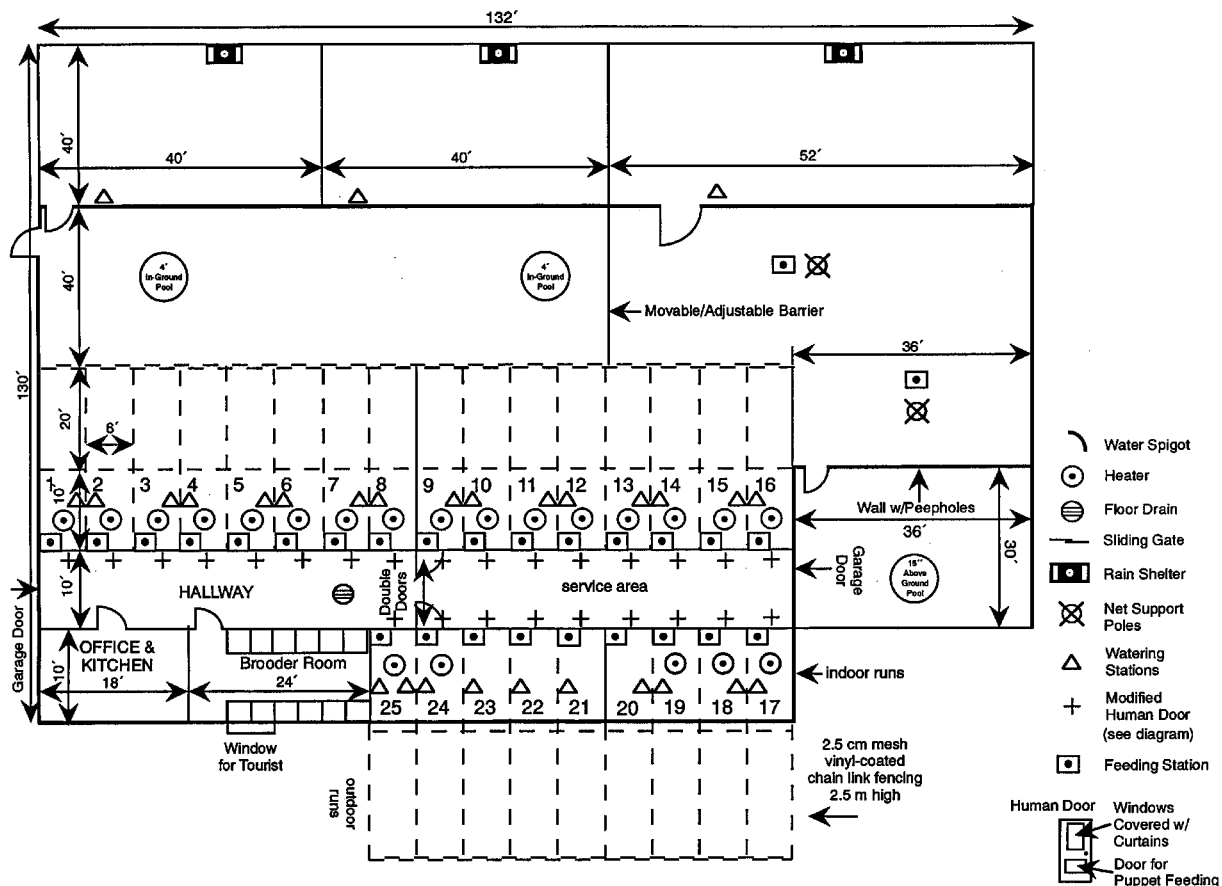
Avoid using rough or **abrasive surfaces** for walls inside the brooders to minimize chick injuries. Provide opaque dividers if neighboring chicks are incompatible and install a wall **mirror** to promote proper sexual imprinting of isolated chicks. If one end of the box has a hinged door, design it to swing up or to the side (but *not* down, because gravity could cause poorly-latched doors to fall and injure a chick). It is also advisable to place a **chick guard** that provides a 15-cm high rim in front of the door so that chicks cannot fall out when the box is opened. The top and/or sides of the box should be well **ventilated** to prevent overheating, to promote air circulation, and to hasten the drying of wet surfaces.

Any reliable **heating system** may be used, but we suggest **electric coil heaters** suspended in the sheet metal roof and controlled by **solid-state or wafer-operated** switches. A **back-up thermostat** set a few degrees above the desired temperature prevents overheating in case the main thermostat fails. This is

especially important if wafer thermostats are used. The heating coil should be **at least 35 cm** above the box floor to prevent chicks from touching the coil with their heads as they grow taller. A **thermometer** should be mounted on one side of each box to allow frequent temperature checks. If the brooders are placed near windows where they receive sunlight, install **adjustable shades** (venetian blinds work best) over the windows to prevent the boxes from overheating.

Non-toxic **carpets**, cut to size, cover the floor of the boxes. Carpets should have relatively short fibers that do not abrade chicks. Rubber-backed carpeting is preferable, because it will not slide on the smooth brooder box floor. Two carpets are needed for each box to allow for replacement during daily cleaning. These carpets can be readily cleaned by hosing them on an inclined surface.

Chick Pens (Figs. 12.2 and 12.3). When chicks are 3-14 days old, they may be allowed access to **outdoor pens** (see Chapter 5). A door separates the inside and outside runs so chicks can be locked inside at night or



CHICK HOUSE PLAN

Fig. 12.2. Diagram of chick-rearing building at ICF; runs are termed pens in Fig. 12.3.

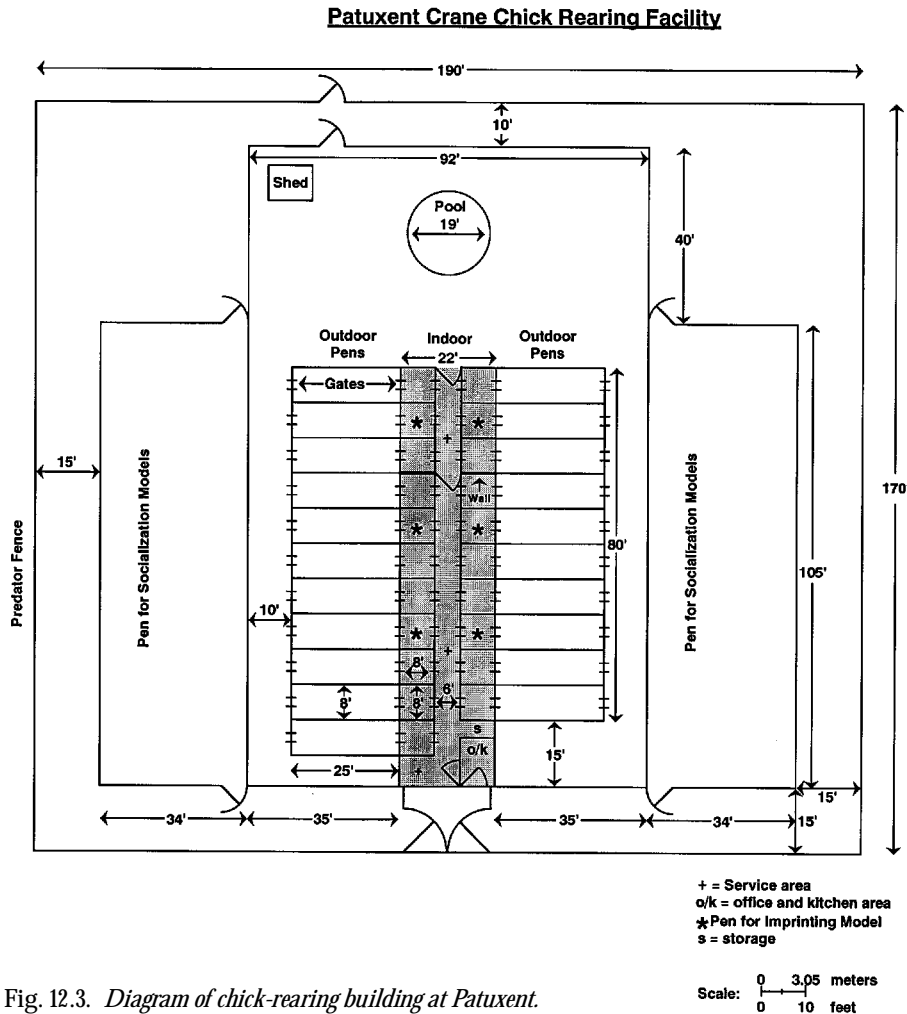


Fig. 12.3. Diagram of chick-rearing building at Patuxent.

during inclement weather. Pens should be placed in a series to provide visual contact between chicks or with imprinting models (adults).

Inside runs should be at least 2 m wide and 2-4 m long to provide adequate exercise space when the chicks are kept inside for much of the day. Outside runs should be the same width but 5 m or more long to give the larger chicks room to exercise. If the chicks do not have access to a large exercise area during the day, their outdoor pens should be larger (e.g., 2.5 x 8 m). Pens should be 2 to 2.5 m high, with net-covered outside runs.

The roof of the chick building should project (>0.5 m) over the edges of the outside runs and have a gutter to catch rainwater. This overhang prevents water from flooding the indoor pens and also provides additional shade.

Indoor chick pens should have **mesh no larger than 2.5 cm** separating adjacent pens. Chicks can get their heads caught in gaps larger than this, and small

predators such as weasels (*Mustela* sp.) or the paws of some larger mammals can pass through 3 cm gaps. **Vinyl-coated welded wire or chain link** can be used for the outside run, while finer mesh vinyl fencing is better for inside runs, where small, nervous chicks often pace the fence and would damage their bills on rougher fencing. If existing pens are constructed with uncoated chain link, hardware cloth, or chicken wire, attach smaller mesh (1-2.5 cm) vinyl fencing material or sheets of plexiglass to the lowest 0.5 m of fencing using stainless steel hog rings or plastic ties. These measures prevent injury from either aggressive chicks or imprinting models (adults) in neighboring pens.

Chick pens should not have **sharp projecting edges** and should be easily cleaned. **Eliminate gaps**

where chicks can get their heads, bills, or feet caught.

Ideally, a door operated remotely by a rope inside the service area allows chicks to pass between their indoor and outdoor runs. This door should be large enough to allow a person to pass through it while bending over (ca 50 cm x 120 cm). The door between the service area and the chick runs should have a window in it to allow observation of the chicks without disturbing them.

The indoor pens should have an overhead **light** and two electrical outlets for operating two **heat lamps** or space heaters. The heat lamps should be on separate circuit breakers to ensure that both heat lamps in one pen do not go out when one circuit fails. Small chicks should also have two lamps in case one bulb burns out (fails). Heat lamps, hung from the ceiling on **adjustable chains**, can be raised as the chicks grow taller. These lamps may be thermostatically controlled, or they can be adjusted in height above the ground and checked each day.

Clear or red 125 or 250 W heat lamps work well. Stone lamps, which do not produce visible light, should not be used for small chicks because the chicks may become chilled. Stone lamps are acceptable for chicks that are more than 40 days old.

Chick Exercise Yard. Hand-reared chicks benefit from a large pen (Figs. 12.4 and 12.5) where they can be socialized and exercised. This yard gives chicks running room and space for short practice flights. If the yard is **near the outside chick runs**, it is easy to transfer the chicks between their pens and the yard. The chick pens and exercise pen should have gates that stay open and have no high objects to step over. The exercise pen should have at least two gates, or one gate for every 50 m of perimeter. Several gates are needed for rapid pursuit of fledging chicks. Exercise pens should be at least 10 x 20 m, but preferably larger (20 x 40 m).

Walls of the exercise yard should be **soft, 2.5-cm mesh fence** like that recommended for outdoor chick runs. The pen need not have an overhead net if the chicks are well supervised (Fig. 12.5). The fence should be **1.2-1.5 m high** if it is not covered, or **2.5 m high** if it is flight netted. Place fence posts outside the fence so that they do not project into

the pen. Place **shade trees** in the exercise pen to protect the chicks on hot days. Small (1.5-2 m diameter) **pools** with washed gravel drainage systems allow the chicks to bathe and cool off during hot weather. These should be drained daily and disinfected periodically. The yard should have a grass or dirt surface. During warm or humid weather, remove grass clippings after mowing to prevent the growth of *Aspergillus*.

Swimming Pool. Swimming therapy (Fig. 12.6) can be used to provide exercise for any chick, but is especially important for chicks that show leg growth problems. The pool should be at least 0.7 m deep and 3-5 m in length to accommodate large chicks. The pool must have a filtration system, should be **cleaned regularly**, and be kept properly **chlorinated**. Pools may not be needed frequently so are optional for small crane breeding operations.

Chick Treatment Pens. The chick house should contain one or more pens where sick or injured chicks can be isolated from disturbance. This pen should be 2 x 1.5 m. The room should have a window for viewing the chick without disturbing it. The pen should have a ceiling-mounted **heat lamp or other heating system**.

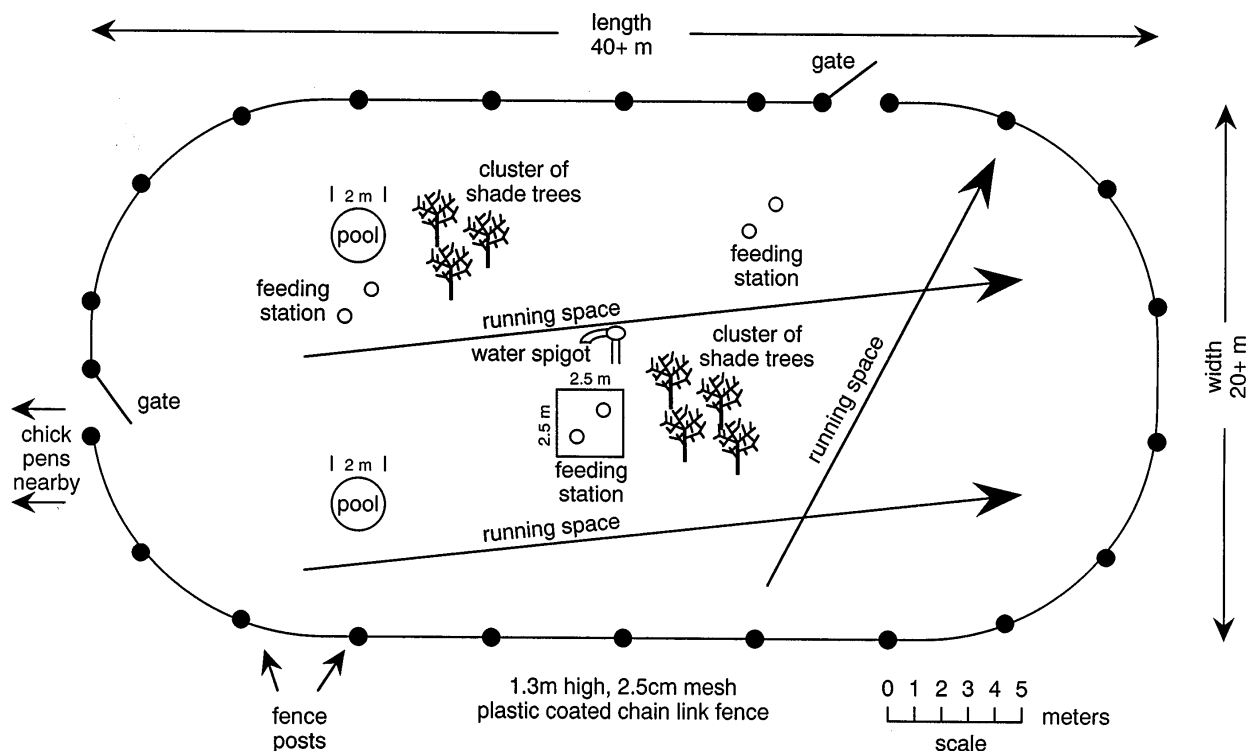


Fig. 12.4. Diagram of chick exercise yard at ICF.



Fig. 12.5. *The chick exercise yard at ICF is also used as an interactive center (Susan McDonald demonstrates use of the puppet head).*

Photo David H. Thompson



Fig. 12.6. *Marianne Wellington attends to chicks during aqua therapy.*

Photo David H. Thompson

Costume-rearing Facilities

The rearing of chicks in isolation from humans requires special facilities that allow people to hand raise chicks using the proper imprinting cues while preventing the chicks from seeing uncostumed humans (Chapters 5 and 11D). At ICF, the brooders and pens are equipped with **one-way mirrors** and small **feeding doors** within the larger door or wall to allow caretakers to see and reach the chicks without being seen themselves. Two one-way mirrors in the door are recommended: one 0.6 m high for watching the chicks during feeding, and one 1.2 to 1.4 m high to allow standing persons to easily check the chick's status. One-way mirrors must be covered with a lightweight opaque cloth to prevent the human caretaker from being seen by the chick when light intensity is greater in the service area than in the pen.

The door for feeding chicks (Figs. 12.2 and 12.7) has an 18-cm diameter hole covered by a small hatch that swings outward into the service area. This door should be 30 cm above the ground. A person should be able to reach nearly any place within the brooder through the feeding door or use a puppet to lure the chick near the door for capture.

At Patuxent, the pen walls along the service area are not opaque but are covered by a tennis-netting visual barrier. This allows the costumed caretaker to view the chicks easily, but it partially obstructs the chick's view of the caretaker. Chicks are fed by costumed caretakers in the pen itself. Small, plexiglass pet incubators are used for the chick's first day; they are kept covered by an opaque cloth unless a costumed caretaker is feeding the chick with a puppet head.

At ICF, we provide an **exercise yard** adjacent to the outdoor chick runs (Fig. 12.2) and accessible from each chick pen. Here chicks can also approach live adult socialization models, a pair or a few subadult conspecifics penned next to the exercise yard. The pen for these models should span the length of all the outdoor runs so that all chicks can see the cranes when outdoors (Fig. 12.3). This pen should be 10 m or less in width to ensure that the adults are near the outdoor runs and constantly in view. Because the adults will often try to attack the chicks, it is imperative to have either a two fence barrier (Fig. 11F.3) or a plexiglass or fine mesh wire barrier to separate the chicks from the adults.

The perimeter fence around the costume-rearing facility should be opaque or should have some visual barrier (e.g., tennis netting) to prevent the chicks from seeing uncostumed humans. Where practical, the costume-rearing pen complex should be adjacent to a wetland so chicks learn to use natural habitats.



Fig. 12.7. *Props for rearing a chick in isolation from humans*

Photo ICF

Parent-rearing Facilities

Pens in which adult cranes raise chicks must be modified for safety of caretakers and chicks. **Flight netting** (Fig. 12.8) is important in areas where aerial predators are a serious danger. Because chicks are not thermo-competent, they also benefit greatly from **shade and wind shelters** (Fig. 12.8). **Safety features** to protect caretakers from the extreme aggression of some parent cranes should be built into pens for chick rearing. Food and water can be supplied inside a shelter when the adults are locked out, or supplied outside when the parents are locked inside. Remotely operated (either vertically or horizontally) sliding doors (Fig. 12.9) are ideal for this purpose. Locking one or both parents in the shelter when their chick is still outside allows safer capture of the chick for health exams.



Fig. 12.8. A flight-netted pen showing shade shelter (near) and food/wind shelter (far).

Photo Jonathan P. Male

Chick safety is further facilitated by having the lowermost 30 cm of the fence covered by a fine mesh (0.5-2.5 cm) **chick-proof fencing** (Fig. 12.10) which also prevents chicks from getting their heads caught in the fencing or being injured by cranes in adjacent pens. This finer mesh should be extended at least 10 cm underground to prevent



Fig. 12.9. The sliding door on a Red-crowned Crane shelter can be operated remotely.

Photo Patty McCourt

the adult cranes from digging beneath it. An alternative chick proofing system uses opaque tennis netting that reaches all the way to ground level, preventing chicks from getting out below it (see Visual Barriers and Capture Corners).

Adult Crane Facilities

General Features of Crane Pens

Materials and Specifications. Recommended fence height for pens is 2.3-2.6 m. Vinyl-coated 16 gauge over 11 gauge **chain link** is the safest fencing material for large cranes. Perhaps the most practical fencing material for crane pens is 5 cm mesh 11 gauge **galvanized steel chain link** (Fig. 12.10). **Aluminum chain link** is much more expensive but is otherwise better than **galvanized steel** because it is smoother and causes fewer injuries. Specify “knuckled” when ordering chain link to avoid the hazard of twisted barbs at the top or bottom of the fence. **Poultry wire** (16 ga or thicker) is an acceptable alternative but causes more cuts to bills, wings, and legs. Small mesh (2.5 cm) poultry wire causes fewer injuries and is stronger than 5 cm mesh.

Fences should be supported at 5 to 8 m intervals with steel posts or pressure-treated (i.e., rot resistant) wooden posts preferably set in concrete. Where possible, **posts** should be placed along the outside of the pen or at least outside of two walls of the pen. Corner posts require stabilization. Place support braces outside



Fig. 12.10. Chain link fence. Photo also illustrates 0.5 m tall chick guard, and Patuxent's new double-door, feed/wind shed.

Photo George F. Gee

the pen whenever possible; when they must be inside a pen, make sure they lie against the fencing so that a crane's head or foot cannot be caught behind them.

Toprite XL, 5 cm mesh, nylon **flight netting** is recommended (see Appendix). For smaller species, 2.5-3 cm netting is recommended. Flight netting is attached to the pen perimeter and supported by **guy wires** crossing the pen at 4-5 m intervals (Figs. 2.1 and 12.8), depending on the size of the pen and anticipated snow loads. Larger pens require more guy wires per unit area, and require internal fence posts at 8 m intervals to support the guy wires. Guy wires should always cross fence boundaries over a fence post.

Flight netting must be **strung tightly** and without gaps along the fence and guy wires. The flight netting should be attached at 5 to 10 cm intervals along fence tops. Some institutions leave the netting free along the wires, while others attach it to wires at 20-40 cm intervals using small hog rings. If you anticipate the need to remove a net, secure it by hooking it over the cut end of the chain link fabric (Fig. 12.11). Otherwise, stainless steel hog rings or plastic clips can be used to permanently attach flight netting to fences.

Visual Barriers and Capture Corners. Some pairs of cranes require visual barriers on one or more sides of their pens to reduce **disturbance** from humans or neighboring cranes. Visual barriers also help prevent injuries when introducing cranes into new pens or when capturing cranes. Barriers can be made of **tennis netting** (Fig. 12.12) or **reed mats** tied or clipped to the fence. The flexibility of these surfaces helps prevent trauma to cranes when they collide with the fence or try to attack something on the other side. In cases of extreme aggression between neighboring cranes, a visual barrier between them must be combined with a gap of more than 1 m between the adjacent pens to prevent stress-related pacing or fence pecking. Ropes or cables are the best permanent attachment system, but clips are preferred for temporary attachment.

Cranes that are handled frequently for AI or some other purpose may require a **capture corner**. This is a portion of the pen wall that is padded or covered with a soft material (such as tennis netting) to prevent cranes from becoming injured during the capture process (Figs. 2.4 and 12.12). Discarded Christmas trees tied to the fence extending 3 m in each direction from a corner work well for this purpose.

Landscaping. Cranes adjust better to pens with **natural cover and shade**. In netted pens, shrubs or artificial shade structures are needed. Tennis netting or



Fig. 12.11. Nets are attached for easy release by hooking the mesh over the cut-ends of the chain link fabric. Photo George F. Gee

reed mats with an area of 10 m² attached to the top of the flight netting works well in summer; remove them each autumn before the first snow. A 2.5 x 2.5 m aluminum shade roof (Fig. 12.8) supported by four posts is an alternate method. Open-topped pens allow for larger shade trees. Landscape pens to prevent **standing water** where pathogens are likely to develop.

Cranes prefer seclusion during the breeding season so it is best to **mow grass after the breeding season**. If short grasses (like buffalo grass, *Buchloe dactyloides*) are used, little or no mowing is required.

Predator Proofing. Crane pens should be built to exclude digging predators (dogs, foxes, etc.) and climbing predators (raccoons). Pens should be surrounded by a perimeter fence with **electrical wires** (Figs. 11F.2 and 11F.3). The wires can be supported by fiberglass rods or attached to insulators on support brackets projecting from the fence posts or chain link fabric. At ICF, electrical and ground wires alternate at 5 cm intervals on brackets that extend 30 mm out and 45° up from the point of attachment.

The perimeter fences (Fig. 11F.3) of crane pens should be **buried** 0.5 m in the ground and be back-filled with 5 mm washed gravel. The base of the perimeter fence includes a 50-cm wide skirt of 2-cm mesh fencing **extending horizontally** outside the perimeter fence to deter digging predators.

Shelters. A shelter provides a dry place for the food and protects the cranes during inclement weather. For cold-hardy cranes a **3-sided shelter** is sufficient. A fully **closed shelter** (Fig. 12.12) is useful for locking cranes inside during heavy snow or ice storms, when pen repairs are needed, for medical

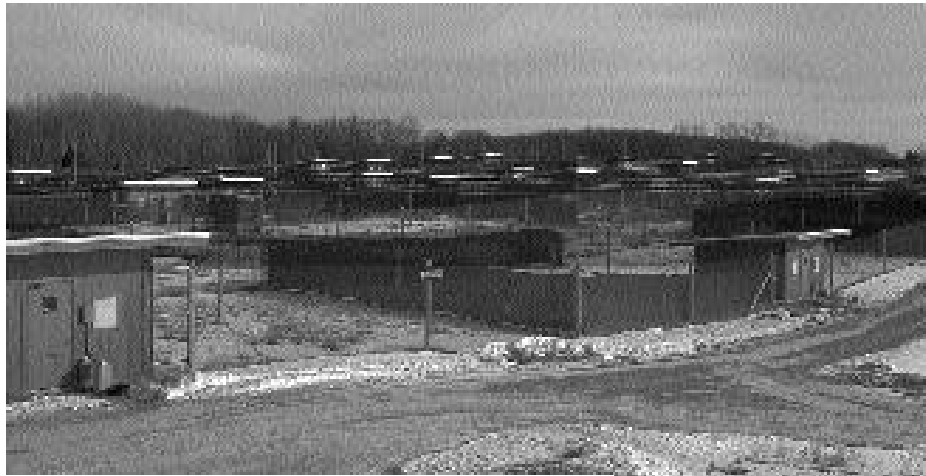


Fig. 12.12. Crane City at ICF. Breeding pens are provided with tennis netting visual barriers that also create non-abrasive capture corners.

Photo David H. Thompson

confinement, or during brief periods when there is excessive risk of nocturnal predators. If the shelter is primarily or solely a feeding station, only a simple structure that keeps the food dry is required (see Fig. 12.10).

If warm climate cranes are kept at temperate latitudes, they require **insulated shelters** with ca 10 m² of floor space per crane. Some cranes require heated shelters. A 1300-1500 W heater hung from the ceiling is enough to heat a 4 x 4 m insulated shelter (see Table 2.1 for guidelines on when to lock cranes inside or provide heat). Insulated shelters also offer the advantage of staying cooler during the summer. If cranes will be confined for days or weeks at a time (e.g., during quarantine), shelters must be well ventilated.

