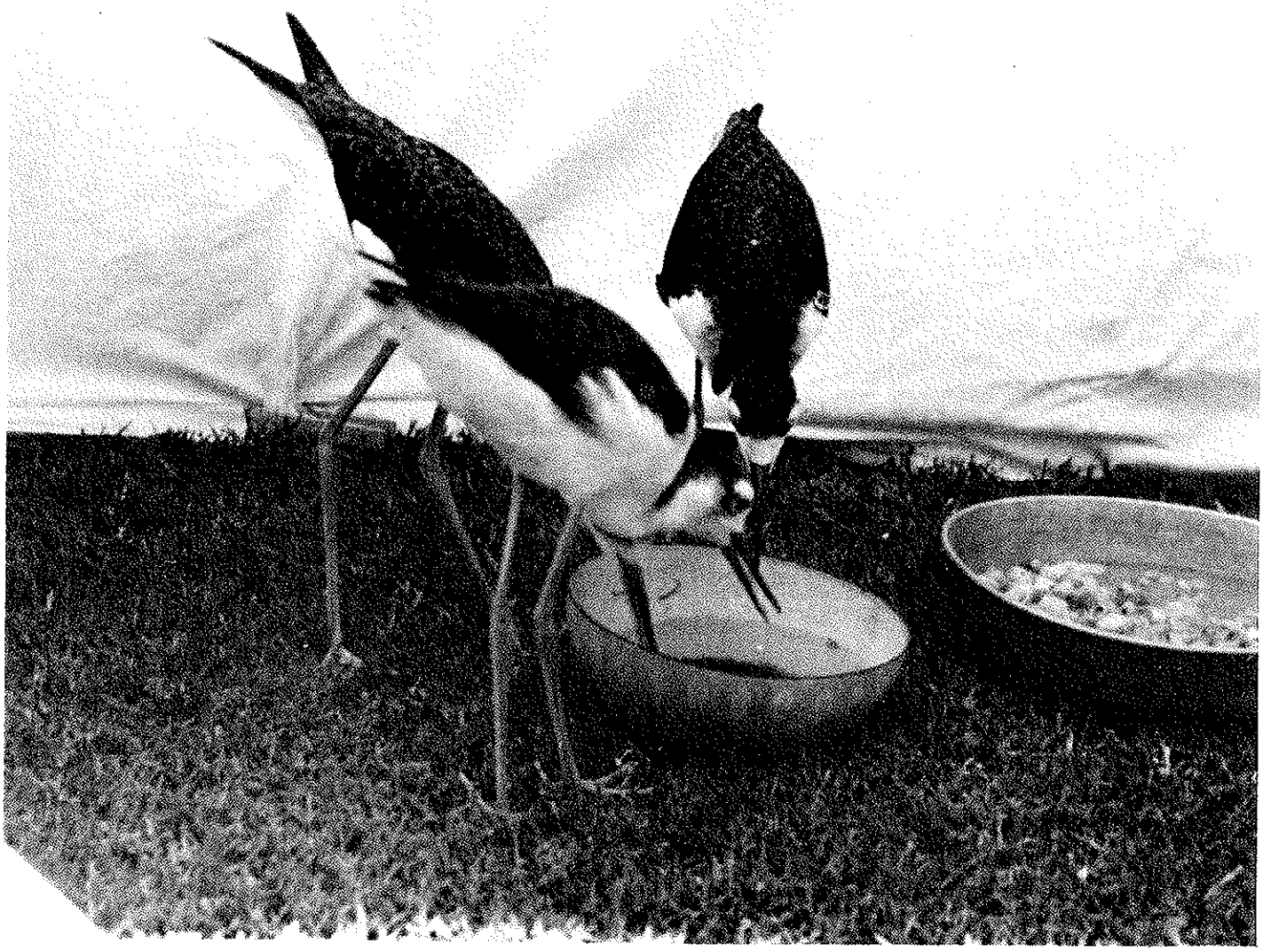
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Vol. 9, No. 1

WILDLIFE JOURNAL

Journal of the International Wildlife Rehabilitation Council



Stilt Rehabilitation . . . Page 7

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4. The writer's name and address and organization should be typed on the first page, and name and page number should appear on all consecutive pages.
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WILDLIFE JOURNAL

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EDITORIAL . . .