Shelters should have sloping roofs and adequate drainage. A non-breakable window should be provided for viewing cranes and for natural light when cranes are locked inside. An overhead light may also be necessary for some servicing activities and to provide supplemental light during extended confinement. **Sliding guillotine doors** (Fig. 12.9) operable by ropes or wires from outside the pen are useful for controlling access without having to enter the pen.

Some shelters require **floors**. Concrete is both durable and easy to wash, but requires a soft bedding (e.g., 5 cm of wood shavings or sand, or 10-15 cm of wood shavings during extremely cold weather). A heat pad may be buried in the floor if the climate demands it. Slope the floor slightly toward the main service door to provide drainage.

Special Pen Features.

Some cranes may breed better with a pool in their pens. Pools can be constructed of concrete or plastic. The floor of the pool should slope gradually to a depth of 20 to 60 cm. Each pool should have a **drain**, unless water flow is sufficient to prevent stagnation. Pools with natural vegetation are more likely to stimulate normal reproductive behavior. Pools

without water circulation should be drained and cleaned every few days. Even pools as small as 3 m in diameter may be effective. At Patuxent, continuously **flowing water** is provided in stainless steel cups (Fig. 12.13).

Overhead **sprinklers** (Fig. 3.3) controlled by timers have been used to simulate a **rainy season** (LaRue 1979) and promote breeding in Brolgas. A durable hosing such as 16-mm polyvinylchloride (PVC) must be used if the cranes can reach the pipes. The hose can be attached to overhead wires and have **sprinkler heads** connected at ca 8-m intervals to sprinkle a large area. Be sure that sprinkled pens have **adequate drainage**.



Fig. 12.13. A specially fabricated stainless steel VanEs water cup (15 cm x 15 cm) and Sandhill Crane. Water rises from a supply line at the base of an inverted cone, and drains through holes at water line. Each supply line has a valve accessible through the drain line.

Photo David H. Ellis

Photoperiod lights (Figs. 2.1, 2.3, and 3.2) are sometimes needed to simulate the long days experienced by arctic and subarctic nesting cranes and thereby promote breeding. The recommended **light intensity** at ground level is an average of 16 foot-candles (Gee and Pendleton 1992). However, lights are somewhat effective at intensities as low as 1 foot-candle. Either many small light bulbs or one large one can be used. Ten 125 W incandescent light bulbs mounted 2.3 m high (just below the flight netting) around the perimeter of a pen or two 400-1000 W metal halide bulbs mounted 8-10 m high in opposite corners of the pen will adequately illuminate a 200-300 m² pen. Be aware that light spillage into adjacent pens may affect non-target birds. Metal halide bulbs are very expensive, but they use less energy than incandescent bulbs. **Electric timers** can be used to control the duration of artificial light.

Types of Pens

Breeding Pens. Paired cranes should be provided with two adjacent outdoor enclosures and a shelter accessible from both outdoor pens and from a service area. This arrangement allows for annual **pen rotation** (see Chapter 2). Separate pens allow paired birds to be separated if they become temporarily incompatible (due to injury, etc.). We recommend that breeding pens be **at least 150 m² for smaller species**, and **200-300 m² for larger cranes**.

For some purposes, flight-netted pens are required. We recommend that flight netting be 2.5 m high. This height is a compromise: if nets are higher cranes can do more flying and dancing, but they can also attain higher speeds thereby increasing the chance of injury. In regions where there are no aerial predators of concern, pens may be left unnetted for flightless cranes.

Some pairs benefit from larger pens. Birds that are full-winged and fly frequently should be kept in smaller pens to prevent injuries. Nervous birds that require **more privacy** may breed better if given larger pens with more visual barriers along the perimeter and screening vegetation within the pen. If the pens are in rows, leave a **5-10 m buffer zone** between rows.

Group Pens. Subadult cranes may be kept in groups of 2-15 in larger pens, such as 30 x 60 m. Standard breeding pens are also adequate for small groups (2-5) of compatible subadults. The pen size should be proportionate to the number of cranes occupying it. Group pens must have **two or more feeding stations and water sources** (see Chapter 6).

Pens should have an 80-100 cm wide **service gate** through which mowers and other equipment can pass. Gates should not have a sill that would impair a hasty exit when escaping from an aggressive crane or that would impede movement of equipment.

Pairing Pens. Pens intended for use in establishing **new pairs** should be built so birds can be **easily moved between pen halves**. Doors between the pens should open a full 180°. This enables nervous birds to be moved between adjacent pens without handling and allows a crane to retreat to its own pen if it fears its prospective mate. If the cranes share a common shelter, it is useful to have a door between the two halves of the shelter. Pairing pens should be at least 15 x 15 m. If pairs of cranes have two pens to allow for rotation, place cranes in **alternate pens** rather than in the halves of a single pen. If this arrangement results in same sex birds sharing a common fence, a visual barrier is normally required.

Exhibit Pens. Because cranes that are on display are frequently not intended to breed, their pens have fewer design constraints. If exhibit cranes are intended to breed, their pens must provide a “safe” place for the nest. Elongated pens (Fig. 12.14) allow cranes to stay further from the public and are more likely to result in breeding (see Chapter 6).

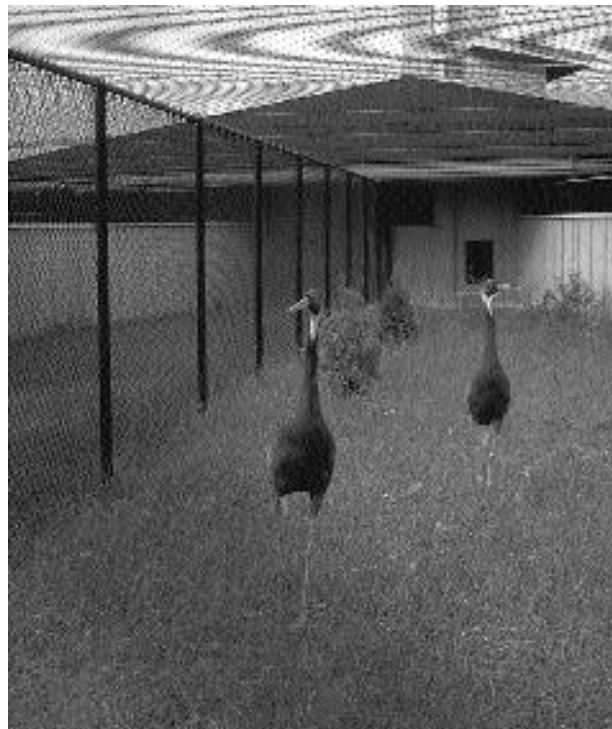


Fig. 12.14. *The display pod at ICF showing a wedge-shaped pen with a pair of White-naped Cranes.*

Photo David H. Ellis

Exhibit pens should allow the public to view cranes without overly disturbing the birds. Breeding pens, described earlier, can make good display pens if no visual barriers are erected on the public viewing end. When people approach, some cranes choose to stay inside their shelters and must be locked outside during viewing hours. Feeding the cranes at the viewing end of the pen encourages them to stay near the visitors. This practice also confines disturbances to one area, leaving the back of the pen secluded for nesting.

Cranes can also be encouraged to stay closer to the public if their shade is near the public end of the pen. For pens that have elevated viewing points, use the type of shade that does not block the view of the cranes. Moated pens or pens with elevated viewing points make good displays. However, elevated viewing points offer less of an opportunity to appreciate the size of cranes.

Tall grass and shrubs obscure the view of cranes but help make the pen look more natural and can be placed to promote a sense of security in the resident cranes. Water pools (Fig. 3.5) make pens appear more natural and allow cranes to exhibit their aquatic tendencies, but may increase the risk of disease (see Chapter 3).

Special Facilities

Quarantine Facilities

New arrivals are quarantined before being housed near other cranes. Ideally, the quarantine facility should be located at least 1 km **away from other crane pens** and should be serviced by separate personnel or be **the last area serviced each day**. The walls and floor should be **easy to disinfect**. A **sealed concrete floor** is advisable. The quarantine facility should either have **no outside run** to avoid long-term contamination of the soil, or the contaminated soil should be left idle for at least one year before reuse (see Chapter 2).

Pens for Holding Release Cranes

Crane release projects usually require an acclimation pen where the cranes are kept for the last few weeks before transfer to a release site. These pens are normally flight-netted and should be large enough to

allow for the entire release cohort (5-15 birds) to be housed together. The details of pen design depend on the type of release (see Chapter 11D).

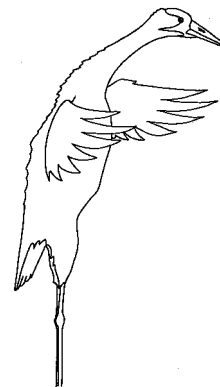
Veterinary Facilities

The hospital facility should include a small **surgery room**, and a series of small **treatment pens** (ca 3 x 2 m) with outdoor runs (ca 5 x 2 m) where cranes can be readily captured for frequent treatments. These pens should have lights and heat lamps. A small (ca 3 x 2 m), easily darkened **recovery room** (may be the same facility as the treatment rooms) should have indoor-outdoor carpeting with a thick rubber backing.

Crane **hospital equipment** should have as a minimum: a walk-on scale with 125 kg capacity, a gram scale with 0.01 g accuracy, a 400 X (preferably 1000 X) microscope, a micro-hematocrit centrifuge, and an autoclave. Larger facilities should have: a radiograph machine, anesthesia equipment, a centrifuge, a medical laboratory that contains additional diagnostic equipment, and an incinerator.

Literature Cited

- Gee, G. F., and G. W. Pendleton. 1992. Effect of extended photoperiod on crane reproduction. Pages 149-159 *in* D. A. Wood, editor. Proceedings 1988 North American Crane Workshop. Florida Nongame Wildlife Program Technical Report #12.
- LaRue, C. 1979. Breeding the Brolga. *Aviculture Magazine* 85(3):139-140.



Ecology, Status, and Conservation

Curt Meine and George W. Archibald



Cranes constitute one of the world's most endangered families of birds. At the same time, their cultural value, high visibility, extraordinary beauty, dramatic migrations, and striking behavior have inspired widespread conservation efforts. Cranes often serve as “umbrella” and “flagship” species in conserving wetlands and grasslands around the world. As such, they draw attention to, and provide protection for, a broad array of species and ecosystems (Schoff 1991). Cranes have stimulated innovative conservation measures at the international level (Lewis 1991), while also providing a focus for local conservation programs (e.g., Harris 1994a, 1994b). Captive propagation and reintroduction programs have provided important experience in the conservation not only of cranes, but other endangered species as well. Cranes have also proven to be effective in focusing information for environmental education programs (e.g., Dietzman and Swengel 1994; Landfried et al. 1995).

In all of these areas, cranes present excellent opportunities to develop programs that combine varied conservation goals, activities, and techniques. Such integrated programs will become even more vital as cranes face growing challenges in a world of accelerating environmental change.

This chapter reviews the status of the fifteen species of cranes, assesses current conservation activities, and identifies future conservation needs. Table 13.1 provides a summary of population estimates and population trends. Our species accounts are derived from *The Cranes: Status Survey and Conservation Action Plan* (Meine and Archibald 1996), which was compiled in consultation with the Crane Specialist Group of the World Conservation Union (IUCN) and Birdlife International. We thank the many individuals around the world who contributed information, advice, and text for the species accounts.

Black Crowned Crane

The Black-Crowned Crane (Fig. 1.1) inhabits the Sahel and Sudan Savanna region of Africa from the Atlantic coast to the upper Nile River basin (Fig. 13.1). Two subspecies are recognized. *B. p. pavonina* (the West African Crowned Crane), with an estimated population of 11,500–17,500, occupies the western part of this range and is divided into eight or more disjunct populations. *B. p. ceciliae* (the Sudan Crowned Crane), with an estimated population of 55,000–60,000, occurs in eastern Africa, with the largest concentrations in southern Sudan (Urban 1996). Historically, the Black Crowned Crane was more numerous and more evenly distributed than at present. In the eastern part of its range, the species remains relatively abundant. In the western portions of the range, however, both its numbers and its range have been reduced dramatically over the last two decades (Mustafa and Durbunde 1992 unpubl.). The species is classified as Vulnerable under the revised IUCN Red List criteria. *B. p. pavonina* is classified Endangered, and *B. p. ceciliae* Vulnerable.

Black Crowned Cranes use both wet and dry open habitats, but prefer a mixture of shallow wetlands and grasslands (especially flooded lowlands in the sub-Saharan savannas). They are both year-round residents and local migrants, flocking together during the dry (non-breeding) season and moving from large permanent wetlands to smaller temporary wetlands during the rainy season. Although they are non-migratory, daily and seasonal movements may, in some areas, range up to several dozen kilometers (Urban 1981).

The principal threat facing the Black Crowned Crane is the loss, transformation, and degradation of its habitat (Tréca 1996). Behind this threat lies a combination of causal factors: (1) extended drought in the Sahel and sub-Saharan savannas, (2) expanding

TABLE 13.1

Population estimates for crane taxa.¹

Species Subspecies, population, or wintering population	Number	Trend
Black Crowned Crane		
<i>B. p. pavonina</i>	11,500-17,500	Declining. Extirpated (or nearly extirpated) in some range countries.
<i>B. p. ceciliae</i>	55,000-60,000	Unknown. Generally stable, but possibly declining locally. Still fairly abundant in Sudan.
Total	66,500-77,500	Declining
Gray Crowned Crane		
<i>B. r. gibbericeps</i>	85,000-90,000	Declining
<i>B. r. regulorum</i>	<10,000	Unknown
Total	85,000-95,000	Declining
Wattled Crane		
South African population	250-300	Declining
South-central African population	13,000-15,000	Declining
Ethiopian population	several hundred	Unknown
Total	13,000-15,000	Declining
Blue Crane		
Southern population	21,000	Declining
Namibia (Etosha Pan) population	<100	Stable
Total	21,000	Declining
Demoiselle Crane		
Atlas population (N Africa)	<50	Declining
Black Sea population	~500	Declining
Turkey population	<100	Unknown
Kalmykia population	30,000-35,000	Stable
Kazakhstan/Central Asia population	100,000	Stable to increasing
Eastern Asia population	70,000-100,000	Stable to declining
Total	200,000-240,000	Stable
Siberian Crane		
Eastern population	2,900-3,000	Unknown.
Central population	5?	Steadily declining. Not observed on the traditional wintering grounds in India since the winter of 1992-93.
Western population	10	Holding at 8-14 birds on the wintering grounds since mid-1980s. Highly vulnerable.
Total	2,900-3,000	

TABLE 13.1 CONTINUED

Population estimates for crane taxa.¹

Species Subspecies, population, or wintering population	Number	Trend
Sandhill Crane		
<i>G. c. canadensis</i> and <i>G. c. rowani</i>	~450,000 ²	Probably stable.
<i>G. c. tabida</i>	65,000-75,000	Unknown due to difficulty in distinguishing from Lesser Sandhill Cranes; probably stable.
<i>G. c. tabida</i>	65,000-75,000	Increasing rapidly in the eastern portion of its range. Generally stable elsewhere. Some western populations may be declining.
<i>G. c. pratensis</i>	4,000-6,000	Increasing rapidly in the eastern portion of its range. Generally stable elsewhere. Some western populations may be declining.
<i>G. c. pulla</i>	120	Generally stable, with local increases and declines. Includes the Okefenokee portion of the population (about 400 individuals).
<i>G. c. nesiotis</i>	300	Numbers in wild increasing through augmentation. Reproduction in the wild is below replacement level.
<i>G. c. nesiotis</i>	300	Generally stable. New populations recently discovered.
Total	520,000	Stable to increasing
White-naped Crane (winter counts)		
Japan (Izumi)	1,800-2,100	Increasing
Korean Peninsula	100-200	Decreasing
China (Poyang Lake)	~3,000	Unknown
Total	4,900-5,300	Stable to decreasing (based on loss of breeding habitat)
Sarus Crane		
<i>G. a. antigone</i>	8,000-10,000	Declining
<i>G. a. sharpii</i>	500-1,500	Unknown; likely declining
<i>G. a. gilli</i>	<5,000	Unknown
Total	13,500-15,500	Declining
Brolga		
Total	20,000-100,000	Stable through most of its range. Decreasing in southeastern Australia.
Eurasian Crane		
West European population	60,000-70,000	Stable to increasing
East European population	>60,000	Stable to increasing
European Russia population	~35,000	Decreasing
Turkish population (non-migratory)	200-500	Decreasing
West Siberia population	~55,000	Decreasing
C Siberia/NE China population	5,000	Decreasing
Tibetan Plateau population	1,000	Stable
Total	220,000-250,000	Increasing overall, but with local declines

TABLE 13.1 CONTINUED

Population estimates for crane taxa.¹

Species Subspecies, population, or wintering population	Number	Trend
Hooded Crane (winter counts)		
Hubei (China)	up to 425	Unknown
Dongting Lake (China)	up to 200	Unknown
Poyang Lake (China)	up to 360	Unknown
Shengjin Lake (China)	300	Stable, but habitat declining
West Taegu (South Korea)	180-250	Unknown
Yashiro (Japan)	50	Declining
Izumi (Japan)	~8,000	Stable
Total	9,400-9,600	Stable
Black-necked Crane (winter counts)		
NE Yunnan/W Guizhou	1,300-1,600	Unknown
NW Yunnan	<100	Stable to declining
SC Tibet	3,900	Stable
E Tibet	<20	Declining
Bhutan	360	Stable
India-Arunachal Pradesh	<10	Declining
Total	5,600-6,000	Stable but vulnerable
Red-crowned Crane (winter counts)		
China	600-800	Unknown
North Korea	300-350	Increasing
South Korea	200-300	Unknown
Japan	594	Stable to increasing
Total	1,700-2,000	Stable to decreasing (based on loss of breeding habitat)
Whooping Crane (as of August 1995)		
Aransas-Wood Buffalo population	150	Increasing slowly.
Rocky Mountain population	4	Decreasing.
Florida population	24	Increasing through artificial augmentation.
Wild sub-total	178	
Patuxent	78	
ICF	44	
Calgary Zoo	19	
San Antonio Zoo	4	
Captive sub-total	145	
Total	324	Slowly increasing

¹ For details of proposed population status categories and criteria see IUCN (1994) and Meine and Archibald (1996).

² In mid-continental population estimates, Lesser and Canadian Sandhill Cranes are not distinguished. Also a relatively small number of Greater Sandhill Cranes are included in the total. Estimates are based on 3-year running averages of spring counts conducted on the Platte River during migration. The figure given here represents the 1995 survey results for the mid-continental populations (420,866) plus about 25,000 Lesser Sandhill Cranes from California.

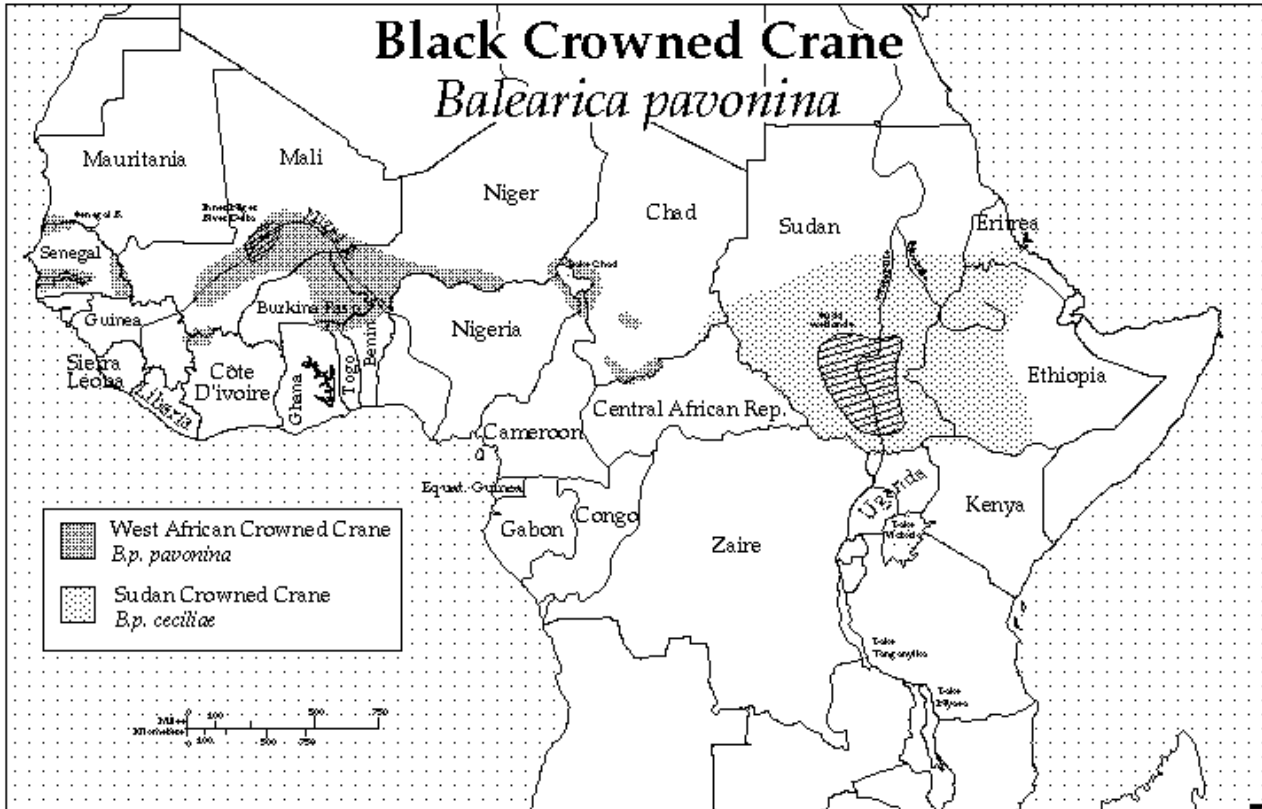


Fig. 13.1. Distribution of the Black Crowned Crane.

human populations, (3) intensive agricultural development and expansion, and (4) extensive changes in hydrological systems as a result of dams, drainage, and irrigation projects (Fry 1987; Daddy and Ayeni 1996). These factors are most pressing in West Africa, but also affect the species in the east. In some areas, these cranes are hunted for meat or captured and sold for trade. Ineffective law enforcement and the lack of long-term population monitoring leave the species in jeopardy.

The decline of the Black Crowned Crane in West Africa has begun to stimulate conservation efforts on behalf of the species. It is legally protected in most countries where it occurs, and many of the protected areas in these countries harbor cranes. Several local surveys have recently been undertaken. In 1992, Nigeria hosted an international conference on the Black Crowned Crane and its habitat. A Black Crowned Crane Coordinating Centre was established as a result. No reintroduction program has been undertaken for the species, but the potential for reintroduction of the West African subspecies is under discussion, and an experimental release took place in Nigeria in 1992 (Garba 1996).

Priority conservation needs for the Black Crowned Crane include: (1) listing the species under CITES Appendix I; (2) ratification of the Ramsar Convention by countries within the species' range and adoption of stronger national wetland protection policies and legislation; (3) mandatory assessment for environmental impact of all large-scale land development schemes affecting Black Crowned Crane habitat; (4) increased support for existing protected areas and designation of new areas used by cranes; (5) ecological research on wetlands and crane habitat requirements; (6) a coordinated surveying and monitoring program for the species; (7) collaborative projects involving local communities in the conservation and sustainable use of wetlands; (8) establishment of a West African Crowned Crane Recovery Team; (9) development of educational programs involving Black Crowned Cranes and wetlands; and (10) expanded training opportunities for crane and wetland conservation specialists.

Gray Crowned Crane

The Gray Crowned Crane (Fig. 13.2) is the most abundant of the resident African cranes. Although precise population numbers are not available, recent estimates place the total population at 85,000-95,000 (Urban 1996), down from more than 100,000 over the last decade. It no longer occurs in certain portions of its historical range, especially the drier areas (Fig. 13.2). Two subspecies are recognized. Most are *B. r. gibbericeps* (the East African Crowned Crane). This race occurs in East Africa from northern Uganda and Kenya south to Zimbabwe, Botswana, and Namibia. *B. r. regulorum* (the South African Crowned Crane) is found in Zimbabwe and South Africa. The species is classified as Vulnerable under the revised IUCN Red

List criteria. *B. r. regulorum* is classified Endangered, and *B. r. gibbericeps* Vulnerable.

Gray Crowned Cranes use mixed wetland-grassland habitats for nesting and foraging, and, along with Black Crowned Cranes, are the only cranes able to roost in trees. The species' feeding strategy (i.e., generalist) has allowed it to adjust to human settlement and activity; most populations in East Africa now live in human-modified habitats (Pomeroy 1987). Abundance and distribution of food and nest sites are the key ecological factors determining the size of the home range. These, in turn, are largely influenced by local rainfall. Gray Crowned Cranes are non-migratory, but undertake local and seasonal movements in response to changing water conditions and food availability (Gichuki and Gichuki 1991; Gichuki 1993).

Although Gray Crowned Cranes and people have long coexisted, population declines over the last decade

reflect widespread threats to the species' habitat due to rapid human population growth, drought-related changes in land use, and intensified agricultural practices (Archibald 1992a). Loss and deterioration of wetland breeding habitat constitute the most significant threat to the species. Other problems include increased use of agricultural pesticides, declines in the fallowing of croplands, high rates of wetland sedimentation due to deforestation, and altered flooding regimes due to dam construction. The capturing of Gray Crowned Cranes for domestication and for export is also a serious threat (Katondo 1996).