Executive Directors and Leadership

New wildlife organizations usually start off with inexperienced directors. Even established groups will occasionally select a relatively new director. These folks may have two or more years of rehabilitation experience, but management and supervision are entirely new to them. Additionally, due to the tight economics of the field, these people are frequently placed into leadership positions with little or no planned training in management and supervision. Herein lies the problem for volunteers and staff, because a person without leadership background will often feel threatened if his or her knowledge is challenged or questioned.

Having found myself in this position years ago, I did the only thing I could think of — found more experienced directors and consulted with them. I visited their facilities, called them often, and tried to avoid some of the mistakes they related to me. Then I went to work for the state and learned much about both being supervised and supervising others. Working for the Employment Development Department also gave me the opportunity to learn about employment issues involved with hiring, firing, workman's compensation, and unemployment insurance. I made plenty of mistakes, in spite of my best efforts, and I lost a few battles.

Then I learned that you don't always have to be right. In fact, people may think they want their director to always be right, but they are relieved to know you are human also. If you are wrong, admit it and then do the appropriate thing. No one respects a leader who has made a bad decision and will not admit it, never mind correct it. People begin to wonder whose interests you are serving, your own or the animal's.

Sharing leadership within the organization is good management. Let people know that you are implementing their ideas, and give them credit. Building a strong organization will generally require the placement of key responsibilities on many more people than the director. This will not mean loss of control. Although the amount of power increases with the amount of responsibility, levels of control can be securely built into the director's job description which is appropriate since the buck stops on the director's desk.

I have learned too, the difference between dealing with friends who are volunteers and employees who would like to be friends. As an employer, you cannot ask employees to work on the basis of friendship. The boundaries of the employer-employee relationship must be honored to avoid the sticky work-related issues that have nothing to do with friendship. There is just about no worse feeling than knowing you have to address employee non-performance with someone who expects you to understand and ignore the problem because you are friends.

Finally, when times are hard, it is so easy to vent your frustrations on those you work with but it is really not their job to act as a shock absorber. Such treatment will only increase their job-related stresses and may adversely affect their performance, thereby worsening the director's problems. You can network with other directors or find a neutral party (not your family either!) to share your woes. People in analogous positions in other industries may be good ones to share ideas and problems with. If you really get depressed, see a counselor. Just keep your staff relationships on a professional basis. They will appreciate it.

Learning to be a good director takes a great deal of effort and time. The main concept is that of caring for the people you work with, not just being the boss to the people who work for you. And it requires patience and understanding from them, in turn, as you develop your skills. Building a good team with the goal of making the organization the best it can be is possible if the goal is not allowed to be obscured. Although workloads will fluctuate with the season, with good management, your organization will be ready for the next busy season.

Jan White

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Pre-Anesthetic, Anesthetic and Post-Anesthetic Care for Raptors

Roark R. Freeman, D.V.M.
Crocker Animal Clinic, Santa Clara, California

The purpose of this paper is to provide the rehabilitator with some fundamental information required in the successful handling and management of the raptor requiring anesthesia. The focus of this article is the period of time beginning with the receipt of the avian anesthetic candidate through the anesthesia itself and immediate post-anesthesia care. Detailed information on long-term raptor care, housing and rehabilitation may be found elsewhere. Much of the information in this applies equally well to non-raptorial avian species.

HISTORY

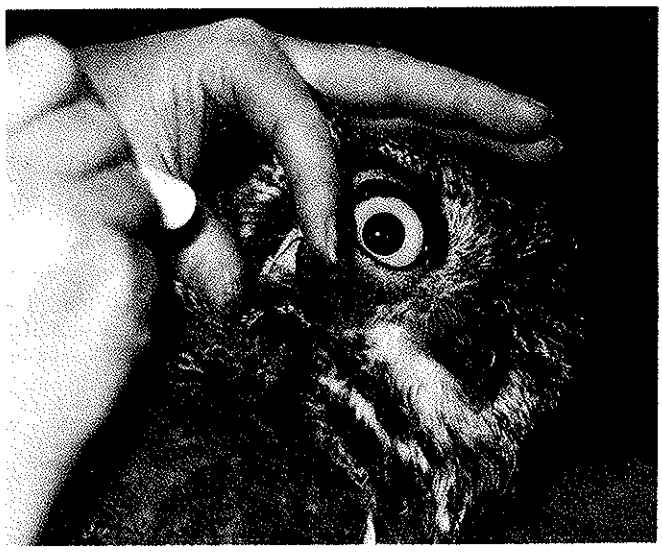
Wild bird medicine and surgery provides unique challenges to the veterinary practitioner and rehabilitator. While other patients are usually presented complete with detailed information concerning such biological data as age, sex, medical history, background, nutritional status and specifics relating to the nature and duration of the animal's illness or injury, raptorial patients are often presented with only rudimentary historical backrounds. Frequently (except for the most obvious cases), the only thing known about the bird is that it was too sick, injured or debilitated to elude capture. It is, therefore, as important to the rehabilitator as it is the the veterinarian to learn as much as possible about the history of the anesthetic candidate when the raptor is first presented. Although the condition that may require anesthesia (and presumably surgical correction) may be the most obvious problem in the patient presented, it must never be overlooked that many patients have *multiple* problems, some of which may influence the raptor's ability to withstand anesthesia, or ultimately, to successfully recover. Information about the bird including the location and details of its capture, and suspected nature and duration of the illness or injury that led to the capture can be invaluable in forming an early assessment and plan of action for the case. A history of the bird should include information about immediate care the bird may have received following capture.

PHYSICAL EXAM

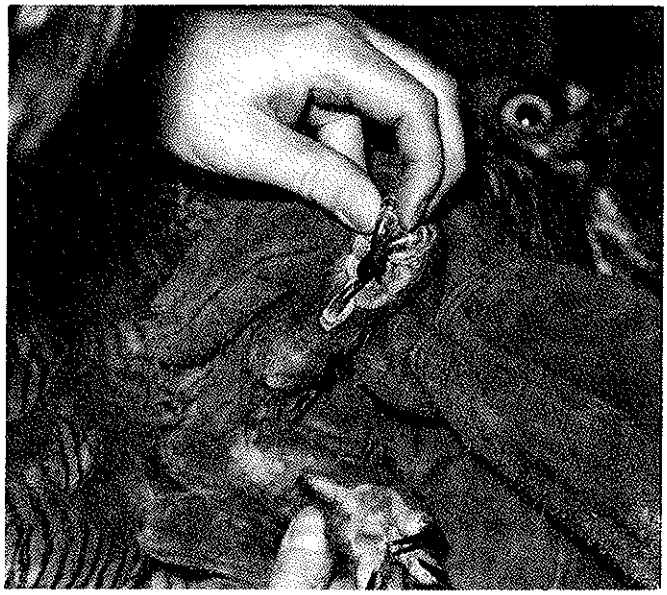
Once a complete history of the raptor is obtained, a thorough physical exam is in order. Extreme care must be taken with debilitated individuals not to compound their stress any more than absolutely necessary. The stress of handling may be more than these individuals can take. Most have been grievously stressed before they ever arrive in the hands of the rehabilitator. A good physical exam, however, can be performed rapidly and gently and is vital (along with a good history) in devising the treatment plan for the patient. Of course, any immediate life-threatening conditions such as hemorrhage or shock should be attended before completing a physical exam.

Before handling the raptor for a physical examination, observe the bird on its own. Observe its stance and how it holds its head. Is it alert? Ask yourself if the bird appears to have vision. Be aware of any abnormalities in body posture including placement of the feet and how the bird holds its wings in relation to the body. Listen for any abnormal respiratory sounds. Look at the droppings and see how dry they are and note their color and consistency.