Many native people revere the Gray Crowned Crane as sacred and strictly protect it. No range-wide surveys of the species have been undertaken, but crane counts

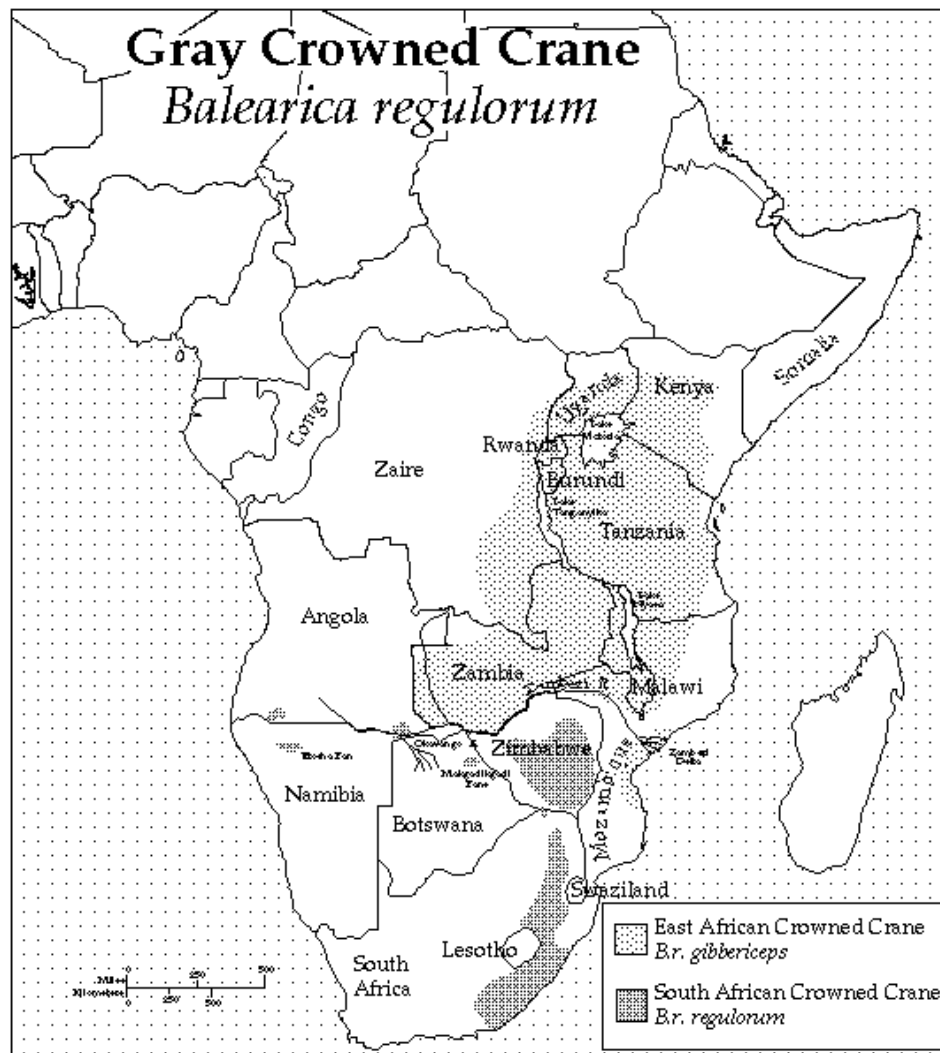


Fig. 13.2. Distribution of the Gray Crowned Crane.

and localized surveys have been undertaken intermittently in several countries. In recent years, field studies have begun to provide basic biological information, although our knowledge of this species remains relatively limited compared to other cranes. The increasing number and effectiveness of protected areas, especially in East Africa, has benefitted the species (Pomeroy 1987). However, most Gray Crowned Cranes nest and forage outside protected areas, so the overriding conservation challenge is to develop sustainable alternatives to the overexploitation of non-reserve wetlands. This goal has stimulated many community-based wetland conservation projects and several nationwide crane and wetland conservation plans (Wanjala 1996). Non-governmental organizations have often played a key role in these efforts.

Priority conservation measures for the species include: (1) listing of the species under CITES Appendix I; (2) strengthening laws to restrict trade and protect wild cranes; (3) expanding community-based wetland conservation programs; (4) designating additional reserves to protect key breeding areas; (5) developing and implementing national crane and wetland conservation plans and more specific management programs for key breeding habitats outside protected areas; (6) organizing national-level crane counts and long-term monitoring programs; (7) implementing research on the basic ecology of the species (e.g., critical habitat, local and regional movement patterns, and the extent of crop damage); and finally, (8) developing broad-based public awareness programs.

Wattled Crane

contributed by Ann Burke

The Wattled Crane (Fig. 1.3) is the largest and rarest of the six African cranes. The three main populations are in south-central Africa, with smaller populations found in Ethiopia and South Africa (Fig. 13.3). Over the last several decades, the species has been declining over much of its range. The total population estimate of 13,000–15,000 has remained constant over the last decade, but this is due largely to the discovery of ca 2,500 birds in Mozambique in the early 1990s (Urban 1996). Historically, the species was more abundant and more widely distributed across southern Africa,

with the greatest losses occurring in South Africa (Brooke and Vernon 1988). The species as a whole is classified as Endangered under the revised IUCN Red List criteria. The South Africa population is Critically Endangered.

The Wattled Crane is the most wetland-dependent of Africa's cranes. The extensive riparian wetlands of southern Africa's large river basins (especially the Zambezi and Okavango) are their preferred habitat, but they also use smaller upland wetlands throughout their range (Konrad 1981). The Ethiopian birds may make greater use of drier habitats during the non-breeding season. Nesting pairs establish large (often >1 km²) territories, generally in shallow wetlands with minimal human disturbance. Their diet consists primarily of aquatic vegetation, but in drier habitats also includes seeds, insects, and waste grain. Wattled Cranes are non-migratory, but do undertake irregular local movements in response to water availability (Urban and Gichuki 1991).

Loss and degradation of wetlands constitute the most important threats to the species (Macdonald 1989; Allan 1994). Habitat loss in South Africa is due mainly to intensified agriculture, dam construction, industrialization, and other pressures. In other portions of the range, dams and other water development schemes have caused fundamental changes in the species' floodplain habitats (Beilfuss 1995). Human disturbance at or near breeding sites is also a major threat (Eksteen 1996); breeding success declines when human settlements are too close to wetlands. Because Wattled Cranes occasionally forage on agricultural fields alongside Blue and Gray Crowned Cranes, they are also vulnerable to poisoning (Allan 1994).

Conservation measures have been undertaken most extensively in South Africa, but are increasing in other range countries. These measures include: (1) strict legal protection; (2) establishment of protected areas in key wetlands, especially in Zambia, Namibia, and Botswana; (3) identification and communication of appropriate habitat conservation practices for farmers and other private landholders; (4) marking and relocation of electrical utility lines; (5) expanded counts and surveys (especially since the early 1980s); (6) expanded research, especially in South Africa, Zambia, and Namibia; (7) establishment, in 1982, of a Wattled Crane Steering Group in South Africa; and (8) development (mainly by non-governmental organizations) of education and public awareness programs. A limited release program for the species has been initiated in South Africa.

Priority conservation measures for the species include: (1) enforcing existing protective legislation; (2) strengthening key protected areas, especially in the Bangweulu Swamps and Kafue Flats in Zambia; (3) identifying additional areas of critical habitat for protection; (4) assessing large-scale habitat threats (mainly from water development schemes) in the Kafue Flats, Okavango Delta, Makgadikgadi Pans, and Zambezi Delta; (5) developing a coordinated program for the protection of breeding habitat on privately owned farmland; (6) organizing a range-wide census and local Wattled Crane counts; (7) expanding field research outside South Africa; and (8) developing education programs aimed at private landowners, farm laborers, and students.

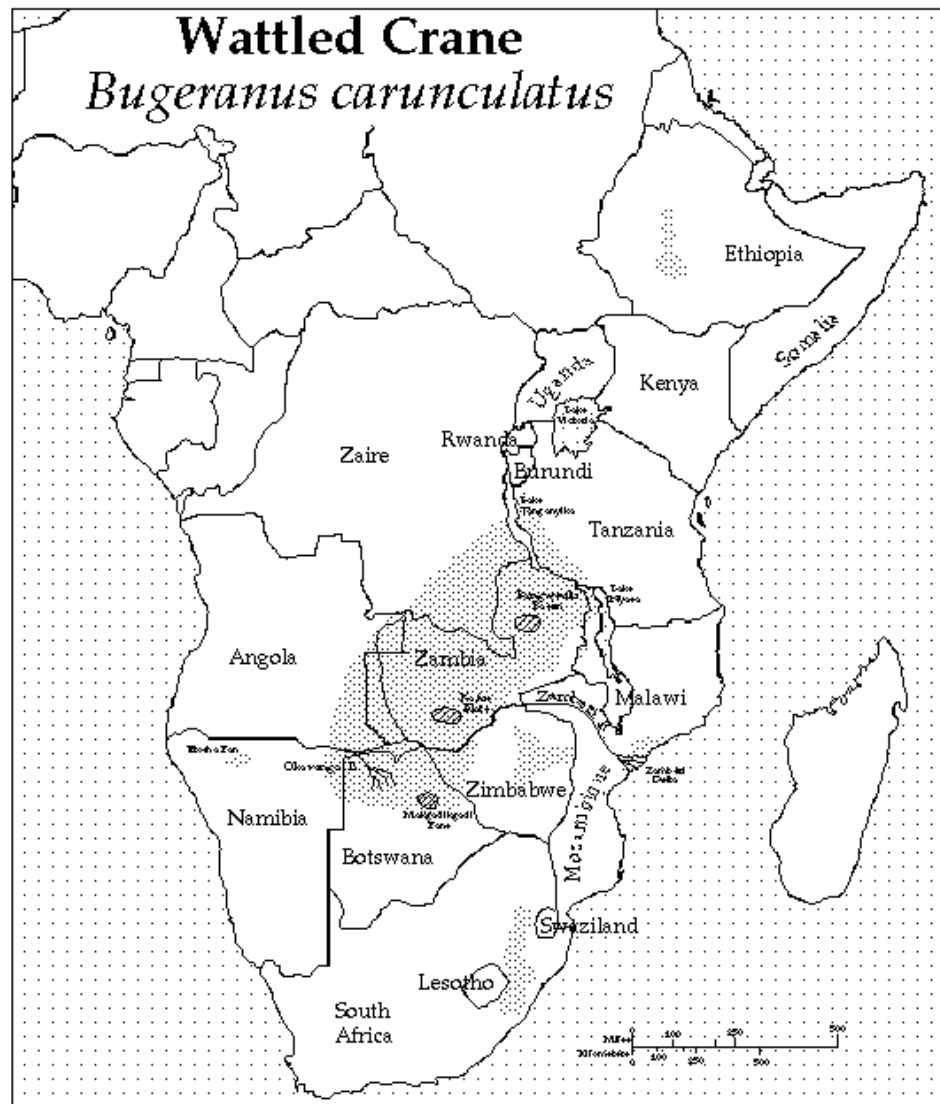


Fig. 13.3. Distribution of the Wattled Crane.

Blue Crane

The Blue Crane (Fig. 1.4), the national bird of South Africa, is still abundant in parts of its historical range (Fig. 13.4), but has experienced significant declines in many areas over the last twenty years. Its distribution is the most restricted of the fifteen crane species. It is endemic to southern Africa, with the vast majority of the population occurring in eastern and southern South Africa (Allan 1993). A small disjunct population occurs in the Etosha Pan of northern Namibia, while breeding pairs are occasionally found in five other countries. As recently as 1980, there was little concern about the Blue Crane from a conservation standpoint.

Since then, however, the species has largely disappeared from the Transkei region, Lesotho, and Swaziland. In other areas, including eastern Cape Province, Natal, northern Orange Free State, and Transvaal, populations have declined by as much as 90% (Allan 1994; Urban 1996). The total population is estimated at 21,000 and is declining (Allan 1993). Due to its rapid decline, the species is classified as Critically Endangered under the revised IUCN Red List criteria. Both the main South African population and the Namibian population (because of its limited numbers) are Critically Endangered.

The Blue Crane is primarily a bird of dry, upland grasslands. In South Africa, the species occurs in the grassland, Karoo, and fynbos biomes. Blue Cranes use natural grass- and sedge-dominated habitats in these biomes for both

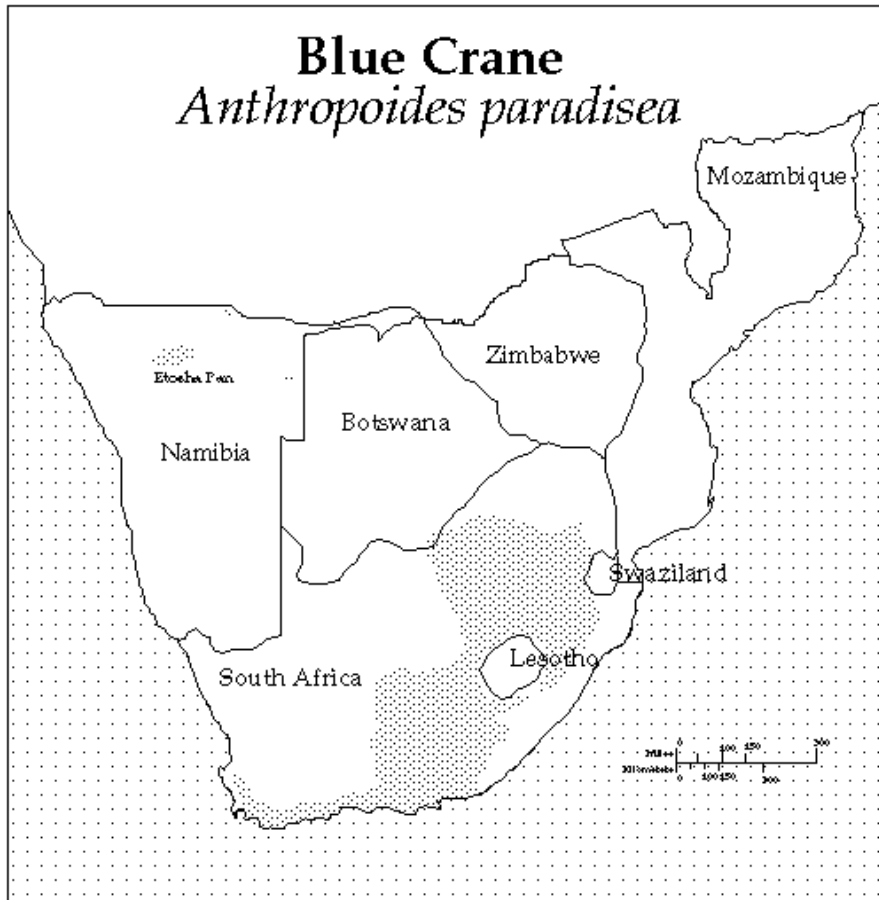


Fig. 13.4. *Distribution of the Blue Crane.*

nesting and feeding, but will roost in wetlands if available. Preferred nesting sites are secluded grasslands in higher elevations, although they also nest in wetlands. In agricultural areas (especially converted farms in the fynbos region), Blue Cranes nest in pastures, fallow fields, and crop fields (Allan 1993; Aucamp 1996). They migrate locally across elevation gradients, spending the breeding season in higher elevation grasslands and moving to lower elevations for the fall and winter (Vernon et al. 1992). Flocking occurs year-round, but intensifies in the winter (Vernon et al. 1992; Allan 1993).

Intentional and unintentional poisoning, afforestation of South Africa's grasslands, and growing human populations constitute the most significant threats to the Blue Crane (Johnson 1992; Tarboton 1992; Allan 1994). As these threats have taken their toll, conservation efforts have accelerated. These measures include: stricter legal protection for the species; local and national surveys of the population; expanded research on the species' biology, ecology, and conservation status; increased attention to habitat management,

especially on private lands; the emergence and active involvement of non-governmental organizations in Blue Crane conservation programs; and the development of new education programs focussing on the status and needs of the species.

Priority conservation measures include: (1) stronger enforcement of existing conservation laws; (2) listing of the species under CITES Appendix I; (3) development of a coordinated plan to halt the poisoning of cranes; (4) identification and protection of critical habitat, especially traditional wintering grounds; (5) adoption of habitat management programs on farms and other private lands; (6) implementation of requirements for environmental impact assessments preceding timber plantation projects; (7) expansion of surveys and monitoring programs

throughout the species' range; (8) extension of research on population dynamics, demographics, seasonal movements, breeding habitat requirements, and the threats posed by poisoning and commercial afforestation; and (9) development of educational programs specifically directed toward private landowners, farm laborers, and students.

Demoiselle Crane

The Demoiselle Crane (Fig. 1.5) is the second most abundant of the world's cranes. Only the Sandhill Crane is more numerous. The total population is estimated at 200,000–240,000, but reliable surveys are available for only limited portions of its range. Six main populations are distinguishable. The three eastern populations (the eastern Asia, Kazakhstan/Central Asia, and Kalmykia) are abundant, each numbering in the tens of thousands. The Black Sea population consists of approximately 500 individuals. A disjunct

non-migratory population in the Atlas Plateau of northern Africa is believed to include no more than 50 birds, and a small breeding population (fewer than 100 cranes) exists in Turkey.

Historical records indicate that the species' range (Fig. 13.8) has contracted substantially (Sudilovskaya 1963; Kovshar 1987; Winter et al. 1995). The species is classified as Lower Risk (Near Threatened) under the revised IUCN Red List criteria. However, the Atlas and Turkey populations are classified Critically Endangered, the Black Sea population is Endangered, and the East Asia population is listed as Vulnerable.

Demoiselle Cranes breed in the Eurasian steppes from the Black Sea to northeastern China. The main wintering grounds are in India, Sudan, and other parts of eastern Africa to Chad. They are primarily grassland birds, but are usually found within a few hundred meters of rivers, shallow lakes, depressions, or other natural wetlands. If water is available, they will inhabit even true deserts. Their winter habitats in

east-central Africa include acacia savannas, grasslands, and riparian areas. In India, they feed in agricultural and stubble fields, and roost in shallow water or on sandbars and mudflats surrounded by water.

The future of the Demoiselle Crane is more secure than for most cranes because of its large total population, broad range, abundant breeding habitat, adaptability, and high rate of breeding success (even in areas inhabited by people). However, the species faces several serious threats. First, much of its breeding habitat in steppe areas is suitable for agricultural conversion. Although the species sometimes successfully adapts to agricultural fields (Winter et al. 1995; Bold et al. 1995), some population reduction is expected as a result of this trend. Its wintering grounds are subject to increased disturbance and agricultural development due to rising human populations. Other threats include sport hunting and persecution in response to occasional crop damage (Ahmad and Shah 1991; Khachar et al. 1991). These threats have brought about the species' decline in the

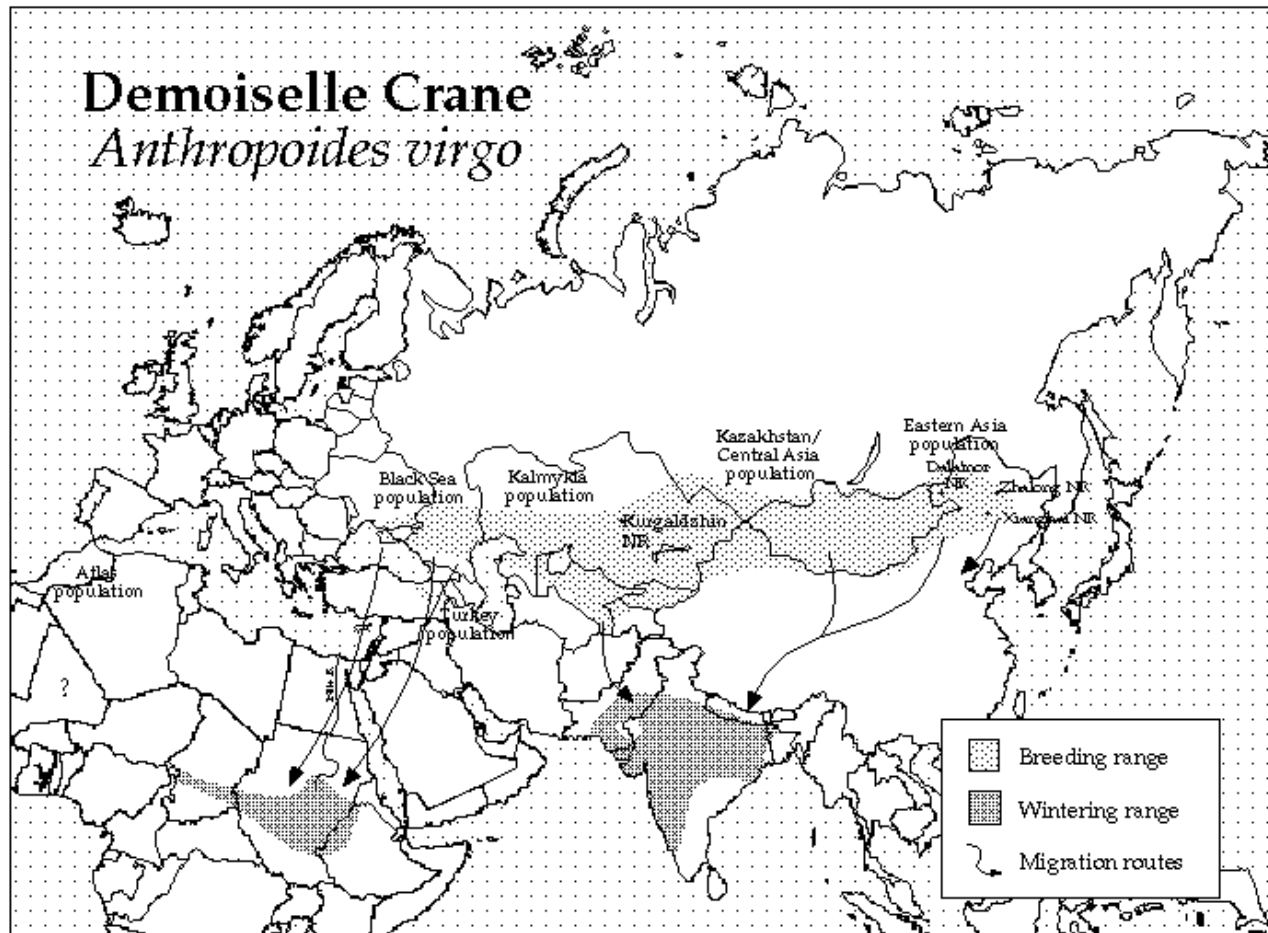


Fig. 13.5. Distribution of the Demoiselle Crane.

western part of its range, and have endangered local populations in other areas.

Conservation measures that have benefitted the Demoiselle Crane include: (1) protection, either through cultural traditions or formal legal restrictions, in many range countries; (2) establishment of numerous protected areas; (3) extensive local surveys and studies of several key migration routes; (4) development of a monitoring program for the threatened Black Sea population; (5) exchange of information on the species in several international forums; and (6) intensive crane education programs in India and Pakistan. No release or reintroduction programs are underway, but releases into areas where it has been severely reduced or extirpated have been considered.

Priority conservation measures for the species include: (1) expanded management programs for the Atlas, Turkey, and Black Sea populations and their habitats; (2) protection of key reserves and establishment of new protected areas in important habitats; (3) development and adoption of agricultural practices that minimize the conflict between cranes and farmers; (4) coordinated international surveys of the species; (5) studies of the migration routes, resting areas, and wintering grounds of the various populations; (6) public education programs in the species' breeding range and along its migration routes; and (7) development of a more specialized education program involving hunters in Pakistan and Afghanistan.

Siberian Crane

The Siberian Crane (Fig. 1.6) is the third rarest species after the Whooping and Red-crowned Cranes. Until 1981, the species was believed to be more endangered than it is today. Then, in 1981, Chinese biologists discovered a wintering flock of 830-850 cranes at Poyang Lake along the middle Yangtze River in China (Zhou et al. 1981). Subsequent field surveys have revised the total population estimate upward to 2,900-3,000 (Gui 1995; Song et al. 1995). These numbers, although encouraging, do not negate the conservation challenges the Siberian Crane faces. Archibald (1992b) noted that "from the tundra to the subtropics, few endangered species involve so many complex problems in so many countries as does the Siberian Crane." The species is classified as Endangered under the revised

IUCN Red List criteria. The central and western populations, because of their extremely small numbers, are Critically Endangered.

Siberian Cranes are divided into three populations. All but a few belong to the eastern population (Fig. 13.6). These birds breed in northeastern Siberia and winter along the middle Yangtze River in China. The central population (Fig. 13.7) breeds in the lower basin of the Kunovat River in western Siberia and winters in the Indian state of Rajasthan, most regularly at Keoladeo National Park. When this population was last observed on its wintering grounds in 1992-93, it included just five birds. Only four birds were observed in the Kunovat breeding grounds in 1995. The western population (Fig. 13.7), which has apparently held at 8-14 birds over the last 8-10 years, winters at a single site along the southern coast of the Caspian Sea in Iran. The exact location of the breeding grounds is still unknown, but recent reports indicate that they lie in the extreme northern portion of European Russia. This population remains extremely vulnerable.

The Siberian Crane is unique among the cranes in its morphology, vocalizations, and behavior (Sauey 1985). It is the most aquatic of the cranes, exclusively using wetlands for nesting, feeding, and roosting. It nests in bogs, marshes, and other wetlands of the lowland tundra, taiga/tundra transition zone, and taiga, preferring wide expanses of shallow fresh water with good visibility. Although its migration and wintering habitats are somewhat more varied, it still feeds and roosts only in shallow wetlands, including artificial water impoundments in India and Iran. Its preferred foods are the roots, tubers, sprouts, and stems of sedges and other aquatic plants, and it seldom forages above water line.

The three populations of Siberian Cranes face an array of threats. The traditional migratory and wintering habitats of the species (especially in China) are under constant pressure from the demands of the growing human population on wetland systems and resources (Harris 1992). Large portions of the eastern population's wintering grounds in China have been lost to drainage, reclamation, and agricultural development. These areas are also threatened by oil exploration and by construction of the Three Gorges Dam on the Yangtze River (Su 1992; Topping 1995). Oil exploration also poses a broad-scale threat to the known breeding grounds of the species. Hunting is believed to be the major cause of the rapid decline of the central population, and is of continuing concern

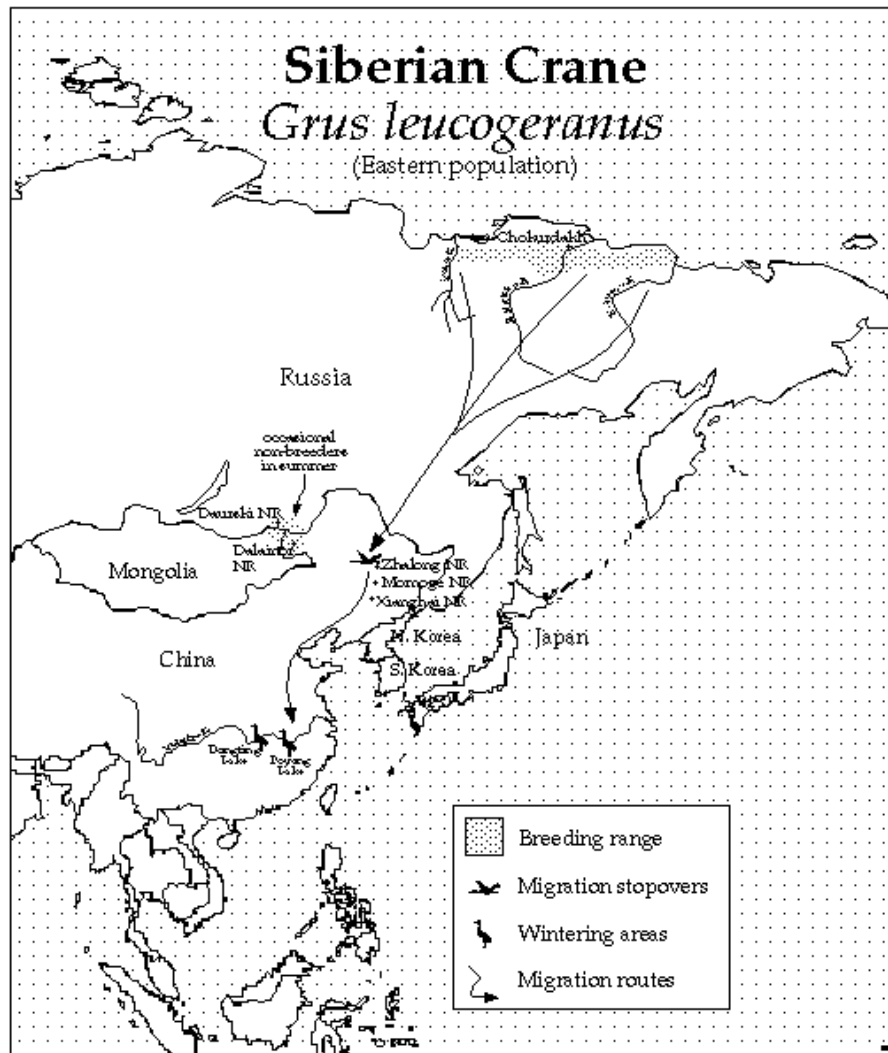


Fig. 13.6. Distribution of the eastern population of the Siberian Crane.

in Pakistan, Afghanistan, and other portions of the species' range (Landfried et al. 1995). The central and western populations are especially vulnerable because of their extremely low numbers.