To proceed further with a physical exam, restraint is necessary. Hooding the bird is valuable in many raptors and the feet should be immobilized. A complete exam of the head and mouth should not be overlooked. Look at the eyes. Check for any signs of injury, discharge, swelling or inflammation of the mucous membranes. Check pupillary function (i.e., are both pupils the same diameter and do both respond equally to light?). Are the eyes bright? Glassy? Do the eyelids appear wrinkled (this is a clue that the raptor is dehydrated). Look at the nostrils and note any swelling, crusts or discharge. Examine the external openings of the ears. Open the mouth and observe the color of the



Examining the ear.



Examining the feet.

mucous membranes and note whether they appear moist. Check the capillary blood flow and note how long it takes to return once pressure is released. This should take ½ to 1 second. Observe the opening to the glottis. Note any swelling, plaques or ulcerations visible in the throat or choanal slit. Part the feathers on the head and look for any bruising under the skin that may be present.

Examine the skin and plumage of the body. Look carefully for any injuries. Gunshot punctures may be very small and detectable only from tell-tale small hemorrhages or crusts. Look closely for evidence of skin and feather parasites. Assess the elasticity of the skin and tendency to "tent" when pinched. This is an indication of the hydration status of the bird.

Palpate the chest, assessing the pectoral musculature and prominence of the keel. This information will yield an indication of the nutritional status of the bird.

Examine the legs and feet, including the talons. Can the raptor grasp normally with the feet? Expect a reflex closure of the toes when the bottom of the foot is stimulated. Look at the skin of the foot and check to see if it "tents" when pinched. This is a sign of dehydration. Move all joints through their ranges of motion to detect any difficulties there.

Observe the vent. Is it clean and dry? Are surrounding feathers contaminated with fecal material?

Examine the wings. Observe the plumage. Note any damaged feathers, especially primary or secondary feathers. Damaged flight feathers must be repaired or replaced before the raptor can be returned to the wild. Move all wing joints through their ranges of motion and compare one to the other to help detect any abnormalities.

Auscult the chest with a stethoscope. Listen for harsh sounds including clicks, squeaks, or moist rattling sounds. Palpate the abdomen and note any swelling massses or ascites (fluid accumulation).

Weigh the bird. An accurate weight is an invaluable aid in the assessment of the raptor when it is admitted, as well as a tool for evaluation of progress in treatment and rehabilitation. Weight should be taken *daily* where possible for raptors in acute stages of illness or injury, especially for birds who are not maintaining themselves.

Sheath the tail of the raptor to protect the feathers from soiling and damage in the cages (see illustrations in Garcelon & Bogue, 1977).

ASSESSMENT, PROBLEM LIST AND TREATMENT PLAN

Once a thorough physical exam is completed, an assessment of the patient may be made and a preliminary treatment plan initiated. Make a problem list that includes all abnormalities gleaned from the history and physical exam. A treatment plan can be made so that all problems may be dealt with according to severity. It is at this time that treatments indicating the use of anesthesia are noted. Anesthesia-related procedures are often several notches down on the priority list of treatments with many raptor patients. There are few situations in raptor medicine and surgery that call for the emergency use of an anesthetic. Rather, more emergencies will occur *under* the influence of an anesthetic in raptor patients who are not up to their maximum possible fitness levels before subjecting them to anesthesia.

It takes only a few minutes after receiving a raptor for treatment to gather a history, perform a physical exam and tabulate a problem list and treatment plan. When these necessary steps have been completed, the true rehabilitator must ask the following questions:

(1) Are there injuries or illness(es) that are too severe to allow full recovery of the bird and return to the wild?

(2) If there are conditions that may allow recovery to health but not fitness, is there a *need* for the physically disadvantaged raptor for example, in captive breeding and fostering programs or wildlife collections?

(3) If the raptor is nonreleasable or not of potential value for various reasons, are we doing the raptor or ourselves any favors by treating it?

The answers to these questions are often difficult to obtain and not readily apparent, but the true rehabilitator must consider these points for every patient received.

Because raptor patients are commonly presented to the rehabilitator in terrible physical shape (shock, hypothermia, hypoglycemia and extreme states of emaciation are all too frequently encountered), it is often upon the rehabilitator's shoulders to make decisions that may eventually effect whether the raptor lives or dies. It is in this area of medicine alone that the nonmedical professional is often responsible for the primary and urgent care of critically ill patients. One of the earliest decisions that must be made by the rehabilitator upon receipt of the avian patient is exactly *when* the patient must see the veterinarian for care. Definitive care of the situation requiring anesthesia must often wait while the raptor receives more primary and lifesaving urgent care. Often a veterinarian is not readily

available to administer this primary care and the rehabilitator must make the decision that the risks of further transporting and stressing the critically ill patient may outweigh the benefits of the raptor receiving primary care from the veterinarian. Each situation is unique: the most important consideration is that the debilitated raptor receive the best possible and most expedient care.

URGENT CARE

Primary or urgent care begins with the treatment of the raptor's most life-threatening problems. Often, the most serious problem in the raptor is shock. Shock is a complicated physical state brought on by a great number of differing stimuli. It is a life-threatening condition that must be treated immediately. A raptor presented for treatment may be in shock due to trauma, blood loss, or a number of other physiological stressors including advanced emaciation, dehydration, hypothermia, sepsis or the stress of handling. A quiet, somnolent bird with pale mucous membranes and a slower than normal capillary refill time should be considered to be in shock. The primary physiologic abnormality in shock is decreased cardiac output and circulatory collapse. Correction of this condition is of paramount importance.

To prevent further and deepening cardiovascular collapse in shock, veterinarians often use corticosteroids. While the use of corticosteroids in some instances is controversial, high-dose corticosteroids have proven their value in treating shock. Glucocorticoids frequently act to increase the strength of the heart in the late stages of shock, help to guard against deterioration of body cells by stabilizing lysosomal membranes and preventing release of harmful enzymes into the cells as well as aiding damaged cells in the metabolism of glucose for energy. There are a number of different corticosteroids used to treat shock with their major differences being the speed of onset of action and longevity of action within the body. Beginning with corticosteroids most frequently used (because of their speed of action) typical dosages are: hydrocortisone sodium succinate 8-20 mg/kg intravenously (IV), prednisolone sodium succinate 11-25 mg/kg IV, and dexamethasone 1-4 mg/kg IV.

For raptors in shock, fluid therapy may be just as important as corticosteroids. While transfusions of whole blood or plasma may be essential in birds suffering acute blood loss, balanced fluids and electrolytes are invaluable in replacing fluid deficits, expanding blood volume (which enhances venous return to the heart and ultimately cardiac output) and speeding return of blood flow and nutrition to the body tissues. The intravenous route of fluid administration is preferred for birds in shock. Place an IV catheter if possible. Methods have been described by Simms (1983). For shock therapy as well as for replacement of fluid deficits, a balanced electrolyte solution is used (balanced to mimic the natural toxicity and electrolyte components of normal extracellular fluid (ECF). Using a balanced electrolyte solution such as Lactated or Acetated Ringer's, recommended infusion rates to combat shock can be as high as 50-100 ml/kg/20 minutes in severe states. Intramuscular injections of fluids (in multiple sites) combined with gavage (oral administration) of fluids at rates of 50 ml/kg/hour may be used where necessary in situations that prevent intravenous administrations. Further information on avian fluid therapy is detailed in a set of articles by Evans (1984).