Concerted conservation efforts on behalf of the Siberian Crane began in the early 1970s. Since then, extensive research has been conducted on the ecology, ethology, breeding and wintering grounds, and migration routes of the species. Annual censuses are available for all three wintering areas and on the breeding grounds of the eastern and central populations. Based on this data, a Population and Habitat Viability Assessment (PHVA) was prepared for the species in 1992 (Mirande et al. *In prep.*). Protected areas have been established at several migration stopover points in Russia, Pakistan, and China, and at the wintering grounds in China and India.

Information about the species has been shared at several international conferences and through expanded communications among biologists. Efforts are now underway to establish an international Siberian Crane Recovery Team and to develop a Recovery Plan. A Memorandum of Understanding Concerning Conservation Measures for the Siberian Crane has been developed. However, it has not yet been signed by all the range countries (UNEP/CMS 1995). An intensive captive propagation program, involving three separate facilities, was initiated in the mid-1970s. Captive-raised birds are now being released in an effort to maintain the central population, and releases are also planned for the western population.

Priority conservation measures for the species include: (1) active participation of all range countries in the Memorandum of Understanding; (2) full development of the Recovery Team and Recovery Plan; (3) creation of protected areas on the breeding grounds and at key staging areas and stopover points; (4) upgrading habitat protection and management efforts at the wintering grounds in Iran and China; (5) continuation of annual winter counts; (6) identification of the western population's breeding grounds in European Russia; (7) identification of migration routes, important staging areas, stopover points, and alternative wintering grounds (including those used by Eurasian Cranes); (8) studies of breeding, migration, wintering, ecology, causes of mortality, and other crucial aspects of Siberian Crane biology; and (9) development of special educational programs involving hunters along the migration route of the central population and communities near the wintering areas in Iran, India, and China. Captive propagation and

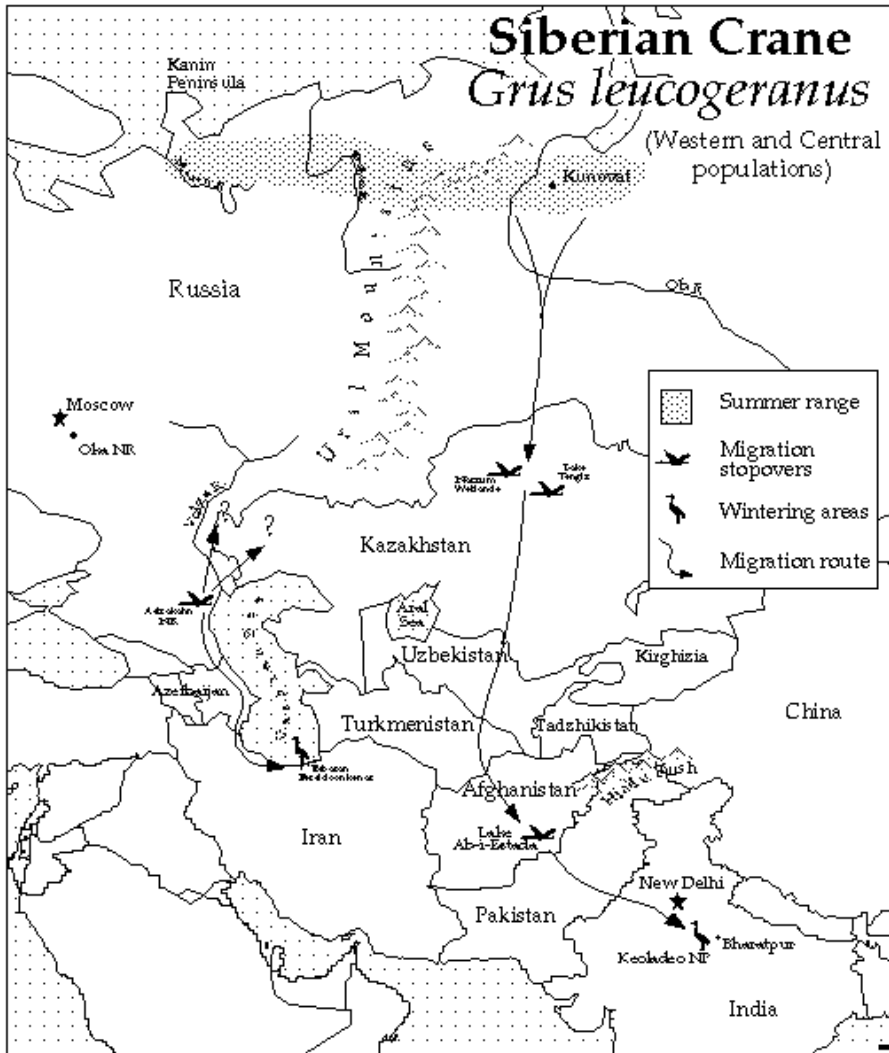


Fig. 13.7. Distribution of the central and western populations of the Siberian Crane.

reintroduction efforts should focus on bolstering the western and central populations, maintaining a genetically diverse captive population, and perfecting rearing and release techniques.

Sandhill Crane

With a total population estimated at more than 500,000, the Sandhill Crane (Fig. 1.7) is the most abundant of the world's cranes. It is widely (though intermittently) distributed throughout North America, with Cuba and northeastern Siberia at the range extremes (Fig. 13.8) (Tacha et al. 1992). Six subspecies have been described. The three migratory subspecies (the Lesser, Greater, and Canadian Sandhill

Cranes) are relatively abundant. They are distributed across a broad breeding range in northern North America and eastern Siberia, with wintering grounds in the southern United States and northern Mexico. The other three subspecies (the Mississippi, Florida, and Cuban Sandhill Cranes) exist as small, non-migratory populations with restricted ranges in the southern United States (southeastern Mississippi, Florida, and southern Georgia) and Cuba. Although some local populations may be declining, the total population is increasing. While the species is classified as Lower Risk under the revised IUCN Red List criteria, the Mississippi and Cuban subspecies are classified as Critically Endangered.

Prior to European settlement of North America, the Sandhill Crane was more widely distributed than at present (Walkinshaw 1949, 1973). While the remote arctic and subarctic breeding grounds of the Lesser and Canadian Sandhill Cranes have been relatively free from human disturbance, the wintering grounds of these subspecies have been extensively altered. Hunting, agricultural expansion, drainage of wetlands, and other habitat changes in the 18th and 19th centuries led to the extirpation of the Greater Sandhill Crane from many parts of its breeding range in the United States and Canada (Walkinshaw 1949). In recent decades, conservation efforts have allowed some of these populations to recover. The numbers and distribution of the two non-migratory races of the Sandhill Crane in the southern United States have diminished due to hunting, loss of wetlands, and other changes in its habitat. The Cuban Sandhill Crane was probably more widely distributed in the Cuban archipelago than at present.

Sandhill Cranes are primarily birds of open fresh-water wetlands and shallow marshes, but the different subspecies use a broad range of habitat types from bogs, sedge meadows, and fens to open grasslands, pine savannas, and cultivated lands (Tacha et al. 1992). During the breeding season, the three migratory subspecies are found in a wide variety of northern wetlands. Habitats along migration routes tend to be large, open palustrine and riparian wetlands near agricultural areas, while wintering habitats include riparian wetlands, wet meadows, playas, and pastures (Krapu et al. 1984; Iverson et al. 1987). The non-migratory subspecies use seasonally variable wetlands, grasslands, and palm and pine savannas (Smith and Valentine 1987; Nesbitt and Williams 1990; Galvez

and Perera 1995). Sandhill Cranes are omnivorous, feeding on a wide variety of plant materials (including waste grains) and small vertebrates and invertebrates, both on land and in shallow wetlands.

The leading threat to Sandhill Cranes is the loss and degradation of wetland habitats, especially ecological and hydrological changes in important staging areas. Of special concern are the spring staging areas along the central Platte River. The areas favored by the cranes have diminished due to anthropogenic effects on the river's flow. Current plans, if carried out, would result in more dams and excessive water withdrawals (Currier et al. 1985; Faanes and Bowman 1992). Continuing loss of roosting habitat has concentrated the migrating cranes, increasing the risks associated

with disease, disturbance, and other threats. Habitat loss continues on breeding grounds of the Greater Sandhill Crane and on the year-round habitats of the non-migratory subspecies. Overhunting poses a potential threat to certain segments of the mid-continental Sandhill Crane populations. Lead and mycotoxin poisoning, abnormal predation pressures, and collisions with fences, vehicles, and utility lines are of local concern.

Since the decline of the Sandhill Crane in the first half of the 1900s, extensive conservation measures have been undertaken on its behalf. These include: (1) legal protection under the Migratory Bird Treaty of 1916; (2) establishment of protected areas in key breeding, migration, and wintering habitats; (3) stronger national wetland protection policies and programs; (4) annual surveys and counts of many populations; (5) wide-ranging research on many aspects of the species' biology and ecology; (6) management guidelines and plans for mid-continental and Rocky Mountain populations; (7) development of a recovery plan, PHVA, and captive propagation and release program for the

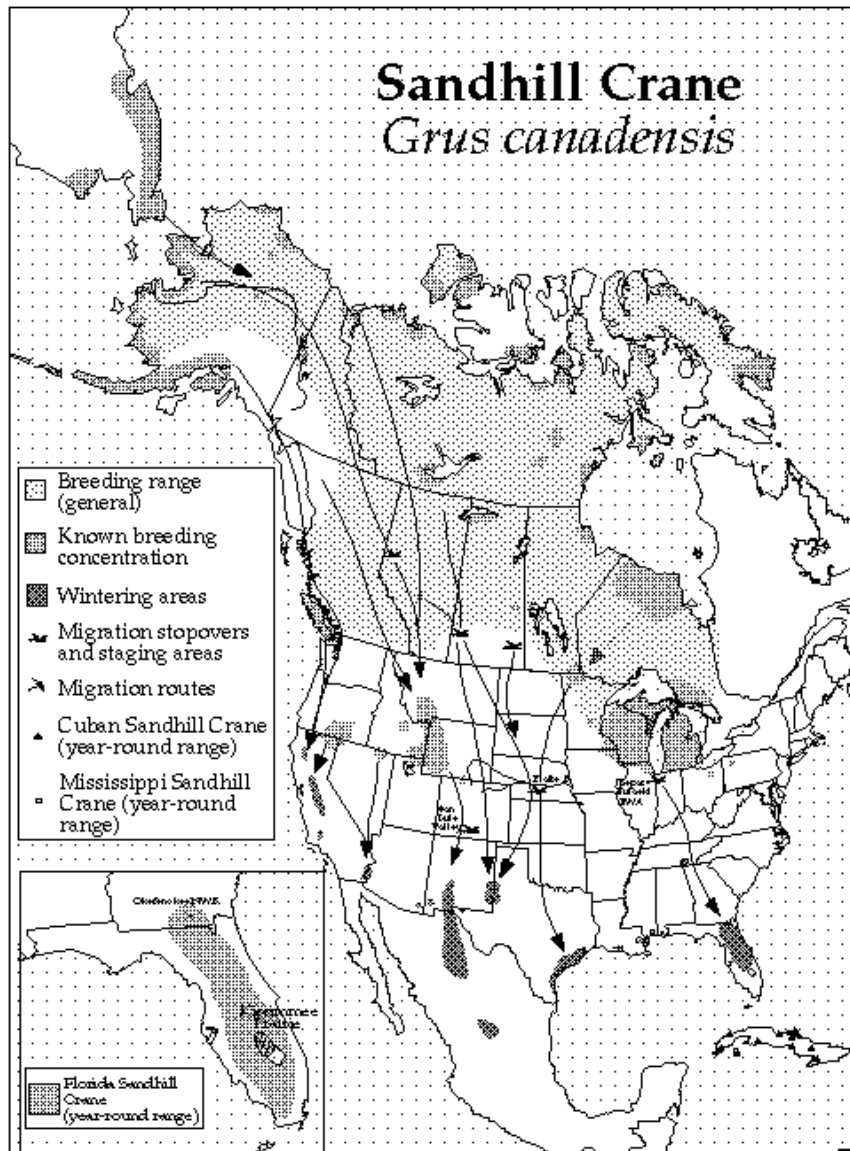


Fig. 13.8. Distribution of the Sandhill Crane.

Mississippi Sandhill Crane; (8) initiation of a research and management program for the Cuban Sandhill Crane; and (9) a wide variety of public education programs.

Priority conservation measures for the Sandhill Crane include: (1) protection, restoration, and management of critical breeding, migration, and wintering habitat for the migratory subspecies (especially along the Platte River) and of the year-round use areas for the non-migratory subspecies; (2) implementation of conservation programs and incentives that involve private landowners; (3) research to improve understanding of the size, status, dynamics, distribution, and movements of populations; (4) continued implementation and updating of the recovery plan for the Mississippi Sandhill Crane; (5) development of a comprehensive Cuban Sandhill Crane conservation program; (6) greater attention to problems associated with crop depredation; (7) greater attention to the long-term effects of hunting; and (8) clarification of subspecies phylogeny.

White-naped Crane

The total population of White-naped Cranes (Fig. 1.8) is estimated at 4,900–5,300. The species breeds in northeastern Mongolia, northeastern China, and adjacent areas of southeastern Russia (Fig. 13.9). Birds in the western portion of the breeding range (about 3,000 individuals) migrate south through China, resting along the Yellow River delta, and wintering in the middle Yangtze River valley (Harris et al. 1995). Approximately 2,000 birds in the eastern portion of the breeding range migrate south through the Korean peninsula (Chonget al. 1994; Ohsako 1994). Several hundred remain on wintering grounds in the Demilitarized Zone between North and South Korea; the remainder continue onto the Japanese island of Kyushu. In the past, the White-naped Crane was more numerous and more extensively distributed than at present (Flint 1978; Won 1981). The population reached its nadir in the years following World War I and the Korean War. Since then, it has increased in many parts of its range, although it may again be declining in parts of Russia and China. The species is classified as Vulnerable under the revised IUCN Red List criteria.

Typical White-naped Crane breeding habitat includes shallow wetlands and wet meadows in broad river valleys, along lake edges, and in lowland steppes

or mixed forest steppes (Su et al. 1991; Fujita et al. 1994). White-naped Cranes feed in their breeding habitat and in adjacent grasslands or farmlands. During migration and on their wintering grounds, they use rice paddies, mudflats, other wetlands, and agricultural fields, where they feed on waste grains, seeds, and tubers (Chen et al. 1987; Halvorson and Kaliher 1995).

The loss of wetlands to agricultural expansion, especially in the breeding grounds of the Amur River basin and other parts of northern China, is the most significant threat to the species. Its preferred habitats are especially prone to drainage and conversion to agriculture (Harris 1994a). The Korean Demilitarized Zone, which has served as a critical refuge for White-naped and Red-crowned Cranes, is highly vulnerable to armed conflict or to development should political tensions between North and South Korea be resolved (Halvorson and Kaliher 1995). Other threats include overexploitation of wetland resources, ineffective management of key protected areas, indiscriminate pesticide use, and the proposed dams on the Amur River and on the Yangtze River at Three Gorges. Wintering Hooded and White-naped Cranes at Izumi in Japan are highly concentrated and susceptible to disease outbreaks.

Current conservation measures for the species include: legal protection in all range countries; international cooperation to protect the species and to manage key reserves in the China-Russia-Mongolia border region; establishment of protected areas in important breeding and wintering habitats; regular surveys of the population at migration stopover points and on the main wintering grounds; expanded research on the species throughout its range; and the involvement of non-governmental organizations in research, habitat protection, and captive propagation programs. Limited releases of captive-reared birds have been carried out at the Zhalong Nature Reserve in China and the Khinganski Nature Reserve in Russia.

Priority conservation measures include: (1) expanded international cooperation in managing nature reserves and in research on migration patterns and timing; (2) expansion of existing reserves and establishment of new protected areas (especially in Mongolia, northeastern China, and the Korean Peninsula); (3) dispersion of the wintering crane populations at Izumi; (4) development of integrated land use and conservation programs in key watersheds; (5) assessment of the environmental impacts of large-scale dam and development projects; (6) continuing sur-



Fig. 13.9. Distribution of the White-naped Crane.

veys of the population; (7) more complete identification of the species' breeding grounds, especially in northeastern China; (8) professional training opportunities for reserve managers and conservation officials; (9) improved agricultural information services for farmers; and (10) community-based conservation education programs involving cranes and wetlands.

Sarus Crane

At 1.8 m, the Sarus Crane (Fig. 1.9) is the world's tallest flying bird. It is also the only resident breeding crane in India and southeast Asia (Fig. 13.10). The total population for the three subspecies is between 13,500 and 15,500. The Indian Sarus Crane (*G. a. antigone*) is still common in northern India, but has

been extirpated from large portions of its historical range and continues to decline in areas where it still exists (Gole 1989, 1991). The Eastern Sarus Crane (*G. a. sharpi*) has been decimated throughout its historical range in southeast Asia. One known population, estimated at between 500 and 1,500, breeds in Cambodia and Laos (and possibly surrounding areas) and winters in Vietnam (Duc 1991; Barzen 1994). The Australian Sarus Crane (*G. a. gillii*) is limited to northeastern Australia, and probably numbers less than 5,000 (A. Haffenden, Australian Nature, Atlanta, Georgia, personal communication). Sarus Cranes, possibly of a distinct subspecies, formerly occurred in the Philippines. This population is now presumed extinct. The species is classified as Endangered under the revised IUCN Red List criteria. The Indian and Eastern subspecies are also classified as Endangered. We know too little about the Australian subspecies to classify it at this time.

The three subspecies use widely varying habitats. The Indian race is highly tolerant of human activity. These birds use even very small wetlands if they are not persecuted or heavily disturbed (Gole 1989, 1991). Breeding pairs and families with pre-fledged chicks are typically dispersed among scattered natural and artificial wetlands. Adult pairs will use drier habitats such as cultivated and fallow fields. Eastern Sarus Cranes are less tolerant of people and are almost completely dependent on natural wetlands in both wet and dry seasons. Australian Sarus Cranes nest in open wetlands during Australia's wet season and feed in upland agricultural fields and grasslands at other times of the year (Marchant and Higgins 1993).

Loss and degradation of wetlands (due to agricultural expansion, industrial development, river basin development, pollution, warfare, heavy use of pesticides, and other factors) are the most important

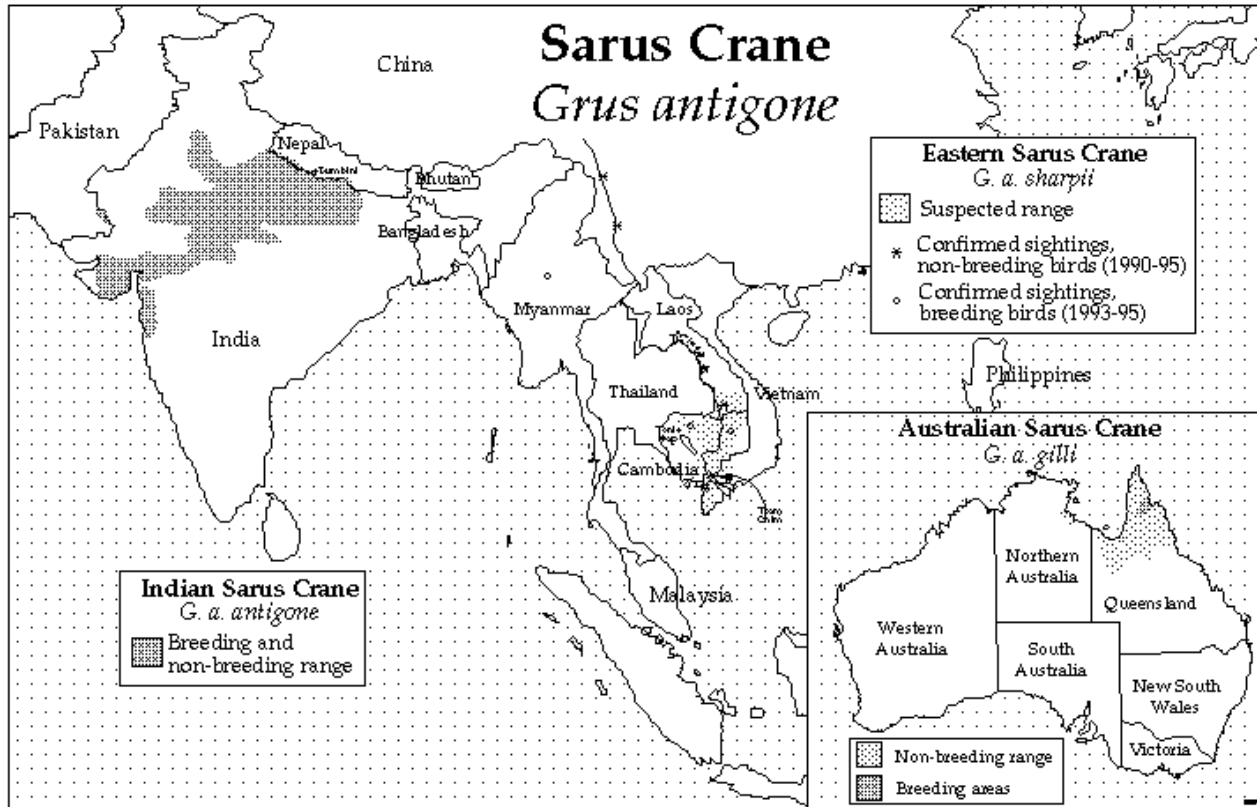


Fig. 13.10. Distribution of the Sarus Crane.

threats to the species, especially in India and southeast Asia. In many areas, high human population pressures compound these threats by increasing disturbance (Gole 1991; Suwal 1995). Human population growth and planned development projects on the Mekong River are acute threats to the Eastern Sarus Crane (Lohmann 1990). Hunting, egg stealing, and the capturing of chicks are also significant problems in some areas, and especially affect the Eastern Sarus Crane. Trading in adults and chicks has been reported in India, Cambodia, and Thailand.

Local traditions and religious beliefs have protected the Sarus Crane in many parts of its range, especially northern India, Nepal's western Terai, and Vietnam (Gole 1993). The species has been the focus of increased conservation activity in recent years, including: (1) international agreements and collaborative conservation projects in southeast Asia; (2) field studies of the species in India and Nepal; (3) intensive surveys of the Eastern Sarus Crane during the dry season in Vietnam and during the breeding season in Cambodia; (4) establishment of the Tram Chim National Reserve in Vietnam and efforts to restore the reserve's wetlands; (5) convening (in 1990)

an International Sarus Crane and Wetland Conservation Workshop; (6) development of a preliminary PHVA for the Eastern Sarus Crane; and (7) establishment of education programs in Nepal and Vietnam. Sarus Cranes are not currently being reintroduced, but plans for reintroduction are underway in Thailand and other portions of the species' historical range.

Priority conservation measures for the species include: (1) identification and protection of breeding areas in India, Cambodia, Myanmar, and Laos, and of non-breeding habitat in Vietnam, Laos, and Cambodia; (2) full implementation of the management plan for Vietnam's Tram Chim National Reserve; (3) protection, maintenance, and restoration of village ponds and other small wetlands in India; (4) improved pesticide management and regulation in agricultural areas used by cranes, especially in India and Nepal; (5) watershed-level conservation planning in the Mekong River basin; (6) expanded efforts to survey and monitor Eastern Sarus Cranes; (7) further research on distribution, ecology, movement, and habitat needs throughout the species range; (8) expanded surveys and basic ecological studies of the

Australian Sarus Crane; (9) development and implementation of national-level wetland conservation plans in range countries; (10) preparation of full PHVAs for both the Indian and Eastern Sarus Crane; and (11) assessments of existing habitat and the potential for natural recolonization in areas where the species is now rare or extinct.

Brolga

The Brolga (Fig. 13.10) occurs throughout northern and eastern Australia and in limited areas of New Guinea (Fig. 13.11) (White 1987; Marchant and Higgins 1993). The Brolgas in northern and southern Australia can be regarded as discrete populations, but are no longer recognized as distinct subspecies. Because no systematic, range-wide survey of the species has been undertaken, no reliable population estimate is available and trends are poorly understood. The total population may range from 20,000 to 100,000 and is probably stable overall. This species still occupies much of its historical range. In recent decades, the Brolga has declined in southeastern

Australia, while apparently expanding (due to increasing use of croplands) in the Northern Territory, on the Kimberley Plateau, and elsewhere in western Australia (Blakers et al. 1984; White 1987). Little is known about the status of, or trends in, the New Guinea populations. The species is classified as Lower Risk (Least Concern) under the revised IUCN Red List criteria.

Brolgas are non-migratory, but do move in response to seasonal rains. Ecologically, they are perhaps the most opportunistic of the cranes, having evolved to cope with Australia's extreme climatic variations. Northern populations are concentrated during the dry season in coastal freshwater wetlands where they subsist on the tubers of the bulkuru sedge (*Eleocharis dulcis*). In the wet season, they disperse to breeding territories in freshwater and brackish marshes, wet meadows, and other seasonal wetlands (Lavery and Blackman 1969; Blackman 1977). Although the wet and dry seasons in southern Australia are less marked, southern Brolga populations also move between wet season breeding territories and traditional dry season flocking areas. They inhabit a similarly wide range of available wetland types, but generally use salt marshes far less than the northern Brolgas.