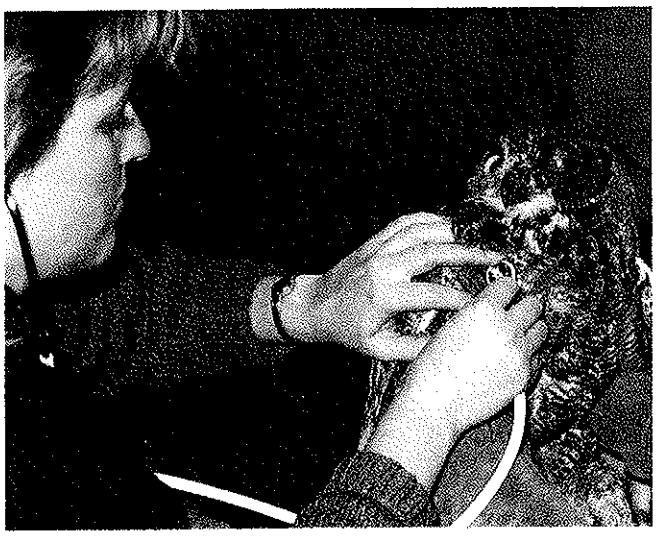
Antibiotics and their use in shock, like corticosteroids are controversial. Since shock is often accompanied (or in some cases caused) by sepsis, many veterinarians administer broad spectrum antibiotics to their patients under therapy for shock. There are numerous sources for dosages of antibiotics in avian patients including Emanuelson (1983). The author most often uses chloramphenicol succinate intravenously or intramuscularly at the dose of 0.08 mg/g BID or TID or amoxicillin 0.01 mg/g BID or TID with or without the addition of an aminoglycoside such as tobramycin 0.0035 mg/g BID, amikacin 0.01 mg/g SID of BID, or gentamicin 0.01 mg/g SID or BID to achieve maximum antimicrobial spectrum. It is important to be aware that antibiotics, like any drugs, require intact and functioning renal (kidney) and hepatic (liver) systems for their excretions and caution must be made in administration of drugs to any debilitated raptor.

PRIMARY CARE

FLUIDS AND FOOD

Many raptors presented to the rehabilitator or veterinarian for care are not in shock but require primary care (even if recovered from shock) before any anesthetic procedures are contemplated. For small raptors or birds subjected to extremes in environmental conditions, as little as 24 hours of food and water deprivation can lead to clinical signs from depletion of stored body reserves. Frequently, raptors are presented in extreme states of emaciation and dehydration from deprivation longer than 72 hours. For these birds who naturally have a very high metabolic rate, 72 hours or more of deprivation can bring them near death.

Before food- and water-deprived birds can receive definitive treatments for conditions that may require



Using a stethoscope to auscult the bird.

anesthesia, they must be stabilized. Open wounds should be treated, cleaned as well as possible, packed with antiseptic and wrapped. Limb injuries should be immobilized and wrapped to prevent further damage pending response to supportive care. For further information on limb immobilization see Welsh (1980).

Water-deprived (dehydrated) raptors must have their fluid deficits replaced before their gastrointestinal, cardiovascular and renal systems return to normal operation. It takes 12 to 24 hours to correct fluid deficits in severely deprived raptors. For replacement therapy, balanced electrolyte solutions must be used (i.e., those that mimic the extracellular fluid makeup of the patient). To estimate fluid requirements, the percentage of dehydration must be known. There are several ways to estimate percent dehydration in debilitated patients. One way is by observations gained in the initial physical exam. If the raptor's oral mucous membranes are dry, the eyes appear glassy or the skin of the eyelids is wrinkled, or if the skin tends to "tent" when pinched, it can be estimated that the bird is at least 5% (perhaps as much as 10%) dehydrated by body weight. The bird's initial weight divided by the percentage hydration estimated will yield the "normal" weight of the bird when rehydrated. The "normal" weight (in g) minus the dehydrated weight will yield the volume of fluids (in cc's) required to rehydrate the bird (Evans, 1984).

When available, a packed cell volume (PCV) and plasma protein (PP) can be a valuable aid in determining the degree of dehydration in raptors. All that is required is a microhematocrit centrifuge, small heparinized (anti-coagulant) hematocrit tubes and a few drops of blood, easily obtainable from a superficial vein or by clipping a toenail of the bird. By comparing the PCV and the PP of the patient to the normals for that species, dehydration levels may be evaluated, as well as response to therapy. In dehydration, the PCV and PP will be elevated (except in severe states of emaciation when they may be artificially low due to cachexia and mobilization of proteins to meet metabolic requirements for energy).

The exact quantitation of electrolytes in the extracellular fluid pool (ECF) (i.e., sodium, potassium, chloride, sodium bicarbonate) may be easily measured by any medical or veterinary laboratory so that accurate replacement therapy can be initiated. With smaller birds (those under 500 g) or in field situations where exact measurement of the ECF components is impractical, the best alternative is replacement fluids designed to mimic as closely as possible the normal ECF makeup of the patient. Recommended rehydration products include Ringer's solution, Acetated Ringer's, and Lactated Ringer's solutions. Through intravenous administration or oral gavage one-half the estimated fluid deficit may be replaced in 4-6 hours and the remainder in 12-24 hours. Maximum rates in gavage therapy are 50-100 mg/kg/hour (Evans, 1984). Maintenance solutions such as Pedialyte M, Kool-Aid and 1 tsp. salt/qt water and Gatorade are generally *not* suitable for *replacement* therapy since they do not approach normal electrolyte makeup of extracellular fluid.

The dehydrated raptor all too often is also the victim of emaciation. Emaciated raptors are *not* good anesthetic candidates. Smaller raptors (such as American Kestrels or Burrowing Owls) may suffer advanced states of wasting when food deprived for as little as 48-72 hours. Primary care for these animals is centered around correcting their caloric deprivations as well as fluid deprivations. Available body fat is utilized for energy in all but the largest food-deprived raptors in a very short period of time. This leads to catabolism of muscle tissues for energy and breakdown of glycogen (carbohydrate) stores in the liver as well. Hypoglycemia and hypocalcemia are common in food-deprived raptors and can lead to tremors and convulsive states. Those birds prone to convulsion from hypoglycemia (i.e., small falcons, and accipiters especially) should be supplemented as soon as possible with carbohydrate calories to prevent energy-wasting and life-threatening seizures. The seizuring bird *requires* veterinary care. Although emaciated raptors have a vital need for caloric supplementation, it is very important to carefully choose the type, amount and frequency of caloric supplementation so as not to overly tax the limited ability that debilitated raptors have to metabolize food. Digestion requires both free calories (emaciated raptors may not have calories to spare), and an intact and functioning gastrointestinal tract (severely emaciated raptors may take prolonged support before they can ever metabolize complex foods).

The emaciated bird should begin receiving calories in the form of simple sugars as soon as possible (often in the rehydrating solutions if administered by gavage). Birds who weigh less than 90% of their normal weight may require a succession of simple sugars foodstuffs in solution such as 5% dextrose and water, 10% or 20% dextrose, before receiving more complex carbohydrates, amino acids, proteins and fats. It may take days before these birds regain the ability to digest complicated foodstuffs or their regular diets. There are many methods and products available for rehabilitating the emaciated captive raptor. Experience is a good teacher; it would be wise for the rehabilitator to consult others in the field who have had success in bringing the emaciated raptor back to normal weight (White & Emanuelson, 1983).

HOUSING

The debilitated raptor receiving primary (pre-anesthetic) rehabilitative care should be housed in an environment providing the minimum of stress. It is unnatural for these patients to be confined at all and their stress is compounded by any housing that differs significantly from their natural environment.

Care should be taken to place these birds in enclosures that will not cause damage to their feathers or themselves, should they be startled. The best environments are those that are quiet and warm (to reduce the need to burn calories solely for maintenance of body temperature). Humidity levels should be maintained to avoid further desiccation in raptors being rehydrated. Appropriate perches, preferably natural ones or artificial perches covered with material providing large surface areas for maximum contact with the feet and distribution of weight, should be included in the enclosures.

continued on page 12

Rehabilitation Notes: Black-Necked Stilt: *(Himantropus mexicanus)*

Dorothy F. Johnson
Youth Science Institute, San Jose, CA

INTRODUCTION

The Black-necked Stilt is found in many climates throughout the world, from temperate, to tropical and rainforest, to desert conditions. The North American species *mexicanus* is found in western North America from Oregon east to Louisiana and Florida, and south to Mexico and Central America to Peru and Brazil.