The most significant threat to the Brolga across its range is the loss and degradation of wetland habitat. In northern Australia (especially along the eastern coast), wetlands used by Brolgas have been extensively degraded by livestock grazing, disruption of hydrological processes, and changes in vegetation (A. Haffenden, Australian Nature, Atlanta, Georgia, personal communication). In the south, loss of wetlands to drainage and reclamation for agriculture is probably the main factor behind the dramatic decline in Brolgas there (Arnold et al. 1984). Other threats include the subdivision (and subsequent fencing) of large private land holdings, pre-

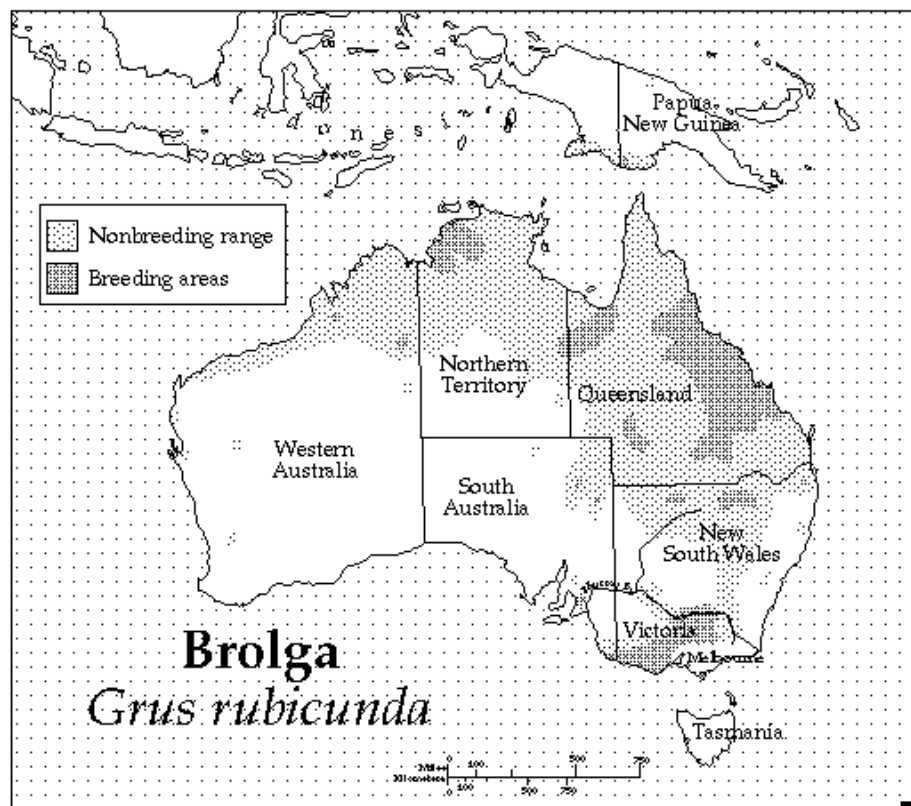


Fig. 13.11. Distribution of the Brolga.

dation by the introduced red fox (*Vulpes fulva*), incidental poisoning, and collisions with utility lines.

Most research and conservation activity involving the Brolga has taken place in the southeast, where the species is no longer as common as in the north. Conservation measures undertaken for the species include: (1) legal protection throughout Australia; (2) local surveys in South Australia, Victoria, and New South Wales; (3) preparation of an Action Statement for Brolgas under the Victorian Flora and Fauna Guarantee Act; (4) programs to protect and restore privately owned wetlands in Victoria; and (5) establishment of a private conservation organization, Friends of the Brolga. A captive propagation program was initiated in Victoria in 1964; surplus birds from this program are to be released in 1995 and 1996.

Priority conservation needs for the Brolga include: (1) adaption of strong watershed-level wetland conservation programs, (2) assessment of the status and conservation needs of the species in New Guinea; (3) enactment of stronger national wetland protection laws and policies; (4) development of incentive and extension programs to encourage and reward private landowners who conserve Brolga habitat; (5) development of a systematic censusing and monitoring program for the species, and inclusion of the species in routine aerial waterfowl counts; (6) expanded research on flocking sites, breeding biology, size and trends of each population, and identification of isolated populations; and (7) expanded education and extension programs.

Eurasian Crane

The Eurasian Crane (Fig. 1.11) is the third most abundant species after the Sandhill and Demoiselle Cranes. The total population, estimated at 220,000–250,000, is probably increasing, although some populations are declining. However, no coordinated survey has been conducted throughout the species' range. The species is not globally threatened, but it is legally protected in many countries. The species is classified Lower Risk (Least Concern) under the revised IUCN Red List criteria. Breeding populations in European Russia and central Siberia are classified Vulnerable, while small populations in Turkey and the Tibetan Plateau are too poorly known to classify at present.

The species' breeding range extends from northern and western Europe across Eurasia to northern

Mongolia, northern China, and eastern Siberia, with isolated breeding populations in eastern Turkey and Tibet (Fig. 13.12). The winter range includes portions of France and the Iberian Peninsula, north and east Africa, the Middle East, India, and southern and eastern China. The species continues to occupy most of its historical range. However, during the last 200–400 years it has been extirpated as a breeding species in much of southern and western Europe, the Balkan Peninsula, and southern Ukraine (Prange 1989, 1995).

The Eurasian Crane nests primarily in bogs, sedge meadows, and other wetlands within Eurasia's boreal and temperate forest zones (Walkinshaw 1973; Johnsgard 1983). Under natural conditions, pairs prefer large, isolated nesting territories. However, in intensively cultivated areas they have adapted to smaller and less wild wetlands (Mewes 1994). During migration, they forage in agricultural fields, pastures, and meadows, and roost in shallow lakes, bogs, rivers, along the edges of reservoirs, and in other wetlands. The widely scattered wintering grounds include a wide spectrum of upland and wetland habitats, from open oak woodlands in the Iberian Peninsula to shallow lakes, agricultural fields, and delta wetlands in China (Alonso and Alonso 1990; Xu et al. 1991). Eurasian Cranes are omnivorous, foraging in wetlands, on dry land, and in agricultural fields for a wide variety of plant and animal foods.

Habitat loss and degradation are the principal threats to the species. Wetlands have been lost to drainage, dams, and other forms of development throughout the species' range, particularly in Europe, European Russia, and central Asia (Prikloński and Markin 1982; Harris 1992; Newton 1996). Although Eurasian Cranes have adapted to human settlement in many areas, continuing changes in land use and agricultural production methods (such as expanded irrigation and conversion of traditional pastures) have had negative impacts. Human disturbance and collision with utility lines are problems in Europe and other heavily developed areas. Hunting is a significant concern for the populations that migrate through Afghanistan and Pakistan (Landfried et al. 1995).

Conservation measures have been undertaken most intensively in the western portions of the species' range. In western and central Europe, the species has benefitted from legal protection, systematic research and monitoring programs, creation and restoration of wetlands, and protection of important staging areas, roosting sites, and wintering grounds. Information

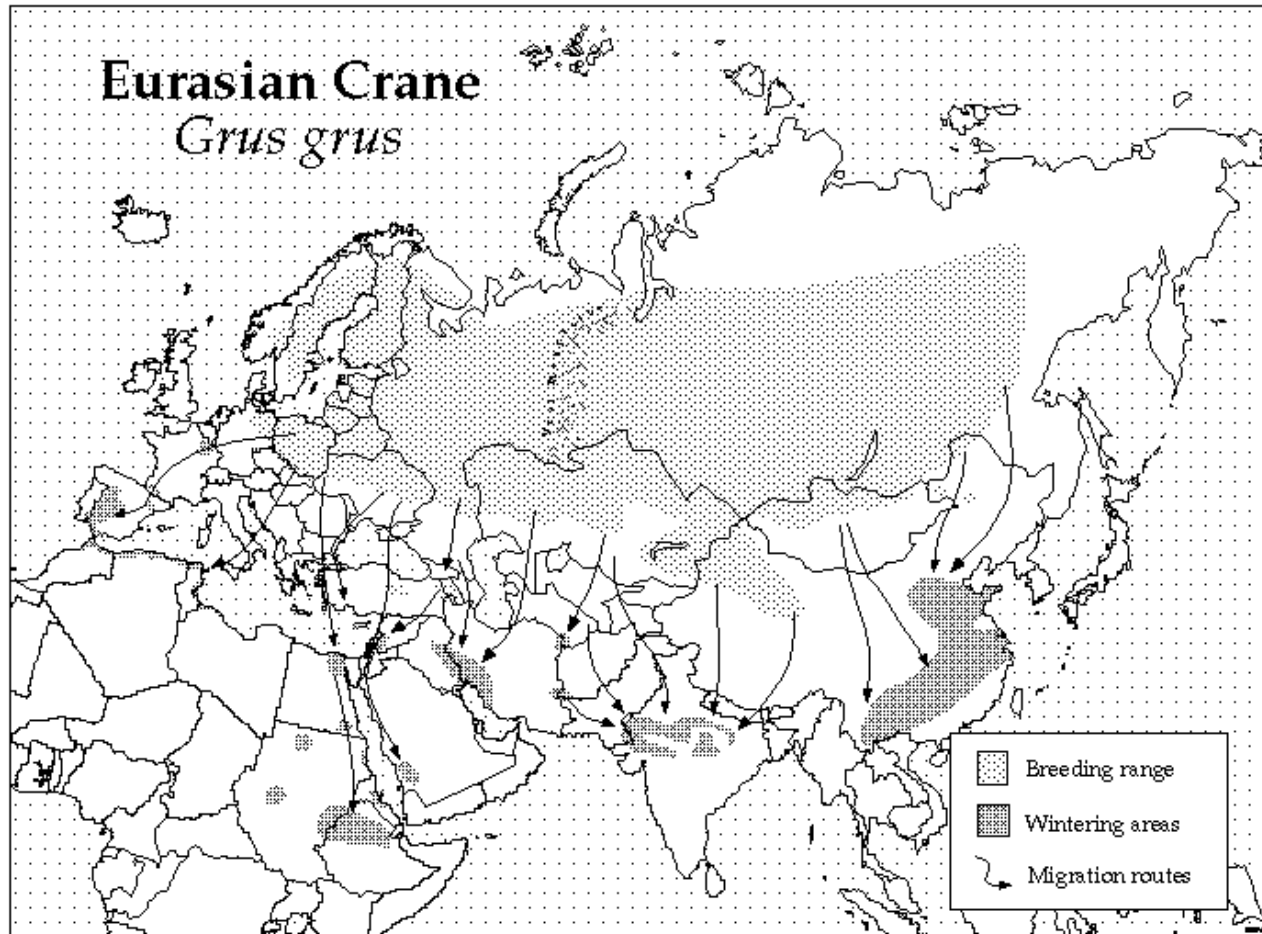


Fig. 13.12. *Distribution of the Eurasian Crane.*

about migration patterns is available due to color banding programs and regular observations along the migration routes (Prange 1989, 1995). International cooperation has played an important role in promoting these measures. In the last decade, such cooperation has expanded into Eastern Europe, where the species has been under greater threat due to recent economic changes. Conservation efforts have been less focused in eastern Russia, Africa, the Middle East, and Asia. In these areas, however, the Eurasian Crane often mixes with other cranes and thereby has benefitted from conservation actions undertaken on their behalf.

Priority conservation measures for the species include: (1) adoption of the Ramsar Convention in all range countries; (2) stronger legal protection for cranes and crane habitats; (3) expanded international research, monitoring, and conservation programs; (4) establishment of protected areas at key breeding, staging, and wintering areas; (5) broad-scale wetland protection and restoration programs (especially in Europe); (6) expanded efforts to survey and census

populations; (7) research on the number, status, distribution, migration routes, and breeding and wintering areas of the main populations; (8) field studies of the isolated populations in the Tibetan Plateau and Turkey; (9) establishment of a central database to maintain information on the species; (10) coordinated efforts to address crop depredation problems; (11) training programs for volunteers working in protected areas established for cranes; and (12) expanded education programs for students and the general public.

Hooded Crane

The total population of Hooded Cranes is estimated at 9,400-9,600. The breeding grounds of the species (Fig. 13.13) are in southeastern Russia and northern China, while non-breeding flocks occur in the Russia-Mongolia-China border region (Neufeldt 1981; Fujimaki et al. 1989; Li 1995; Bold et al. 1995). The

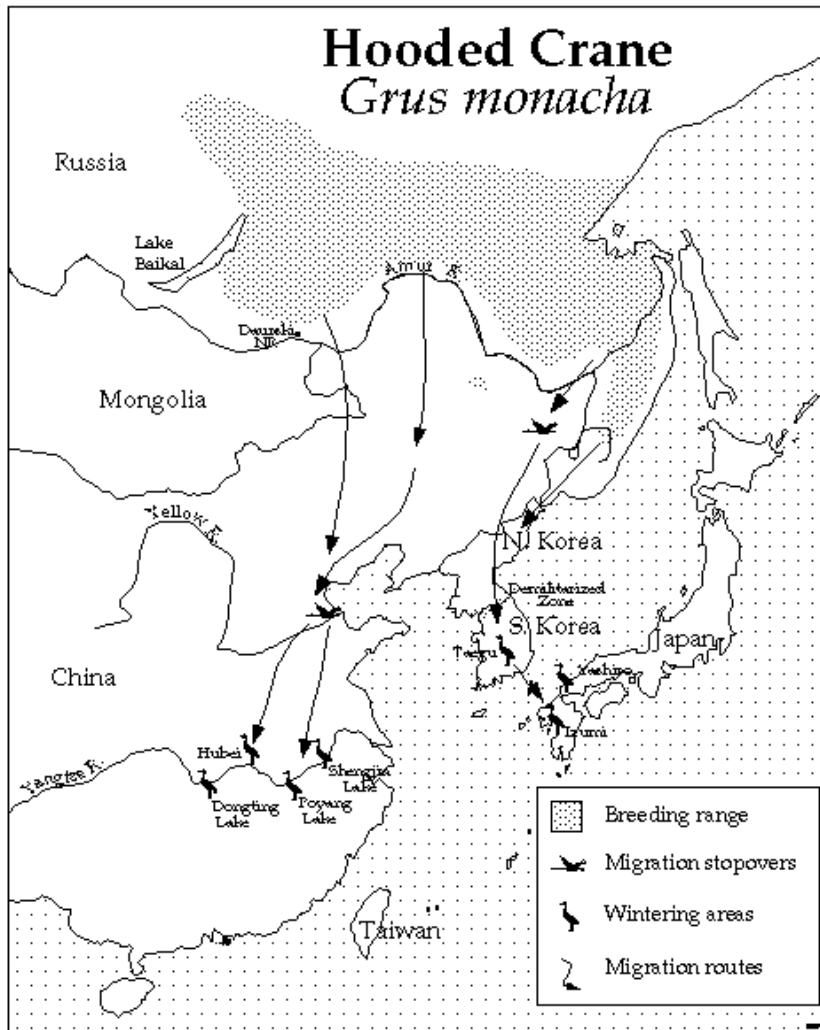


Fig. 13.13. Distribution of the Hooded Crane.

species is divided into several wintering subpopulations. More than 80% of the world's Hooded Cranes (about 8,000 birds) spend the winter at Izumi on the Japanese island of Kyushu, where they are sustained by artificial feeding (Ohsako 1994). Small subpopulations are found at Yashiro in southern Japan, near Taegu in South Korea, and at several sites along the middle Yangtze River in China. Although little is known about historical changes in the distribution of the species, its numbers are known to have fluctuated dramatically since the 1920s (Ohsako 1987). At present, the population is probably as large as at any point this century. The species is classified as Vulnerable under the revised IUCN Red List criteria.

Hooded Cranes (Fig. 1.12) nest in isolated, widely scattered bogs in the taiga and in other forested wetlands, preferring mossy areas with widely scattered larch trees (*Larix* sp.), and avoiding areas that are

either too open or too densely forested (Pukinski 1977). Non-breeding cranes are found in shallow open wetlands, natural grasslands, and agricultural fields in southern Siberia, northeastern Mongolia, and northern China. Wintering Hooded Cranes use a wide variety of habitats. In China, they tend to roost along the shores of rivers and shallow lakes, and to forage in the muddy edges of lakes and in nearby grasslands, grassy marshes, rice paddies, and agricultural fields (Chen and Wang 1991). In Korea and Japan they feed almost exclusively at feeding stations and in agricultural fields (Cho and Won 1990; Ohsako 1987).

Although the Hooded Crane is a threatened species, it is more secure than other threatened cranes of East Asia. This is due mainly to the relative absence of intensive human economic activity in their breeding grounds. Moreover, the species (unlike the other East Asian cranes) winters primarily in Japan rather than China and the Korean Peninsula, where threats are somewhat greater. However, the species does face several critical threats, including: drainage of wetlands and intensified logging in Russia's taiga

forests; reclamation of wintering grounds in China for agriculture and alterations in the hydrology of these areas; the planned Three Gorges dam on the Yangtze River; rapid development of the key wintering grounds in Korea, especially through the construction of greenhouses; and high risk of disease outbreak in the concentrated flocks at the winter feeding stations in Japan.

Conservation measures that have been undertaken include: (1) legal protection throughout the species' range; (2) international agreements to protect the species and key habitat throughout its range; (3) recently expanded research on breeding habitats, winter ecology, and migration routes; (4) annual surveys of populations on the wintering grounds; (5) establishment of protected areas, especially in the winter range; and (6) intensive management (including the artificial feeding programs) at the main wintering area in Japan.

The Hooded Crane has many of the same priority conservation needs as the White-naped, Red-crowned, and Siberian Cranes, including stronger enforcement of existing laws, adoption of an umbrella agreement on the migratory cranes of East Asia, adoption of the Ramsar Convention in all range countries, expanded international conservation programs, continued research on migration routes, and protection of key habitats in China and the Korean Peninsula.

Additional priorities specific to the species include: protection of potential alternative feeding and roosting sites for the wintering populations in southern Japan and Korea; studies of the West Taegu population in Korea and application of this information in creating an adequate protected area for the flock; agreements to bring greenhouse development under control in and near the Hooded Crane Protection Area in Korea; continued winter surveys of all Hooded Crane populations; and development of a program to monitor the status of the breeding grounds in Russia.

Black-necked Crane

contributed by Mary Anne Bishop

The world's Black-necked Crane population is estimated at 5,600–6,000. The species' breeding range (Fig. 13.14) includes much of the Qinghai-Tibetan Plateau in China, with a small breeding population occurring nearby in Ladakh, India (Lu et al. 1980). Six main wintering locations have been identified. These include lower elevations of the Qinghai-Tibet and Yunnan-Guizhou Plateaus in China, with some birds also occurring in Bhutan and Arunachal Pradesh, India (Bishop 1993). Published records and local reports indicate that the species has declined in many breeding and wintering areas over the last seventy years, although the population seems to have stabilized since the 1970s. The species is classified as Vulnerable under the revised IUCN Red List Criteria.

During the breeding season Black-necked Cranes (Fig. 1.13) use high altitude wetlands, nesting in grassy marshlands, sedge meadows, and marshes along the shores of lakes and streams, and foraging in shallow marshes, streams, and pastures (Li 1987). Their diet includes plant roots, tubers, snails, shrimp, small fish, and other small vertebrates and invertebrates. The

cranes winter in lower elevation, agricultural valleys, where they feed mainly on waste grains and other residue in fields and pastures. In both breeding and wintering areas, Black-necked Cranes are quite tolerant of human activity, and regularly feed near human settlements and domestic livestock.

Degradation and loss of habitat are the main threats facing the Black-necked Crane. These problems are most serious in the wintering areas, where wetlands have been extensively affected by irrigation projects, dam construction, drainage and conversion to agriculture, river channelization, heavy grazing pressure, sedimentation, and industrial pollution (Li and Li 1991; Wei et al. 1994; Bishop et al. *In prep.*). In Tibet, widespread changes in traditional agricultural practices have reduced the availability of waste barley and spring wheat, the main winter foods (Bishop 1991). Hunting has become an important threat in several wintering areas with the introduction of firearms and increased accessibility of formerly remote areas. Other factors, including egg collecting and predation by feral dogs, are significant threats in some locales.

Conservation measures for the species have greatly expanded since the late 1970s. These measures include: (1) implementation of an integrated program of conservation and development at Cao Hai Lake, a key wintering area in Guizhou Province, China; (2) establishment of additional protected areas in China and Bhutan; (3) regular population surveys in the main wintering areas; (4) expanded field studies of the species' distribution, habitat use, breeding biology, wintering ecology, and conservation status; (5) support for conservation programs from national and international non-governmental organizations; and (6) training programs for local conservation officials and reserve personnel. Local religious beliefs have also played a critical role in safeguarding the Black-necked Crane across much of its range.

Priority conservation measures for the species include: (1) stronger efforts to control poaching; (2) improved management of existing protected areas (especially Cao Hai Nature Reserve); (3) establishment of protected areas in Yunnan and India; (4) protection of wetlands (especially in wintering areas) against further deterioration and development; (5) establishment of agricultural management areas in key wintering and breeding areas; (6) regular, coordinated counts of the wintering populations; (7) banding and satellite radio studies of the main wintering populations; (8) studies of roosting habitats in Tibet,

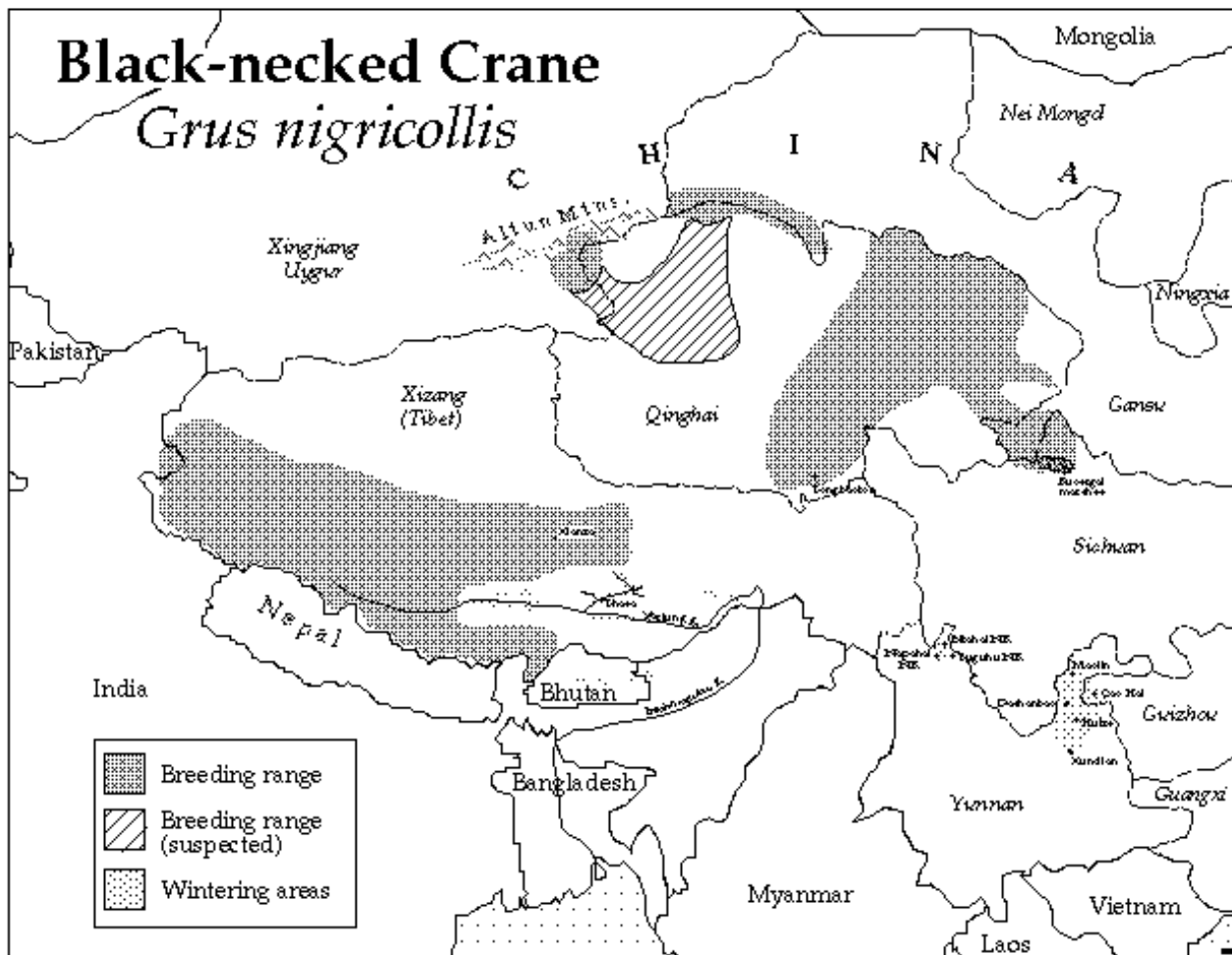


Fig. 13.14. *Distribution of the Black-necked Crane.*

Yunnan, and Bhutan; (9) further development of education programs; and (10) expanded training opportunities for nature reserve personnel.

Red-crowned Crane

contributed by Scott R. Swengel

The Red-crowned Crane (Fig. 13.14) is the second rarest species of crane, with a total population in the wild of 1,700-2,000 birds. The species breeds in large wetlands in temperate East Asia, and winters along rivers and in coastal and freshwater marshes in Japan, China, and the Korean Peninsula (Fig. 13.15). There are two main breeding populations, a migratory population on the East Asia mainland (northeastern China and Russia) and a resident population on the island of

Hokkaido in northern Japan (Masatomi 1981; Su 1993). In the winter, the mainland population divides into two or three wintering subpopulations. The total population has fluctuated over the last century, probably reaching its lowest point in the years following World War II (Masatomi 1981). Although the species has recovered in some areas, much habitat has been lost to agricultural development and other human economic activities. The species is classified as Endangered under the revised IUCN Red List criteria.

Red-crowned Cranes prefer to nest and feed in marshes with relatively deep water, building their nests in areas with standing dead vegetation (Smirenski 1988). They prefer wetter foraging sites, but will also forage on dikes and in croplands. On their wintering grounds, they feed on waste (or human-provided) grain and on aquatic plants and animals in coastal marshes and open watercourses (Andronov et al. 1988; Masatomi 1993).

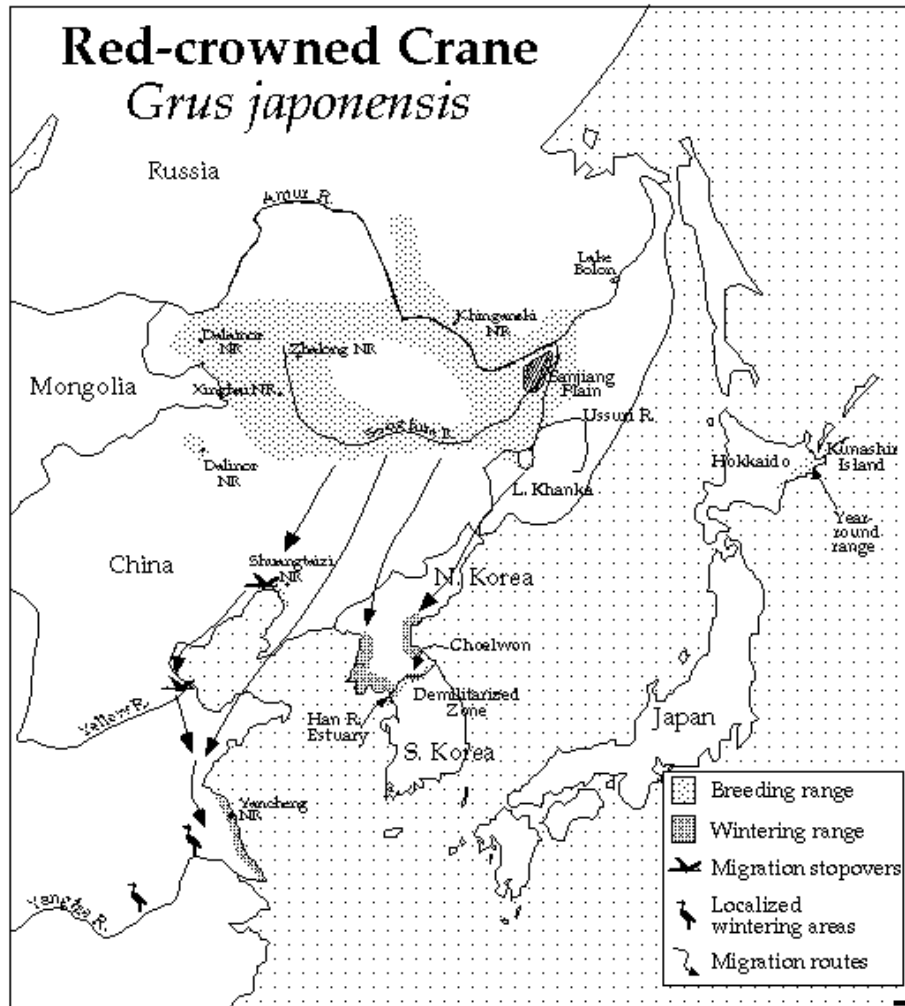


Fig. 13.15. *Distribution of the Red-crowned Crane.*

Habitat loss is the principal threat to the species. Continued agricultural and industrial development affects breeding areas in Hokkaido, the Sanjiang Plain in northeastern China, and the Amur River basin in Russia (Masatomi et al. 1990; Harris 1994a; Smirenski et al. 1995). Water control and diversion projects (including proposed dams on the Amur River and on the Yangtze River) and the potential for conflict or development in the Korean Demilitarized Zone pose large-scale threats to breeding, migration, and wintering habitat. Other anthropogenic threats include disturbance, intentional setting of fires, and overharvest of wetland resources in key breeding areas (Harris 1994a).

Current conservation measures include: (1) international agreements and cooperative research (especially involving migration); (2) establishment of reserves to protect habitat and minimize disturbance; (3) devel-

opment of winter feeding stations and the marking of nearby powerlines in Japan; (4) regular surveys on breeding and wintering grounds; (5) preparation of a PHVA for the species; (6) cooperative conservation and education programs focused on the species; and (7) several limited reintroduction efforts.

Priority conservation needs include: (1) adoption of an umbrella international agreement on the cranes of East Asia; (2) development of a comprehensive recovery plan for the species; (3) continued international cooperation in research on migration routes and patterns; (4) protection of key habitats on the Korean Peninsula; (5) adoption of improved methods of resource management (including both wetland resources and agricultural lands) in and around existing protected areas; (6) annual surveys of the main wintering populations; (7) research

on the impacts of human resource use on breeding habitats and breeding behavior; and (8) development of education programs to encourage farmers and other local residents to adopt sustainable resource use practices.

Whooping Crane

The Whooping Crane (Fig. 1.15) is the rarest of the world's 15 crane species. The species' historical decline, near extinction, and gradual recovery is among the best known and documented cases in the annals of conservation (Allen 1952; McNulty 1966; Doughty 1989). Over the last 50 years, a combination of strict legal protection, habitat preservation, and continuous international cooperation between Canada and the

United States has allowed the only surviving wild population to increase steadily from a nadir of just 15 or 16 individuals in 1940-41 to about 150 today. Since the mid-1960s, captive propagation has become increasingly important and now provides security against extinction of the species while affording opportunities to initiate new populations. The species provides an important case study in the conservation of rare and endangered species, and serves as a symbol for international cooperation in conserving not only threatened cranes, but biodiversity in general. The species is classified as Endangered under the revised IUCN Red List criteria.

The Whooping Crane occurs exclusively in North America (Fig. 13.16). The historical mid-continental

breeding range stretched from Alberta across the northeastern portions of the mid-continental prairies nearly to the southern end of Lake Michigan (Allen 1952). The historical wintering grounds included the highlands of northern Mexico, the Texas Gulf coast, and parts of the Atlantic coast. Non-migratory populations occurred in Louisiana and possibly other areas in the southeastern United States. The species declined rapidly in the late 1800s and early 1900s as a result of hunting, collecting, and the conversion of its habitats to agriculture. By 1940, only one self-sustaining flock remained.

As of August 1995, the Whooping Crane numbered 178 in the wild and another 145 birds in captivity. In the wild, the species exists in three separate popula-

tions: the historical Aransas-Wood Buffalo population, an experimental cross-fostered population of only four surviving birds in the Rocky Mountains, and an experimental non-migratory population of released birds in central Florida. Whooping Cranes are maintained in captivity at four locations.

Historically, the species bred primarily in wetlands of the northern tall- and mixed-grass prairies and aspen parklands (Hjertaas 1994). The remnant wild population breeds at the northernmost extreme of the historical range in intermixed muskeg and boreal forest in Canada's Wood Buffalo National Park (Kuyt 1981). During migration, this population uses a variety of feeding and roosting habitats, including croplands, marshes, and submerged sandbars in rivers along the migration route (Lewis 1995). The birds winter in bays and coastal marshes within and near the Aransas National Wildlife Refuge on the Texas Gulf Coast. A population derived from 289 eggs placed in Sandhill Crane nests in the Rocky Mountains peaked at about 35 birds in 1985 but now consists of only four individuals (Ellis et al. 1992). The experimental non-migratory population of 24 captive-reared

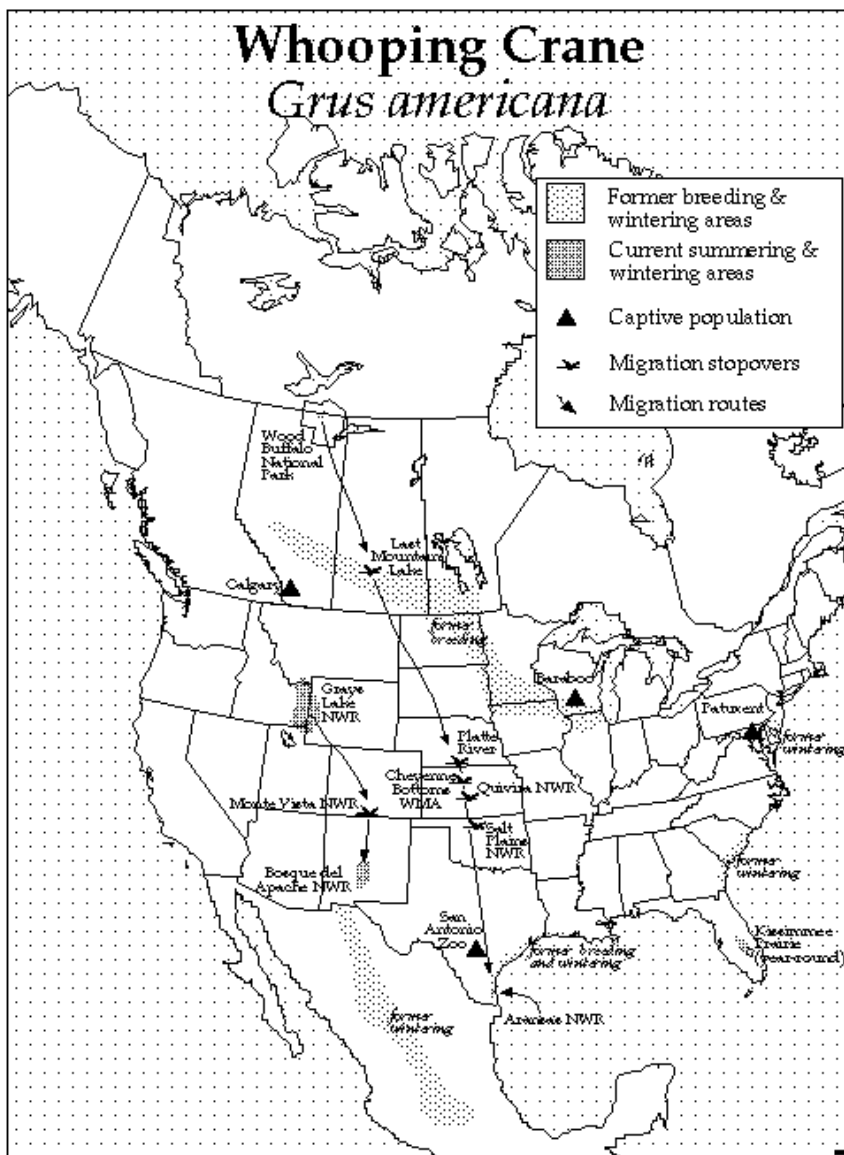


Fig. 13.16. Distribution of the Whooping Crane.

subadults (as of August 1995) results from recent releases in Florida's Kissimmee Prairie. Another 40 birds are due to be released in winter 1995-1996.

Whooping Cranes continue to face multiple threats including habitat loss and air and water pollution in their Texas wintering grounds, collision with utility lines, human disturbance, disease, predation, loss of genetic diversity within the population, and vulnerability to natural and human-caused catastrophes (USFWS 1994). Concern over the near extinction of the Whooping Crane has prompted a broad range of conservation actions including: (1) national and international legal protections; (2) comprehensive scientific research and monitoring programs; (3) protection of key habitats; (4) development of Whooping Crane recovery teams and comprehensive recovery plans in Canada (Edwards et al. 1994) and the United States (USFWS 1994); (5) and extensive public education campaigns.

Priority conservation measures for the future include: (1) full implementation of the U.S. and Canadian Whooping Crane Recovery Plans; (2) special attention to key problems within existing habitats, potential breeding areas, and potential reintroduction sites; (3) continued efforts to establish two additional self-sustaining wild populations and to maintain viable captive populations; and (4) research related to the above topics.

Conclusions

These summary accounts provide only a hint of the tremendous challenges faced by the cranes and those who are working to protect them and the ecosystems that sustain them. Cranes, along with much of the world's biodiversity, will face difficult circumstances in the coming decades. Although their survival, and in some cases recovery, cannot be assured, the steps outlined above will greatly enhance their chances. However, these steps will only be effective if those who are most concerned about, and involved in, crane conservation effectively coordinate their efforts and share their knowledge. Local peoples, as well as the leaders of nations, must participate in these conservation efforts. Only through such shared commitment will cranes continue to grace the world's skies.

Literature Cited

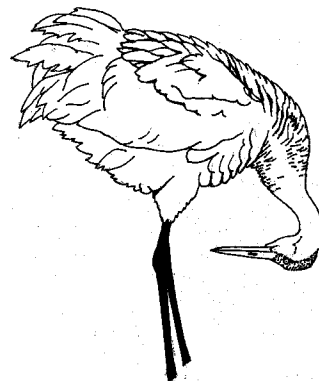
- Ahmad, A. and S. I. H. Shah. 1991. The future of cranes in Pakistan with special reference to the Northwest Frontier Province. Pages 335-339 *in* J. Harris, editor. Proceedings 1987 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Allan, D. G. 1993. Aspects of the biology and conservation status of the Blue Crane *Anthropoides paradiseus* and the Ludwig's *Neotis Ludwigii* and Stanley's *N. denhami stanleyi* Bustards in southern Africa. M. S. thesis, University of Cape Town, Cape Town, South Africa. 242 pp.
- Allan, D. G. 1994. Cranes and farmers. Endangered Wildlife Trust, Parkview, South Africa. 33 pp.
- Allen, R. P. 1952. The Whooping Crane. Research Report No. 3. National Audubon Society, New York. 246 pp.
- Alonso, J. A. and J. C. Alonso, editors. 1990. Distribución y demografía de la Grulla Común (*Grus grus*) en España. ICONA-CSIC, Madrid. 192 pp.
- Andronov, V. A., R. S. Andronova, and L. K. Petrova. 1988. Distribution of Red-crowned Crane nesting territories in the Arkharinski Lowlands. Pages 59-62 *in* N. M. Litvinenko and I. A. Neufeldt, editors. The Palearctic Cranes: biology, morphology, and distribution. Academy of Sciences of the USSR, Vladivostok, USSR. [In Russian with English abstract.]
- Archibald, G. W. 1992a. African cranes for the future. Pages 7-9 *in* D. J. Porter, H. S. Craven, D. N. Johnson, and M. J. Porter, editors. Proceedings of the First Southern African Crane Conference. Southern African Crane Foundation, Durban, South Africa.
- Archibald, G. W. 1992b. Meeting develops plans for Siberians. ICF Bugle 18(1):4-5.
- Arnol, J. D., D. M. White, and I. Hastings. 1984. Management of the Brolga (*Grus rubicundus*) in Victoria. Technical Report Series No. 5. Victoria Fisheries and Wildlife Service, Department of Conservation, Forests, and Lands. 83 pp.
- Aucamp, E. 1996. The breeding biology of the Blue Crane, *Anthropoides paradiseus*, in the Southern Cape, South Africa. R. Beilfuss, W. Tarboton, and N. Gichuki, editors. Proceedings of the 1993 African Crane and Wetland Training Workshop. In press.
- Barzen, J. 1994. ICF team discovers rare wildlife in Cambodia. ICF Bugle 20(4):3-4.
- Beilfuss, R. 1995. Wattled Cranes in the great Zambezi delta. ICF Bugle 21(3):3.
- Bishop, M. A. 1991. A new tune for an ancient harmony. ICF Bugle 17 (4):2-3.
- Bishop, M. A. 1993. The Black-necked Crane winter count 1991-1992. Journal of the Ecological Society (India) 3:55-64.
- Bishop, M. A., Canjue Zhouma, Song Yanling, Gu Binyuan, and J. Harkness. In prep. The status of Black-necked Cranes wintering in Tibet.
- Blackman, J. G. 1977. The development and application of aerial survey methods for population and ecological studies of the Brolga *Grus rubicundus* (Perry) Gruidae. M.S. thesis,

- James Cook University of North Queensland, Townsville, Australia.
- Blakers, M., S. J. J. F. Davies, and P. N. Reilly. 1984. The atlas of Australian birds. Melbourne University Press, Melbourne, Australia.
- Bold, A., N. Tseveenmyadag, and B. M. Zvonov. 1995. Cranes of Mongolia. Pages 42-47 in C. H. Halvorson, J. T. Harris, and S. M. Smirenski, editors. Cranes and Storks of the Amur River: the proceedings of the international workshop. Arts Literature Publishers, Moscow, Russia.
- Brooke, R. K. and C. J. Vernon. 1988. Historical records of the Wattled Crane *Bugeranus carunculatus* (Gmelin) in the Cape Province and the Orange Free State, South Africa. *Annals of the Cape Provincial Museums (Natural History)* 16(15):363-371.
- Chen Bin and Wang Zuoyi. 1991. Observations on wintering ecology of cranes at Poyang Lake. Pages 105-107 in J. Harris, editor. Proceedings 1987 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Chen Bin, Liu Zhiyong, and Tao Fuzhong. 1987. The habits of the wintering White-naped Crane. Pages 57-60 in Poyang Lake Nature, editor. Report on the rare birds winter ecology study at Poyang Lake Migratory Birds Reserve. Jiangxi Scientific Technical Press, Nanchang, Jiangxi.
- Cho, S.-R. and Won, P. 1990. Wintering ecology of the Hooded Crane (*Grus monacha* Temminck) in Korea. *Bulletin of the Institute of Ornithology, Kyung Hee University* 3:1-22.
- Chong, J.-R., H. Higuchi, and P. U-il. 1994. The migration routes and important rest-sites of cranes on the Korean Peninsula. Pages 41-50 in H. Higuchi and J. Minton, editors. The future of cranes and wetlands. Wild Bird Society of Japan, Tokyo, Japan.
- Currier, P. J., G. R. Lingle, and J. G. VanDerwalker. 1985. Migratory bird habitat on the Platte and North Platte Rivers in Nebraska. Platte River Whooping Crane Habitat Maintenance Trust, Grand Island, Neb. 177 pp.
- Daddy, F. and J. S. O. Ayeni. 1996. The status of Nigerian freshwater wetlands and their potential for Black Crowned Crane reintroduction. R. Beilfuss, W. Tarboton, and N. Gichuki, editors. Proceedings of the 1993 African Crane and Wetland Training Workshop. International Crane Foundation, Baraboo, Wis. In press.
- Dietzman, G. and S. Swengel. 1994. 20 years of counting cranes: what have we learned? *ICF Bugle* 20(1):2-3.
- Doughty, R. W. 1989. Return of the Whooping Crane. University of Texas Press, Austin. 182 pp.
- Duc, L. D. 1991. Eastern Sarus Cranes in Indochina. Pages 317-318 in J. Harris, editor. Proceedings 1987 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Edwards, R., S. Brechtel, R. Bromley, D. Hjertaas, B. Johns, E. Kuyt, J. Lewis, N. Manners, R. Stardom, and G. Tarry. 1994. National Recovery Plan for the Whooping Crane. Report No. 6. Recovery of Nationally Endangered Wildlife (RENEW) Committee, Ottawa. 39 pp.
- Eksteen, J. J. 1996. Integrated reserve management at the Verloren Vallei Nature Reserve, Eastern Transvaal, South Africa. R. Beilfuss, W. Tarboton, and N. Gichuki, editors. Proceedings of the 1993 African Crane and Wetland Training Workshop. International Crane Foundation, Baraboo, Wis. In press.
- Ellis, D. H., J. C. Lewis, G. F. Gee, and D. G. Smith. 1992. Population recovery of the Whooping Crane with emphasis on reintroduction efforts: past and future. Proceedings North American Crane Workshop 6:142-150.
- Faanes, C. A. and D. B. Bowman. 1992. Relationship of channel maintenance flows to Whooping Crane use of the Platte River. Pages 111-116 in D. A. Wood, editor. Proceedings 1988 North American Crane Workshop. Florida Nongame Wildlife Program Technical Report #12.
- Flint, V. 1978. Strategy and tactics for the conservation of rare birds. *Priroda* 756 (8):14-29.
- Fry, C. H. 1987. New data on the status of the Black Crowned Crane in West Africa. Pages 331-335 in G. W. Archibald and R. F. Pasquier, editors. Proceedings of the 1983 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Fujimaki, Y., S. Hanawa, K. Ozaki, O. Yunoki, F. Nishijima, V. M. Khrabryi, Y. B. Starikov, and Y. B. Shibaev. 1989. Breeding status of the Hooded Crane (*Grus monacha*) along the Bikin River in the Far East of the USSR. *Strix* 8:199-217.
- Fujita, G., J. Harris, A. Bold, N. Tseveenmayadag, and S. Chuluunbatar. 1994. Habitat preference of Demoiselle and White-naped Cranes, *Anthropoides virgo* and *Grus vipio*, breeding in Mongolia. Pages 93-96 in H. Higuchi and J. Minton, editors. The future of cranes and wetlands. Wild Bird Society of Japan, Tokyo, Japan.
- Galvez, X. and A. Perera. 1995. A crane conservation revival in Cuba. *ICF Bugle* 21(1):2-3.
- Garba, B. 1996. Status and reintroduction potential of the Black Crowned Crane in Nigeria. R. Beilfuss, W. Tarboton, and N. Gichuki, editors. Proceedings of the 1993 African Crane and Wetland Training Workshop. In press.
- Gichuki, N. N. 1993. Factors affecting the reproductive success of the Grey Crowned Crane. Ph.D. dissertation, University of Cambridge, Cambridge, U.K. 196 pp.
- Gichuki, N. N. and C. M. Gichuki. 1991. Relationships between habitat selection and flock characteristics in foraging Grey Crowned Cranes, *Baelearica regulorum*, in Kenya. Pages 357-361 in J. Harris, editor. Proceedings 1987 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Gole, P. 1989. The status and ecological requirements of Sarus Crane: phase I. Ecological Society, Pune, India. 45 pp.
- Gole, P. 1991. The status and ecological requirements of Sarus Crane: phase II. Ecological Society, Pune, India. 35 pp.
- Gole, P. 1993. A Field Guide to Cranes of India. Ecological Society, Pune, India. 46 pp.
- Gui Xiao Jie. 1995. Protection and habitat management for wintering cranes at Dongting Lake of the Yangtze River, China. Pages 163-167 in C. H. Halvorson, J. T. Harris, and S. M. Smirenski, editors. Cranes and storks of the Amur River: proceedings of the international workshop. Arts Literature Publishers, Moscow, Russia.
- Halvorson, C. and F. Kaliher. 1995. A current assessment of cranes wintering in South Korea, January-March, 1992: needs and opportunities. Pages 81-85 in C. H. Halvorson, J. T. Harris, and S. M. Smirenski, editors. Cranes and storks

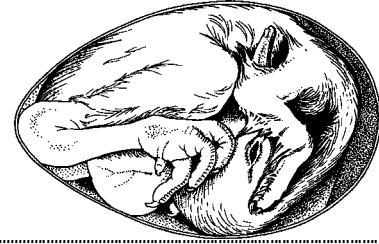
- of the Amur River: proceedings of the international workshop. Arts Literature Publishers, Moscow, Russia.
- Harris, J. 1992. Managing nature reserves for cranes in China. *Proceedings North American Crane Workshop* 6:1-11.
- Harris, J. 1994a. Cranes, people, and nature: preserving the balance. Pages 1-14 *in* H. Higuchi and J. Minton, editors. *The future of cranes and wetlands*. Wild Bird Society of Japan, Tokyo, Japan.
- Harris, J. 1994b. Conservation for people at Cao Hai Nature Reserve. *ICF Bugle* 20(2):1, 4-5.
- Harris, J. T., O. Goroshko, Y. Labutin, A. Degtyarev, N. Germogenov, J. Zhao, N. Zeng, and H. Higuchi. 1995. Results of Chinese-Russian-American investigation of cranes wintering at Poyang Lake Nature Reserve, China. Pages 57-72 *in* C. H. Halvorson, J. T. Harris, and S. M. Smirenski, editors. *Cranes and storks of the Amur River: proceedings of the international workshop*. Arts Literature Publishers, Moscow, Russia.
- Hjertaas, D. 1994. Summer and breeding records of the Whooping Crane in Saskatchewan. *Blue Jay* 52(2):99-115.
- IUCN (World Conservation Union). 1994. IUCN red list categories. IUCN, Gland, Switzerland. 21 pp.
- Iverson, G. C., P. A. Vohs, and T. C. Tacha. 1987. Habitat use by mid-continent Sandhill Cranes during spring migration. *Journal of Wildlife Management* 51:448-458.
- Johnsgard, P. A. 1983. *Cranes of the world*. Indiana University Press, Bloomington. 257 pp.
- Johnson, D. N. 1992. Extent and reasons for crane population declines. Pages 115-116 *in* D. J. Porter, H. S. Craven, D. N. Johnson, and M. J. Porter, editors. *Proceedings of the First Southern African Crane Conference*. Southern African Crane Foundation, Durban, South Africa.
- Katondo, J. 1996. An overview of the status and distribution of the Grey Crowned Cranes in Tanzania. R. Beilfuss, W. Tarboton, and N. Gichuki, editors. *Proceedings of the 1993 African Crane and Wetland Training Workshop*. In press.
- Khachar, S., H. R. Patankar, A. Gaekwad, T. Mundkur, R. Pravez, and R. M. Naik. 1991. Wintering cranes in Gujarat State, India. Pages 327-333 *in* J. Harris, editor. *Proceedings of the 1987 International Crane Workshop*. International Crane Foundation, Baraboo, Wis.
- Konrad, P. M. 1981. Status and ecology of Wattleed Crane in Africa. Pages 220-237 *in* J. C. Lewis and H. Masatomi, editors. *Crane research around the world*. International Crane Foundation, Baraboo, Wis.
- Kovshar, A. V. 1987. The Demoiselle Crane in Kazakhstan and Central Asia. Pages 23-31 *in* G. W. Archibald and R. F. Pasquier, editors. *Proceedings of the 1983 International Crane Workshop*. International Crane Foundation, Baraboo, Wis.
- Krapu, G. L., D. E. Facey, E. K. Fritzell, and D. H. Johnson. 1984. Habitat use by migrant Sandhill Cranes in Nebraska. *Journal of Wildlife Management* 48:407-417.
- Kuyt, E. 1981. Population status, nest site fidelity, and breeding habits of Whooping Cranes. Pages 119-125 *in* J. C. Lewis and H. Masatomi, editors. *Crane research around the world*. International Crane Foundation, Baraboo, Wis.
- Landfried, S. E., A. A. Chaudry, M. M. Malik, and A. Ahmad. 1995. Integrated crane conservation activities in Pakistan: education, research, and public relations. Pages 121-155 *in* S. Jacobson, editor. *Conserving wildlife: international education and communication approaches*. Columbia University Press, New York.
- Lavery, H. J. and J. G. Blackman. 1969. The cranes of Australia. *Queensland Agricultural Journal* 95:156-162.
- Lewis, J. C. 1991. International cooperation in recovery of Whooping Cranes: a model for other nations. Pages 389-394 *in* J. Harris, editor. *Proceedings 1987 International Crane Workshop*. International Crane Foundation, Baraboo, Wis.
- Lewis, J. 1995. Whooping Crane (*Grus americana*). *The Birds of North America*, No. 153. The Academy of Natural Sciences, Philadelphia, and the American Ornithologists' Union, Washington, D. C. 28 pp.
- Li Dehao. 1987. The distribution and status of the Black-necked Crane on the Tibetan Plateau. Pages 45-49 *in* G. W. Archibald and R. F. Pasquier, editors. *Proceedings of the 1983 International Crane Workshop*. International Crane Foundation, Baraboo, Wis.
- Li Fengshan and Li Mingjing. 1991. On the management of Cao Hai, Guizhou Province. Pages 123-126 *in* J. Harris, editor. *Proceedings 1987 International Crane Workshop*. International Crane Foundation, Baraboo, Wis.
- Li Lin. 1995. The first breeding of Hooded Crane in China. Page 55 *in* C. H. Halvorson, J. T. Harris, and S. M. Smirenski, editors. *Cranes and storks of the Amur River: the proceedings of the international workshop*. Arts Literature Publishers, Moscow, Russia.
- Lohmann, L. 1990. Remaking the Mekong. *The Ecologist* 20:61-66.
- Lu Zongbao, Yao Jianchu and Liao Yanfa. 1980. Survey on the breeding biology of the Black-necked Crane. *Chinese Journal of Zoology* 15(1):19-24. [In Chinese].
- Macdonald, I. A. W. 1989. Man's role in changing the face of southern Africa. Pages 51-77 *in* B. J. Huntley, editor. *Biotic diversity in southern Africa: concepts and conservation*. Oxford University Press, Cape Town, South Africa.
- Marchant, S. and P. J. Higgins, editors. 1993. *Handbook of Australian, New Zealand and Antarctic Birds*. Volume 2: Raptors to Lapwings. Oxford University Press, Melbourne.
- Masatomi, H. 1981. The Red-crowned Crane. Pages 81-85 *in* J. C. Lewis and H. Masatomi, editors. *Crane research around the world*. International Crane Foundation, Baraboo, Wis.
- Masatomi, H. 1993. The Tancho of Kushiro Shitsugen. *Japanese Society for Preservation of Birds*, Kushiro, Japan. 34 pp.
- Masatomi, H., S. Ogawa, and K. Miyama. 1990. The relations between land cover monitored by satellite remote sensing and distribution of the Tancho's nests in Kushiro Marsh, Hokkaido. *Journal of the Hokkaido College, Sensu University* 23:209-223.
- McNulty, F. 1966. *The Whooping Crane: the bird that defies extinction*. E. P. Dutton, New York. 190 pp.
- Meine, C. and G. W. Archibald, compilers. 1996. *The cranes: status survey and conservation action plan*. IUCN/SSC Crane Specialist Group, Gland Switzerland. In press.
- Mewes, W. 1994. The increasing population of the Common Crane (*Grus grus*) in Germany and the causes behind this development. Ph.D. dissertation, Martin-Luther-Universität Halle-Whittenberg. 111 pp. [In German].

- Mirande, C., V. Panchenko, R. Belterman, G. Archibald, and U. Seal. In prep. Siberian Crane Population and Habitat Viability Analysis (PHVA) Workshop Report. IUCN Conservation Breeding Specialist Group, IUCN Species Survival Commission, Apple Valley, Minn.
- Mustafa, H. and A. A. Durbunde. 1992 unpubl. Status and distribution of the Black Crowned Crane in West and Central Africa. Paper presented at the International Conference on the Black Crowned Crane and Its Wetlands Habitat in West and Central Africa, February 1992, Kano, Nigeria.
- Nesbitt, S. and K. S. Williams. 1990. Home range and habitat use of Florida Sandhill Cranes. *Journal of Wildlife Management* 54:92-96.
- Neufeldt, I. 1981. The Hooded Crane in the USSR. Pages 239-243 in J. C. Lewis and H. Masatomi, editors. *Crane Research Around the World*. International Crane Foundation, Baraboo, Wis.
- Newton, S. F. 1996. North African crane and wetland action plan. Report of the North and Northeast African Crane and Wetland Working Group. R. Beilfuss, W. Tarboton, and N. Gichuki, editors. Proceedings of the 1993 African Crane and Wetland Training Workshop. International Crane Foundation, Baraboo, Wis. In press.
- Ohsako, Y. 1987. Effects of artificial feeding on cranes wintering in Izumi and Akune, Kyushu, Japan. Pages 89-98 in G. W. Archibald and R. F. Pasquier, editors. Proceedings of the 1983 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Ohsako, Y. 1994. Analysis of crane population change, habitat selection, and human disturbance Pages 107-113 in H. Higuchi and J. Minton, editors. *The future of cranes and wetlands: proceedings of the international symposium*. Wild Bird Society of Japan, Tokyo, Japan.
- Pomeroy, D. E. 1987. The ecology and status of Crowned Cranes in East Africa. Pages 323-330 in G. W. Archibald and R. F. Pasquier, editors. Proceedings of the 1983 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Prange, H., editor. 1989. *Der Graue Kranich. Die Neue Brehm-Bücherei*. A. Ziemsen Verlag, Wittenberg. 272 pp. [In German].
- Prange, H. 1995. Occurrence of cranes in Europe: evolution, protective measures, and future tasks. Pages 393-415 in H. Prange, editor. *Crane research and protection in Europe*. European Crane Working Group and Martin-Luther-Universität, Halle-Wittenberg, Germany.
- Priklonski, S. and Y. Markin. 1982. The number and fluctuation of the Common Crane in the center of European Russia over a twenty-year period. Pages 84-88 in I. A. Neufeldt, editor. *Cranes in the USSR*. Zoological Institute of the Academy of Sciences of the USSR, Leningrad, Russia.
- Pukinski, Y. B. 1977. The Hooded Crane in Primorye. *Okhota i Okhotnichye Kozyaistvo* 1:28-30.
- Sauey, R. 1985. The range, status, and winter ecology of the Siberian Crane (*Grus leucogeranus*). Ph.D. dissertation, Cornell University, Ithaca, New York. 411 pp.
- Schoff, G. 1991. Reflections: the story of cranes. International Crane Foundation, Baraboo, Wis. 40 pp.
- Smirenski, S. M. 1988. Chick relationships and brood sizes in Red-crowned (*Grus japonensis*) and White-naped (*Grus vipio*) Cranes. Pages 49-53 in N. M. Litvinenko and I. A. Neufeldt, editors. *The Palearctic Cranes*. Amur-Ussuri Branch of the USSR Ornithological Society, Vladivostok, USSR. [In Russian. English translation available through ICF].
- Smirenski, S. M., J. T. Harris, C. Gray, K. Strom, and N. Ichida. 1995. Report of the Amur program of [the] Socio-Ecological Union. Pages 205-212 in C. H. Halvorson, J. T. Harris, and S. M. Smirenski, editors. *Cranes and storks of the Amur River: the proceedings of the international workshop*. Arts Literature Publishers, Moscow, Russia.
- Smith, P. J. and J. M. Valentine Jr. 1987. Habitat changes within the Mississippi Sandhill Crane range in Jackson County, Mississippi (1942-1984). Pages 341-354 in G. W. Archibald and R. F. Pasquier, editors. Proceedings of the 1985 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Song Xianjin, Zhao Jinsheng, and Huang Xiangyu. 1995. Crane and stork counts and conservation measures at Poyang Lake Nature Reserve. Pages 73-80 in C. H. Halvorson, J. T. Harris, and S. M. Smirenski, editors. *Cranes and storks of the Amur River: the proceedings of the international workshop*. Arts Literature Publishers, Moscow, Russia.
- Su Liying. 1992. Human impacts on cranes in China. Pages 22-32 in H. Whitaker, editor. *The proceedings of the International Crane Symposium. People, water and wildlife: human population impacts on cranes*. National Audubon Society, Washington, D.C.
- Su Liying. 1993. Comparative feeding ecology of the Red-crowned and White-naped Cranes. M. A. thesis, University of Missouri, Columbia. 91 pp.
- Su Liying, Xu Jie, and Zhou Desheng. 1991. Breeding habits of White-naped Cranes at Zhalong Nature Reserve. Pages 51-57 in J. Harris, editor. Proceedings 1987 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Sudilovskaya, A. M. 1963. Changes in the nesting ranges of the Common Crane, the Siberian Crane, and the Demoiselle Crane in the Soviet Union. *Bulletin of the Moscow Society of Naturalists, Biology Section* 68:125-126.
- Suwal, R. 1995. Crane ecology and bird conservation in Lumbini (1992-93). Pages 35-52 in C. Schimmel, editor. Proceedings of the Third Annual International Crane Symposium: people, water, and wildlife: human population impacts on cranes. National Audubon Society, Boulder, Colo.
- Tacha, T. C., S. A. Nesbitt, and P. A. Vohs. 1992. Sandhill Crane. *The Birds of North America*, No. 31. The Academy of Natural Sciences, Philadelphia, and the American Ornithologists' Union, Washington, D.C. 24 pp.
- Tarboton, W. R. 1992. Extent and reasons for population declines in South Africa's three crane species. Pages 117-118 in D. J. Porter, H. S. Craven, D. N. Johnson, and M. J. Porter, editors. Proceedings of the first southern African Crane Conference. Southern African Crane Foundation, Durban, South Africa.
- Topping, A. 1995. Ecological roulette: damming the Yangtze. *Foreign Affairs* 74(5):132-146.

- Tréca, B. 1996. Wetland habitats for Black Crowned Cranes in west and central Africa. *In* R. Beilfuss, W. Tarboton, and N. Gichuki, editors. Proceedings of the 1993 African Crane and Wetland Training Workshop. In press.
- United Nations Environment Programme/Convention on the Conservation of Migratory Species of Wild Animals (UNEP/CMS). 1995. Memorandum of understanding concerning conservation measures for the Siberian Crane: report of the first meeting of range states. Secretariat of the Convention on the Conservation of Migratory Species of Wild Animals, Bonn, Germany. 113 pp.
- Urban, E. K. 1981. The Sudan Crowned Crane. Page 254 *in* J. C. Lewis and H. Masatomi, editors. Crane research around the world. International Crane Foundation, Baraboo, Wis.
- Urban, E. K. 1996. Status of cranes in Africa, 1994. *In* R. Beilfuss, W. Tarboton, and N. Gichuki, editors. Proceedings of the 1993 African Crane and Wetland Training Workshop. In press.
- Urban, E. K. and N. N. Gichuki. 1991. Recent research and conservation activities with cranes in Africa. Pages 351-355 *in* J. Harris, editor. Proceedings 1987 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- U.S. Fish and Wildlife Service (USFWS). 1994. Whooping Crane Recovery Plan. U.S. Fish and Wildlife Service, Albuquerque, New Mexico. 92 pp.
- Vernon, C. J., A. F. Boshoff, and W. S. Stretton. 1992. The status and conservation of cranes in the Eastern Cape Province. Pages 47-72 *in* D. J. Porter, H. S. Craven, D. N. Johnson, and M. J. Porter, editors. Proceedings of the first southern African crane conference. Southern African Crane Foundation, Durban, South Africa.
- Walkinshaw, L. H. 1949. The Sandhill Cranes. Cranbrook Institute of Science Bulletin 29:1-202.
- Walkinshaw, L. H. 1973. Cranes of the world. Winchester Press, New York. 370 pp.
- Wanjala, M. 1996. The Kipaina Wetland Conservation Organization. *In* R. Beilfuss, W. Tarboton, and N. Gichuki, editors. Proceedings of the 1993 African crane and wetland training workshop. International Crane Foundation, Baraboo, Wis. In press.
- Wei Tianhao, Wu Jingliang, Huang Guozu, Wu Jiyu, Li Zongqiang, and Han Lianxian. 1994. Investigation on Black-necked Crane (*Grus nigricollis*) in Yunnan. Pages 54-60 *in* Waterbird Research in China. Waterbird Specialist Group of the Chinese Ornithological Association. East China Normal University Press, Shanghai.
- White, D. M. 1987. The status and distribution of the Brolga in Victoria, Australia. Pages 115-131 *in* G. W. Archibald and R. F. Pasquier, editors. Proceedings of the 1983 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Winter, S. V., Y. A. Andryushchenko, and P. I. Gorlov. 1995. The Demoiselle Crane in the Ukraine: status, ecology, and conservation prospects. Pages 285-289 *in* H. Prange, editor. Crane research and protection in Europe. European Crane Working Group and Martin-Luther-Universität, Halle-Wittenberg, Germany.
- Won, P. O. 1981. Status and conservation of cranes wintering in Korea. Pages 61-65 *in* J. C. Lewis and H. Masatomi, editors. Crane research around the world. International Crane Foundation, Baraboo, Wis.
- Xu Xinjie, Liu Hongxi, Wang Jiangtang, and Zhang Wancang. 1991. Cranes in the place where the Yellow River flowed long ago in Henan Province. Pages 153-154 *in* G. W. Archibald and R. F. Pasquier, editors. Proceedings of the 1983 International Crane Workshop. International Crane Foundation, Baraboo, Wis.
- Zhou Fuzhang, Ding Wenning, and Wang Zhiyu. 1981. A large flock of White Cranes (*Grus leucogeranus*) wintering in China. Acta Zoologica Sinica 27(2):179.



Equipment and Suppliers



The following is a list of sources of equipment and supplies mentioned in the various chapters. Much of the equipment needed for crane propagation is nonspecialized and can be obtained locally from hardware stores, livestock equipment and feed dealers, laboratory supply stores, and other sources. This list is not meant to be all inclusive, nor is it an endorsement of any particular product or supplier.

Artificial Insemination and Cryopreservation

Beltsville Poultry Semen Extender:

Continental Plastics Corporation
P. O. Box "C"
540 South Second Street
Delvan, WI 53115
(414) 728-4800

International A.I., Inc.
7909 South Fairfax
Bloomington, IN 47401
(812) 824-2473, (800) 274-2824

Tri Bio Laboratories, Inc.
1400 Fox Hill Road
State College, PA 16803
(814) 355-1541

Cryogenic equipment:

Brinkman Instruments, Inc.
1 Cantiague Road
Westbury, NY 11590-9974
(516) 334-7500, (800) 645-3050

MVE Cryogenics
Minnesota Valley Engineering
407 7th Street NW
New Prague, MN 56071
(612) 758-4484

Praxair Inc.
P. O. Box 44
Tonawanda, NY 14150-7891
(716) 879-2000, (800) 621-7100

Histological mounting medium (Permount):

Fisher Scientific Co.
711 Forbes Ave.
Pittsburgh, PA 15219
(714) 669-4600, (800) 766-7000

Insulated multipurpose bath with stirrer (Neslab Agitator):

Neslab Instruments, Inc.
P. O. Box 1178
Portsmouth, NH 03802-1178
(603) 436-8411, (800) 463-7522

Refrigeration compressor immersion probe (Neslab Cryocool Immersion Cooler CC-100II):

Neslab Instruments, Inc. (above)

Temperature recording unit (Honeywell Elektronik II, Type T):

Honeywell Industrial Division
1100 Virginia Ave.
Ft. Washington, PA 19034
(215) 641-3000, (800) 328-5111

Tube sealer and holder (Seal-ease):

Becton Dickinson and Co.
1 Becton Drive
Rutherford, NJ 07070
(201) 847-6800

(Parafilm):

Fisher Scientific Co.
711 Forbes Ave.
Pittsburgh, PA 15219
(714) 669-4600, (800) 766-7000

Chick Rearing/Incubation

Candlers:

Lyon Electric Co.
2765 Main Street
Chula Vista, CA 91911
(619) 585-9900

Georgia Quail Farm
2343 Louisville Road
P. O. Box 1552
Savannah, GA 31498
(912) 236-0651

Valentine Inc.
4259 S. Western Blvd.
Chicago, IL 60609
(312) 650-9050, (800) 438-7883

Chickens (adult stock, chicks, hatching eggs):

Murray McMurray Hatchery
Webster City, Iowa 50595
(515) 832-3280

Stromberg's Chicks and Pets Unlimited
P. O. Box 400
Pine River, MN 56474
(218) 587-2222

Egg repair

(Nexaband, surgical-grade cyanoacrylate):
Tri-Point Medical L.P.
5265 Capital Blvd.
Raleigh, NC 27604
(919) 876-7800, (800) 334-8560

Incubators and brooders:

Georgia Quail Farm (above)

Hawkhead International Inc.
(also sells used, mainly Robbins, equipment)
General Office Suite 158
200 Industrial Loop
Orange Park, FL 32073
(904) 264-4295

Humidaire Incubator Co.
217 West Wayne Street
New Madison, OH 45346
(513) 996-3001

Lyon Electric Co. (above)

Petersime Incubator Co.
300 North Bridge Street
Gettysburg, OH 45328
(513) 447-2151 or 447-7171

Stromberg's Chicks and Pets Unlimited (above)

Valentine Inc. (above)

Feed and Food Supplements

Prepared crane feed—local feed mills may also custom-mix feeds according to formulas given:

Garver Feed Co.
3244 Atwood Ave.
Madison, WI 53701
(608) 244-4739

Zeigler Bros., Inc.
Box 95
Gardners, PA 17324
(717) 677-6181, (800) 248-3007

Supplemental food, complete diet, for tube feeding (Emeraid I and II):

Lafeber
RR #2
Odell, IL 60460
(815) 358-2301

Supplemental food ingredients**(Nutri-cal):**

EVSCO Pharmaceuticals
IGI Affiliate
P. O. Box 209 (Harding Hwy.)
Buena, NJ 08310
(609) 691-2577

(Prosobee):

Mead Johnson Nutritionals
2400 W. Lloyd Expressway
Evansville, IN 47721
(812) 429-5000, (800) 222-9123

(Vionate, vitamin supplement):

Gimborn U.S.
4280 N.E. Expressway
Atlanta, GA 30340
(770) 454-3200, (800) 755-7056

Tubes, for tube feeding:

Henry Schein Inc.
5 Harbor Park Drive
Port Washington, NY 11050
(516) 843-5500, (800) 872-4346

General Husbandry/Facilities

General animal care equipment:

Valentine Inc.
4259 S. Western Blvd.
Chicago, IL 60609
(312) 650-9050, (800) 438-7883

Bands, metal:

Gey Band and Tag Co.
P. O. Box 363
Norristown, PA 19404-0363
(215) 277-3280

Old Dominion Mfg.

19101 Duval Road
Moseley, VA 23120
(804) 739-3944

National Band and Tag Co.

721 York Street, P. O. Box 430
Newport, KY 41072-0430
(606) 261-2035

A. C. Hughes

1 High Street
Hampton Hill
Middlesex, England TW12 1NA

Lambournes, Ltd.

Shallowford Court off High St.
Henley in Arden, Solihull
West Midlands, England B95 5BY

I. O. Mekaniska HB

Eric Ohman
P. O. Box 98
S-560 23 Bankeryd, Sweden

Danieleson's (anodizing)

Box 30-S 194-21
Upplands-Zansby, Sweden

Acraft Sign and Nameplate Co.

10432 59th Avenue
Edmonton, Alberta, Canada
(403) 436-1098

Bands, colored:

(Engraved bands)
Allard Trophy Co.
Valley City, ND 58072
(701) 845-3303

Avinet, Inc.
P. O. Box 1103
Dryden, NY 13052-1103
(607) 844-3277

J. T. W. Anderson (Darvic)
4 Elm Place
Aberdeen, Scotland AB2 335W

National Band and Tag Co. (above)

Gey Band and Tag Co. (above)

A. C. Hughes (above)

Danieleson's (above)

Band materials, laminated plastics:

Chesapeake Wildlife Heritage
P. O. Box 1745
Easton, MD 21601
(410) 822-5100

Lynn Plastics Sales Co.
3 Liberty Street
Merrimac, MA 01860
(508) 346-8182

(Gravoply):
New Hermes, Inc.
2200 Northmont Parkway
Duluth, GA 30136
(404) 623-9697, (800) 843-7637

Spinner Plastics
1108 N. First St.
Springfield, IL 62702
(217) 523-8585; FAX (217) 523-2399

Band materials, lubricant (Pam):

American Home Foods Products Inc.
Five Giralda Farms
Madison, NJ 07940
(800) 544-5680

Disinfectants:

American Scientific Products
1439 Waukegon Road
McGaw Park, IL 60085
(312) 689-8410

(Environ/One Stroke Environ):
Calgon Vestal Laboratories
Box 147
St. Louis, MO 63166
(800) 345-0473

Holt Products, Inc.
613 Atlas Ave.
Box 8185
Madison, WI 53708
(608) 221-4367

Midwest Vet Supply
12012 12th Avenue South
Burnsville, MN 55337
(612) 894-4350, (800) 328-2975

Feeders:

Valentine Equipment Co. (above)

Fencing, wire:

J. A. Cissel
Squankum-Yellowbrook Road
Farmingdale, NJ 07727
(908) 901-0300, (800) 631-2234

Valentine Equipment Co. (above)

Flight netting for pens (Toprite XL netting):

J. A. Cissel (above)

Nicholas Net & Twine
2200 Highway 111
Granite City, IL 62040
(618) 797-0211

Photoperiod lights (use high intensity discharge lamps: mercury, metal halide (Multivapor), or high pressure sodium (Lucalox)):

General Electric
Nela Park
Cleveland, OH 44112
(800) 327-0097

Space heaters:

Animal Spectrum, Inc.
P. O. Box 6307
Lincoln, NB 68506
(800) 228-4005

Transponders (Trovan/A.E.G. Transponder System):

(U.S. source)
InfoPET Identification Systems
415 West Travelers Trail
Burnsville, MN 55337
(618) 890-2080

(European source)
EuroID
Grossbuellesheimer Str. 56
535 Euskirchen 16, Germany

Water heaters, coil and pole-type (tube):

Nelson Manufacturing Co.
Box 636
Cedar Rapids, IA 52406
(319) 363-2607

Genetics/Records

Conservation Breeding Specialist Group
(CBSG)/ISIS
Minnesota Zoological Garden
12101 Johnny Cake Ridge Road
Apple Valley, MN 55124-8199
(612) 431-9295

Medical and Surgical Supplies

General veterinary medical/surgical supplies including instruments, syringes, pharmaceuticals and reagents:

A. J. Buck & Son, Inc.
11407 Cronhill Drive
Owings Mills, MD 21117
(301) 581-1800, (800) 638-8673

Blood counts (Eosinophil Unopette, Unopette, Microtainer tubes):

Becton-Dickinson & Co.
1 Becton Drive
Rutherford, NJ 07417
(201) 847-6800

Calphosan:

Glenwood, Inc.
82 N. Summit St.
Tenafly, NJ 07670
(201) 569-0050

Cameo Quick Stain II:

Cambridge Diagnostic Products
6880 NW 17th Avenue
Ft. Lauderdale, FL 33309
(305) 971-4040

Centrifuge (IEC Centra-4B Centrifuge):

International Equipment Co.
300 2nd Ave.
Neeham Heights, MA 02194
(617) 449-8060, (800) 843-1113

Diff-Quick:

A. J. Buck & Son, Inc. (above)

Dermaclens:

Smith Cline Beecham Clinical Laboratories
2727 W. Baseline, Suite 8
Tempe, AZ 85283
(602) 438-8477, (800) 829-7225

Dremel tool

Dremel
Division of Emerson Electric Co.
4915 21st Street
Racine, WI 53406
(414) 554-1390

Ellman Cautery Unit:

Ellman International Mfg. Inc.
1135 Railroad Ave.
Hewlett, NY 11557
(516) 569-1482, (800) 835-5355

Endoscopes:

Karl Storz Veterinary Endoscopy
175 Cremona Drive
Goleta, CA 93117
(805) 968-7776, (800) 955-7832

Richard Wolf Medical Instruments Corp.
7046 Lyndon Avenue
Rosemont, IL 60018
(312) 298-3950, (800) 329-9653

Schott Fiber Optics, Inc.
122 Charlton St.
Southbridge, MA 01550
(508) 765-9744, (800) 343-6120

Glutaraldehyde (Glutarex):

VWR Scientific
7211 Hanover Parkway, Suite D
Greenbelt, MD 20770-9888
(800) 932-5000

Hemaglobinometer:

Curtin Matheson Scientifics
500 American Road
Morris Plains, NJ 07950
(800) 640-0640

Heparinized capillary tubes:

Midwest Vet Supply
12012 12th Avenue South
Burnsville, MN 55337
(612) 894-4350, (800) 328-2975

Medical diagnostic services:

Medical Diagnostic Services
P. O. Box 1441
Brandon, FL 33509
(813) 653-1180, (800) 435-9352

Moldable cast material

(Orthoplast):
Johnson and Johnson
325 Paramount Drive
Raynham, MA 02767
(800) 526-2459

(Rolyan Polyflex II):
Smith & Nephew Rolyan Inc.
P. O. Box 1005
Germantown, WI 53022-8205
(414) 251-7840, (800) 288-3693

Nebulization equipment

(Ultra-Neb 99):
Sunrise Medical
DeVilbiss Health Care Division
1200 E. Main St.
Somerset, PA 15501
(814) 443-4881, (800) 338-1988

(Snyder Oxygen Cage Model ATC-32):
Snyder Manufacturing Co.
5500 E. Pacific Place
Denver, CO 80222
(303) 756-1932, (800) 422-1932

Nolvasan:

Fort Dodge Laboratories Inc.
800 5th Street NW
Fort Dodge, Iowa 50501
(515) 9554600

Self-curing dental acrylic (Lang's Jet Acrylics):

Lang Dental Manufacturing Co.
175 Messner Drive
P. O. Box 969
Wheeling, IL 60090
(708) 215-6622, (800) 222-5264

Surgical lubricant:

E. Fougera & Co.
60 Baylis Road
Melville, N.Y. 11747
(516) 454-6996

Vetwrap:

3M Animal Care Products
Building 225-1N-07
3M Center
St. Paul, MN 55144
(612) 733-8477, (800) 364-3577

Vitamin A, injectable form (Aquisol A):

Astra U.S.A., Inc.
50 Otis Street
Westboro, MA 01581-4500
(508) 366-1100

Predator and Pest Management

Arthropod control (Wasp Freeze; Permanone):

Summit Chemical Co.
7657 Canton Center Dr.
Baltimore, MD 21224
(410) 282-5200

Frightening agent for birds (Avitrol):

Summit Chemical Co. (above)

Fumigant (Giant Destroyer Cartridge):

Atlas Chemical Corporation
P. O. Box 141
Cedar Rapids, Iowa 52406
(319) 377-8921

Mammal traps**(Tomahawk Model 108; 110A, B, or C):**

Tomahawk Live Trap Co.
P. O. Box 323
Tomahawk, WI 54487
(715) 453-3550, (800) 272-8727

(Live traps, Havahart):

Valentine Inc.
4259 S. Western Blvd.
Chicago, IL 60609
(312) 650-9050, (800) 438-7883

(Victor, Conibear, and Soft Catch traps):

Woodstream Corporation
69 North Locust Street
Lititz, PA 17543
(717) 626-2125, (800) 800-1819

Non-selective herbicide (Roundup):

Monsanto Co.
Agricultural Products
Solaris P. O. Box 5008
San Ramon, CA 94583
(800) 225-2883

**Poisoned bait for rodents (Quintox,
Eaton's Bait Blocks):**

Summit Chemical Co. (above)

Starlicide Complete:

Metro Feeds Ltd.
P. O. Box 64768
Baltimore, MD 21264

Sexing Laboratories

Karyotyping using blood cells:

Avian Genetic Sexing Laboratory (Avigen)
6551 Stage Oaks Dr., Suite 3A
Bartlett, TN 38134
(901) 388-9548

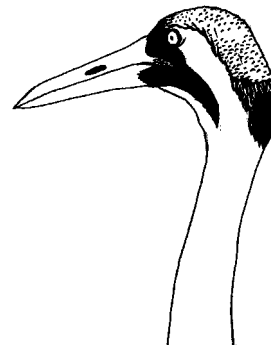
Arlene Kumamoto
San Diego Zoo
P. O. Box 551
San Diego, CA 92112-0551
(619) 231-3953

Commercial feather pulp sexing:

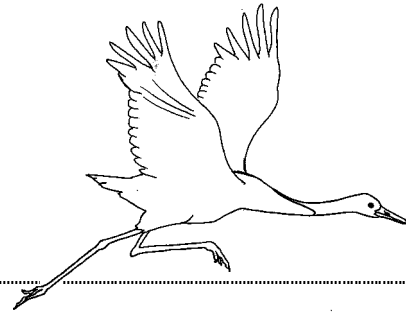
Avigen (above)

Peter van Tuinen
2151-C Kensington Drive
Waukesha, WI 53188
(414) 544-4279

Zoogen, Inc.
1105 Kennedy Place, Suite 4
Davis, CA 95616
(916) 756-8089



Index



- Acanthocephala* (acanthocephalan) 103, 160
acclimation (acclimating) 117, 231-233, 239, 262
acidosis 141, 153, 164
adoption(s) 45-46, 61, 85-86, 267, 271, 273, 282, 284, 286
age distribution 176
age structure 176-177
aggression 23-24, 29, 62, 78, 84-86, 90-92, 101, 108, 110, 113-115, 136, 138, 160-161, 205, 225, 236, 258-259
aggressive displays 106, 109, 114, 119
air sac(s) 138-140, 156, 161, 170, 205, 207
allele(s) 175-178, 181
anatomy 3, 123, 129, 134-136, 155, 166, 168, 205, 207
androgens 124-127, 130, 136
anemia 140-141, 143, 147, 150-151
anesthesia (anesthetized) 137, 141, 146, 154, 168-171, 173, 186, 227, 262
anesthetic 143, 146, 166, 168, 173, 242, 244
angel wing 101
Animal Care and Use Committee 31
Animal Record Keeping System (ARKS) 185-186, 193, 196, 199, 202, 204
Animal Welfare Act 31
Anthropoides paradisea (*A. paradiseus*) 2, 271, 288
Anthropoides virgo 2, 76, 172-173, 272, 289
antibiotic(s) 32, 72, 80, 87, 95-97, 100, 141-142, 155-157, 160-162, 164, 166, 171-172, 210
antifungal 97, 156-157
antihelminthics 96
aqua therapy (see swimming) 90, 257
arthritis (see osteoarthritis) 102, 162, 166
artificial insemination (AI) 25, 34, 37-38, 49, 52, 56, 84, 113-116, 134, 140, 189-190, 192, 197, 199-200, 202-207, 209-217, 221-222, 228, 241, 259
artificial selection 177
Aspergillus (aspergillosis) 32, 97, 140, 152, 156, 256
attachment 90, 117, 119, 247, 259
auscultation 140
avian pox 138, 143, 157, 174
bacteria (bacterial) 32, 35, 60-61, 64, 66, 69, 72, 88-89, 96-98, 140-142, 151-152, 155-157, 165-166, 209-211, 216-217
Balearica pavonina 2, 267
Balearica pavonina ceciliae (*B. p. ceciliae*) 2, 263-264, 267
Balearica pavonina pavonina (*B. p. pavonina*) 2, 263-264, 267
Balearica regulorum 2, 29, 75, 133, 268, 289
Balearica regulorum gibbericeps (*B. r. gibbericeps*) 2, 264, 268
Balearica regulorum regulorum (*B. r. regulorum*) 2, 264, 268
bandage (bandaging) 101-102, 146, 166-167
bands 41-42, 85, 181, 238-239
beak fractures 161, 173
beak repair 169, 171-172
beak trimming 100, 137
Black Crowned Crane 2, 4, 26, 5, 223, 263-264, 267, 289, 291
Black-necked Crane 109, 179, 266, 284-285, 288, 290, 292
blood collection 144, 146
blood glucose 99, 144
blood smears 146-147, 153
blood typing 181-182
Blue Crane 2, 26-27, 29, 8, 76, 179, 225, 264, 270-271, 288
body condition index (BCI) 98, 139
botulism 164, 171-172
bowed legs 83, 102
brailing 41, 114, 239, 241, 243-244
breeding objectives 176, 182, 204
breeding pens 77, 260-262
breeding season 3, 23-24, 33-34, 45, 49, 51-52, 55, 85, 87, 93, 110, 112, 115, 124, 186, 188, 192-193, 196, 200, 202, 228, 251, 259, 271, 276, 279, 284
Brolga 2-3, 26, 14, 34, 39, 48, 50, 55, 67, 83, 107, 224-226, 262, 265, 280-281, 288, 292
brooder 77-78, 85, 87-88, 91-93, 236, 253-254, 257
broom 37, 87

- Bugeranus carunculatus* 2, 120, 270, 289
bumblefoot (see pododermatitis) 102, 140-141, 161, 165-166, 174
- cachexia 98-99
calcium 34-36, 51, 54, 62, 79, 130, 145, 150-152, 162
candling 60, 65-66, 68-69, 72, 74
Capillaria (capillarids) 103, 153, 158, 160
captive management programs 178
capture corners 36, 115, 258-260
capture myopathy 38, 43, 141, 143, 152, 164, 172-174
carcinoma(s) 155, 163
cardiac disease 152
carrying capacity 176-177
central nervous system 124, 152
cestodes 153, 158-159
Chlamydia 142, 152
climate 47, 260
cloaca 98, 128-129, 134, 162-163, 170, 190, 205-210, 212-214, 228
cloacal prolapse 97
Clostridium 32, 156, 164
clutch 23-24, 52-54, 60, 68, 85-86, 116, 130, 200
coccidia (coccidiosis) 97, 103-104, 153, 158-159, 172-173
conditioning 94-95, 115, 118, 205, 207, 233, 237, 253
copulation 45, 48, 106, 110, 114-115, 121, 128, 205-209, 241
cornea (corneal) 37, 100, 104, 161
corticosteroids 141-142, 160-161, 164
costume 93-94, 118, 232, 234-237
costume-rearing (costume-reared) 93-95, 117-119, 234, 236-239, 257
courtship 45, 48
crates 40-41
cross-fostering (cross-fostered) 85, 104, 117-118, 120, 233-234, 240, 287
cryopreservation (cryopreserved) 176, 199, 216, 219, 221-222
culling 176
culture(s) 60, 72, 96-97, 100, 141, 155-156, 162, 166, 193, 226-229
culturing 141, 156
Cyathostoma 103, 153, 158
cytology 97, 172
- daylength (see photoperiod) 46-47, 52
dehydrate 207, 221
dehydration 73, 79-80, 89, 97-98, 101, 123, 136, 140-141, 152, 164, 209
demography (demographic) 175-178, 183, 185-186, 196
Demoiselle Crane 2, 26, 29, 9, 20, 172, 223, 264, 271-273, 290-292
diarrhea 97-98, 139, 153, 155, 158, 162, 171
diet(s) 3, 34-35, 41, 43, 51, 55, 57, 62, 78-79, 83, 89-90, 99, 101-102, 104, 144, 152, 164-166, 174, 237, 269, 284
differential cell count 147
dimethylsulfoxide (DMSO) 220-222
disease 25, 32, 45, 49, 59, 61, 65, 67, 86, 92, 97-99, 103-104, 123, 132, 137-138, 140-141, 150-153, 156-158, 164, 166, 170-175, 182, 192, 204-205, 234, 246, 248-250, 252, 262, 276-277, 283, 288
disinfect 32, 36, 60, 72, 78, 253, 262
disseminated visceral coccidiosis (DVC) 97, 158
disturbance 31-34, 41, 43, 45, 51-52, 54, 56, 112-113, 115-116, 121, 123, 132, 135, 161, 212, 247, 251, 256, 259, 269, 272, 275-276, 279, 281, 286, 288, 291
DNA 3, 28, 181-183, 223, 227, 229
drinking water 34, 36, 61
dummy eggs 45, 54-55, 60, 62, 85-86
- eastern equine encephalitis (EEE) 33, 157, 171-172, 250, 252
ectoparasites 159-160, 250
effective population number 175
egg(s) 3, 22-26, 32-34, 45-57, 59-76, 84-86, 94-96, 103, 106, 110, 112-117, 124-127, 129-132, 134-136, 139, 143, 152-153, 157-158, 160, 162-163, 170, 173, 181-182, 186, 188, 190, 192-193, 196-197, 199-203, 206-213, 215-216, 219, 228, 231, 233, 235-236, 239, 245-246, 248, 251, 279, 284, 287
egg binding 143, 152, 162
egg breakage 51, 53-54, 63
egg laying interval 52-53, 199-200, 202
egg removal 53, 199
egg retention 162
Eimeria 103, 153, 158, 160
electric fencing 246
emaciation 98-99, 139, 144
endocrine disorders 152
endogenous rhythms 46, 124

- endoscope (endoscopic) 170, 174
 energy requirements 98
Escherichia coli 96, 139, 155-156
 estrogens 124-127, 129-130
Eucoleus 153, 158
 Eurasian Crane 2, 25-26, 15, 106, 265, 281-282
 euthanasia 176
 exercise 34, 61-62, 77-78, 83-84, 87, 90-93, 101-103,
 110, 165, 234, 255-257
 exhibit pens 261-262
 eye injuries 78, 100
- facilities 31, 39, 45, 48, 54, 59-60, 77-78, 84, 90-91,
 94-95, 152, 158, 160-161, 211-212, 237, 246-248,
 250-251, 253, 255, 257-259, 261-262, 274
 fallow pen (see pen rotation) 31-32, 158
 fecal examination(s) 103, 153, 193
 feeding (feed) 3, 26, 33-36, 38, 41, 43, 51, 56, 62,
 78-79, 84-85, 88-89, 91-92, 94-95, 97, 99-101,
 108, 113-114, 117-119, 125, 144, 154, 159, 164,
 168-169, 209, 236-239, 248-251, 257-258,
 260-262, 268, 271-273, 276-278, 283-287, 291
 fertility 54, 60, 66-69, 76, 103, 110, 116, 134, 136,
 155, 175-177, 190, 197, 205, 208-213, 215-217,
 219, 221-222
 fitness 177
 flotation 60, 66, 69, 153, 158
 fluid therapy 96-97, 99, 101, 104, 140-141, 144,
 162, 174
 follicle stimulating hormone (FSH) 124-127,
 130-131
 food storage 35, 246
 footbath 32
 foreign body 162, 170-171, 173
 formaldehyde 65
 fossil 1, 28
 founder 176, 182, 219
 fracture(s) 38, 103, 137, 140, 143, 161-162, 165,
 167-169, 172-173
 fumigate 65
 fungal infection(s) 51, 72, 97, 151, 156
- gapeworm(s) (see *Syngamus*) 103, 153, 158
 gastrointestinal 98-99, 150-151, 154-155, 158,
 170-171
 genealogy 56, 177, 185, 204
 generation time 177
 genetic bottleneck 177, 181
 genetic defects 59
 genetic diversity (diversity) 175-178, 181-183, 219,
 288, 290
 genetic drift 176
 genetic fingerprinting 181
 genetic load 177
 genetic management 27, 175-177, 179, 181, 183, 185
 genetic representation 176
 genetics 102, 182-183, 227
Giardia 153
 gonadotropins 124, 130
 gout 140, 152, 162
 Gray Crowned Crane 2, 4, 26, 6, 123, 264, 268
 growth 29, 32, 34-35, 43, 54, 60-61, 66, 78, 80-84,
 86-87, 89-90, 97, 100-104, 121, 124-127, 129-130,
 136, 162-163, 165, 174-177, 186, 188, 256,
 268, 279
Grus americana 2, 104, 121, 173-174, 183, 228,
 287, 290
Grus antigone 2, 172, 279
Grus antigone antigone (*G. a. antigone*) 2, 265,
 278-279
Grus antigone gilli (*G. a. gilli*) 2, 265, 278-279
Grus antigone sharpii (*G. a. sharpii*) 2, 265, 278-279
Grus canadensis 2, 29, 56, 104, 120, 135, 172-174,
 217, 276
Grus canadensis canadensis (*G. c. canadensis*) 2, 265
Grus canadensis nesiototes (*G. c. nesiototes*) 2, 265
Grus canadensis pratensis (*G. c. pratensis*) 2, 265
Grus canadensis pulla (*G. c. pulla*) 2, 56, 172, 265
Grus canadensis rowani (*G. c. rowani*) 2, 265
Grus canadensis tabida (*G. c. tabida*) 2, 104, 120,
 174, 265
Grus grus 2, 27, 282, 288, 290
Grus grus grus (*G. g. grus*) 2
Grus grus lilfordi (*G. g. lilfordi*) 2
Grus japonensis 2, 28-29, 56, 120-121, 183, 229,
 286, 291
Grus leucogeranus 2, 29, 180, 274-275, 291-292
Grus monacha 2, 283, 289
Grus nigricollis 2, 285, 292
Grus rubicunda (*G. rubicundus*) 2, 280, 288
Grus vipio 2, 29, 56, 104, 120, 172, 204, 278,
 289, 291
 Guard-call 112, 223, 225, 237

- hand-rearing (hand-reared) 33, 37, 77-78, 80-84, 87-95, 97, 101-102, 111, 114-117, 120-121, 188, 192, 201, 225, 231-234, 236, 240, 253, 256
- handling 36, 39, 60, 65, 68, 72, 74, 80-81, 84, 86, 89, 95, 98, 101, 116, 152, 154, 160-161, 164, 169, 199, 208-209, 212, 214, 236, 251, 261
- hatch (hatching) 23, 34, 42, 46, 54-56, 59-61, 63, 65-67, 69-73, 75-82, 85-88, 90-91, 94-96, 99, 104, 117, 136, 140, 165, 177, 180, 186, 188, 193, 200, 202-204, 236, 253, 257
- hatchability 54, 59-60, 65, 71-76, 134, 136, 177, 192, 216
- heat stress 29, 40, 98, 135
- hematocrit 141, 143, 145, 147, 150-151
- hematology 144, 153, 172-174
- hematoma 140, 146
- hemoglobin 145, 147-148, 150
- Hemoproteus* 158
- heterozygosity 177, 182
- Hexamita* 153, 158
- hobbling 102
- Hooded Crane 2, 26, 16, 105, 121, 179, 266, 282-284, 289-291
- hormonal status 45
- hormone(s) 52, 124-127, 129-131, 133-136, 228
- humidity 35, 47, 59, 63-66, 71, 73, 96, 98, 124, 193, 209, 253
- hydrotherapy (see swimming, aqua therapy) 78, 103
- hyperglobulinemia 152
- hypoglycemic 99, 141
- impaction 138-139
- imprinting 25, 77-78, 83, 85, 87, 91-95, 117-122, 231-238, 254-255, 257
- inbreeding 59, 61, 67, 177-178, 183, 185, 199
- inclusion body disease (IBDC) 151, 157, 171
- incubation 23, 28, 45, 53-56, 59-71, 73-76, 85-86, 88, 96, 101-102, 104, 111-112, 123, 125, 131-132, 188-189, 193, 199-203, 235, 253
- incubators 33, 53, 59, 61, 63-66, 68, 72-74, 77-78, 200, 253, 257
- infertility 67-68, 205, 209
- inseminating equipment 211
- insemination (inseminate) 25, 33-34, 56, 61, 84, 113, 116, 134, 140, 181, 189, 197, 200, 202, 204-217, 221-222, 228, 241
- International Species Information System (ISIS) 25, 150, 178, 185-186, 202, 204
- intravenous therapy 140-141, 174
- isolation-rearing (isolation-reared) 85, 93-94, 240
- laceration(s) 80, 100, 161, 165, 168, 170
- laparoscopy 155, 173, 223, 227-229
- laparotomy 162
- latitude 46-48, 50, 111, 132, 154
- lead poisoning 150, 152, 163, 173-174
- leg injuries 38, 81, 165-166
- leg rotation 83, 102-103
- Leucocytozoon* 158
- lifespan 51-52
- live-dead count 211
- liver disease 152-153
- longevity 25, 28-29, 52, 176
- luteinizing hormone (LH) 124-127, 130-131, 133-136
- luteinizing hormone releasing hormone (LHRH) 124-127, 130, 132
- luxations 140, 165, 168
- Macracanthorhynchus* 153
- macrochromosomes 226
- Major Histocompatibility Complex (MHC) 182-183
- malpositions 70-72, 75
- massage(s) 205-208, 212-213, 215, 219, 228
- methionine 35, 89, 165
- minimum viable population 176-177
- molt 63, 111, 113, 123, 131-132, 134-135, 228, 242
- mortality 25, 37, 59, 63, 68-69, 71, 75, 84, 87, 157, 163-164, 172-176, 185, 188, 192-193, 196, 199, 202-203, 231, 233, 246-247, 250, 252, 274
- multiple clutching 52, 54
- muscle relaxant 143, 161
- Mycobacterium* 155
- mycotoxins 164, 174
- myopathy 38, 43, 141-143, 152, 164, 171-174
- nebulization (nebulize) 97, 156-157
- neck collars 42
- nematodes 103, 153, 158-159
- neoplasm (neoplasia) 139, 152, 162-163
- nest(s) 21, 23, 25, 29, 34, 45-46, 48-49, 51-52, 54, 56, 59-63, 68, 72-73, 76, 86, 106, 112-113, 115, 117, 125, 130-132, 136, 181, 193, 200, 212, 231, 245, 248-250, 261, 268-269, 271, 273, 278, 281, 283, 285, 287, 290
- nest building 34, 45, 86, 106, 113, 125, 131, 212
- nets 238, 241, 250, 259, 261
- Newcastle disease 156-157, 173

- nutrition (nutritional) 35-36, 43, 51, 54-55, 59, 72-73, 89, 91, 95-96, 98-99, 101, 104, 123, 132, 137, 139, 144, 152, 165-166, 168, 174, 229
nutritional status 139
nutritional support 98-99, 104, 144, 168, 174
- ocular injuries 161
omphalitis 96
orthopedic 100, 166
osteoarthritis (see arthritis) 165
osteomyelitis 103, 161-162, 166
ovary 126, 129-130, 207, 228
ovulation 52, 126-127, 129-130, 132-133, 207, 215
oyster shell 34, 51, 62
- packed cell volume 98
pair bond(s) 22, 45, 51, 59, 108, 112, 114-116
parasite(s) 31, 43, 61, 78, 84, 86, 92, 95, 98, 103-104, 133, 137, 151, 153, 158, 160, 171-173, 245, 248-249
parasitic diseases 43, 153, 158, 172, 252
parasiticides 158
parent-rearing (parent-reared) 33, 37, 77, 79-80, 83-84, 86-87, 89-91, 95, 97, 103, 110-111, 114, 116, 192-193, 202-203, 231, 233-234, 238, 240, 244, 258
parentage 175, 177, 181
pedigree 181, 185
pen rotation (see fallow pen) 31, 33, 39, 158, 251, 261
peritonitis 97, 103, 152, 158
pest 245-247, 249-252
pesticide(s) 151, 163, 173-174, 245-246, 252, 268, 277-279
pets 24
phosphorus 34-35, 51, 145, 150-152
photoperiod (see daylength) 33-34, 46-50, 56, 60-61, 111, 131-132, 134-135, 261-262
physical examination(s) 33, 137, 140, 155, 188, 192, 199, 236
physical restraint 36, 154
physical therapy 164-165
physiology 29, 56-57, 113, 123, 125, 127, 129, 131-136, 216-217
pipping 66, 69, 73, 200
plumage 1, 21, 29, 106-107, 119, 131, 134, 136, 139
pododermatitis (see bumblefoot) 140, 161, 165
pool(s) 32, 49, 51, 78, 87, 89-90, 146, 176, 178, 183, 219, 238, 256, 260, 262
Population and Habitat Viability Analysis (PHVA) 181, 274, 276, 279, 286, 291
population growth 176, 268, 279
predator 77, 84, 88, 237, 245, 247-249, 251, 259
preventative health program 95
preventative medicine 95
progesterones 130
prolactin 45, 125-127, 131, 133-135
prostaglandins 126, 129-130, 134-135
prosthesis 170-171, 173
protein 34-35, 51, 55, 78-79, 99, 101, 129, 134, 144-145, 150-152, 181, 216
protein electrophoresis 181
protozoa 153, 158
proximate factors 46
Pseudomonas 100, 104, 156
pubic bones 62, 190, 207-208, 212-213
public display 45, 105, 116
- quarantine 158, 171, 178, 260, 262
- radio transmitters 238
radiograph 138, 154-155, 162, 165, 171, 193, 244, 262
radiology 154, 193
rainfall 48, 51, 124, 268
records 25, 54, 57, 60, 65, 67, 78, 154, 185-204, 212-213, 272, 284, 289-290
red blood cell count 148
Red-crowned Crane 83, 105, 107, 120, 178-179, 233, 240, 258, 266, 285-286, 288, 290
reintroduction 27-28, 56, 75, 77, 118, 120, 133, 231, 233-235, 237, 239-240, 263, 267, 273, 275, 279, 286, 288-289
releases 117, 125, 127-129, 132, 231-232, 239, 273-274, 277, 288
releases, abrupt 21, 24, 39, 85, 94, 231-232
releases, gentle 98, 231-232, 234, 240, 244
renal disease 152-153
reproductive lifespan 51-52
respiratory disease 97
restraining 36-37, 39, 146, 164, 243-244

- Salmonella* 95, 155-156, 171, 174
Sandhill Crane 2-4, 21-24, 26, 29, 11, 33, 40, 43,
48-49, 51, 53, 56-57, 75-76, 81-83, 85-86, 92-93,
99, 101, 103-105, 107, 109-113, 118-122, 129,
134-135, 138-139, 150-151, 163-164, 168, 171-174,
181, 208-210, 216-217, 219, 221, 225, 231, 233,
238-240, 242, 245, 260, 265, 271, 275-277,
287, 291
sanitation 31, 65, 75, 87, 246, 248
Sarus Crane 2, 26, 83, 163, 172, 225, 265,
278-280, 289
scoliosis 138, 156
screen-rearing 93, 234
selenium 36, 102, 143, 163-164
semen 34, 45, 47, 49, 51-53, 55-57, 124-125, 128,
130, 132, 134, 181, 189-190, 199, 204-217,
219-222
semen, collection 61, 95, 105, 137, 144, 146, 155,
157, 185-186, 193, 199, 202, 205-207, 209-212,
215-217, 219
semen extender 209, 211-213, 217, 219-220, 222
semen, fecal contamination 207, 210
semen, protection 27, 37, 115, 209, 211, 237, 245,
250, 263, 267, 269-271, 273-274, 276-277, 279,
281-284, 286, 288-289, 291-292
semen, quality 52, 55-56, 83, 154-155, 164, 173, 190,
199, 209-210, 215
serology 97
serum chemistries 152
sex ratio 28, 56, 175-177
sexing 34, 110, 173, 181, 223, 226-229
sexing, blood culture 227
sexing, DNA probe 223, 227
sexing, feather pulp 227
sexing, fecal steroid 223, 228
sexing, karyotyping 223, 226
sexing, surgical 173, 227, 229
sexing, vent sexing 228
sexual maturity 51-52, 56, 117, 121, 123, 132, 177
shipping (shipment) 40-41, 158, 160-161, 199, 202,
227, 243
shock 98, 101, 137, 140-142, 153, 160-161, 249
Siberian Crane 2-4, 21, 25-29, 10, 39-40, 46, 52,
56-57, 83, 86, 98, 100, 108, 112, 118, 169,
178-180, 225, 235, 252, 264, 273-275, 291-292
sling 39-40, 164, 168
social display(s) 22, 106, 110-113, 116, 212
socialization 77-78, 91, 93, 95, 188, 233, 238, 257
sonagram 223
sperm 52, 128, 130, 135, 148, 205, 207-211,
213-217, 219-221
sperm concentration 1, 32, 148, 150, 153, 173, 207,
209-210, 213-214, 216, 219-220
sperm density 144, 154-155, 165, 210, 215, 220-221
sperm motility 75, 136, 210-212, 214, 219-221
splayed leg 102
splint(s) (splinting) 100-103, 165-169
sprinklers 48, 260
Staphylococcus 140, 152
Starlicide 250
stress 29, 31, 39-40, 45, 54, 77, 80, 86, 90, 97-98,
115, 123, 127, 132, 134-135, 139-140, 151-152,
154-155, 160, 162, 166, 206-209, 227, 234
stressors 132
subcutaneous emphysema 139, 161, 165, 170
sulphur amino acids 35
supplemental (supplementary) feeding 79, 156
supplemental heat 33-34
surrogate 45, 53, 59, 61, 85, 87, 119-120, 231, 240
surrogate incubation 61
surrogate pair 61
survival 1, 23, 27-28, 49, 56, 63, 95, 103, 113, 117,
141, 158, 168, 175-179, 181-183, 192, 196, 210,
219, 231-234, 237-238, 240, 244, 288, 291
suture(s) 96, 98, 100, 141, 162-163, 166, 168-169,
170-171
swimming (see aqua therapy, hydrotherapy) 78, 90,
103, 256
Syngamus (see gapeworm) 103, 153, 158

taming 84, 115
tattoos 42
taxonomy (taxonomic) 1, 27, 120-121, 182, 204, 228
tennis netting (see visual barriers) 37, 94, 115, 247,
257-260
tenotomy 162, 241-242, 244
tension taping 103
testes 128, 205, 227
toes 67, 83, 101-103, 106, 140, 165
toes, crooked 101-102, 165
toes, curled 101, 165, 168
total protein 145, 150-152
toxicity 141, 151-152, 164, 220, 252
toxins 151, 163
trachea 1, 21, 97, 99, 123, 138, 144, 156, 158, 170
tracheal flushes 156
tracheal washes 158
training 27-28, 82, 87-89, 95, 116, 118, 170, 209,
237, 245, 252, 267, 278, 282, 284-285, 288-292
transfer 40, 73, 180, 185, 199, 205, 219-221, 237,
256, 262

- translocated 231, 247
 transmitter(s) 237-240
 transponders 42-43
 transportation 164, 245
 transporting 36, 39-41, 73, 164
 trap(s) 247-252
 trauma 41, 66, 97, 100-101, 103, 137-143, 152,
 160-162, 164-165, 170, 259
 trematodes 158-159
 tube feeding 79, 89, 97, 99-100, 144, 168-169
 tuberculosis (TB) 151-152, 155, 162, 171, 174,
 207, 214
- ultimate factor 46
 umbilicus 60, 86-87, 96
 Unison-call 4, 108, 111, 223-225
- vaccination 33, 157, 171-172, 186
 vaccines 157
 venipuncture 141, 146
 viral infections 151
 virus 157, 172-174, 250, 252
 visual barriers (visual screening) (see tennis netting)
 31, 34, 45, 54, 115, 161, 247, 258-262
- wading pools 32
 Wattled Crane 2-3, 21, 23, 25-26, 28-29, 7, 66, 75,
 120-121, 145, 149, 179, 225, 264, 269-270,
 289-290
 weighing 39, 81-82, 93, 98-99, 139, 234, 253
 weight 34, 38, 51, 54, 60, 63, 66, 68, 71, 74-75, 78,
 81-86, 88, 95, 97-99, 101, 103, 111, 130, 139, 144,
 146, 155-156, 158, 164-166, 188, 192, 215,
 226, 241
 white blood cell count 148-149
 White-naped Crane 4, 46, 83, 179, 183, 204, 225,
 229, 265, 277-278, 289
 Whooping Crane 2-3, 21-22, 24-29, 19, 33, 38, 47,
 51, 56, 71, 74-75, 82-83, 85-86, 90-91, 99,
 103-108, 110-112, 120-121, 133, 135-136, 154-155,
 160, 163-164, 171-174, 181-182, 197, 204-206,
 211, 215, 223, 225-226, 228-229, 231, 233, 235,
 240-241, 252, 266, 286-290, 292
 wing clipping 33, 241-242
 wing problems 78, 82, 101
 wing tags 42, 85
 wounds 140, 150-151, 160, 168, 244
- yolk sac 60, 72-73, 82, 94, 96-97, 104
- zinc toxicosis 164