Favored habitat includes brackish pools with extensive mudflats, flooded hay and rice fields, brackish areas of coastal lagoons, as well as salt ponds and lakes.

This attractive and vociferous shorebird was at one time considered a rare inhabitant of the San Francisco Bay area. (Sibley 1952).

During the past thirty years, populations of this stilt have begun to adapt to the salt pond regions of the San Francisco Bay area. It is now a regular breeder and winter resident.

The generic name *Himantopus* is derived from the Greek "himantopos" which means thong-foot or crook-shanked in reference to its long legs. The Black-necked Stilt has the longest legs in relation to body size of any bird except the Flamingo. Other common names include Daddy long legs, Long shanks, and Yelper (Terres 1980).

DESCRIPTION

Stilts have no hind toe, and a small web between the middle and outer toe. They can swim and dive, if necessary, but prefer not to.

Adults: The feathers are all white except for black wings, back of neck and crown. There is also a black stripe through the eyes. The black bill is slightly upturned and needle-nosed. The legs are pink and the eyes are crimson. Female plumage is similar with more brown on the back. There are two records of albinistic stilts in the United States (Herron 1980).

Juveniles: Have cinnamon fringes on the feathers of the upper back and the secondaries and inner primaries have whitish tips.

Chicks: Down feathers are white below, pale grey or buff above, and greyer on the neck. A thin black line runs down the fore-edge of the wings. A broken double line runs down the middle of the upper back, terminating in a dark spot on the lower back.

Molting: Young undergo a partial pre-nuptial molt in spring where they become indistinguishable from adults. A few juvenile wing coverts are retained. Adults undergo a partial pre-nuptial molt and a complete post-nuptial molt in late summer.

The longest lived bird in captivity was a specimen at the Bronx Zoo that lived for nineteen years. The oldest ringed bird in Europe: twelve years, two months

Measurements:

Weight: Male 142-179 g
Female 165-189 g
Legs: 8.5-10 inches long
Body: 13.5-15.5 inches long

Migration: Black-necked Stilts are migratory in northern California, Oregon, Nevada, and Colorado, down to New Mexico, Florida and the southern East coast. They are year round residents in the San Francisco Bay area and down the coast to Baja California and the east and west coasts of Mexico.

NATURAL DIET

The stilt feeds with a pecking motion, probing the underlying mud for crustaceans or taking prey from near the surface. Food items consist of 98% animal matter, although a few seeds are taken, as well as small pebbles to aid in digestion. Favorite prey includes brine flies and brine shrimp. They will also eat tadpoles, small fish, crustaceans, dragonflies, mayflies, earwigs, water beetles, and other types of flies. Young stilts eat a large quantity of beetle and fly larvae.



Susan Brain

Young Black-necked Stilt

NESTS AND YOUNG

Stilts nest in loose colonies. They are territorial and will aggresively protect their nest site. They will fly to meet intruders, calling rapidly, bobbing up and down, and striking the surface of the water with their chest to make splashing sounds. They will also pretend to be crippled, dragging one wing as though injured, and walking away from the nest in the hope of distracting intruders.

The breeding season for stilts ranges from mid-April through June in California (Stout 1980). Adults, two years and older, will usually breed (Cramp 1983). The nest may consist of a simple scrape or a more elaborate floating nest depending on habitat. Most nests are lined with vegetation, shell scraps, or twigs.

Stilts usually lay four eggs, occasionally three. Eggs are yellow to buffy in color with irregular splotches. They are subelliptical to oval and smooth, but only slightly glossy. The female lays an egg every 1 to 2 days until the clutch is complete. Incubation begins with the second or third egg, and lasts 24-26 days. Hatching is asynchronous (staggered) (Cramp 1983).

Both parents assist with nest building, incubation, and raising the chicks. Stilts are precocial, staying at the nest only long enough to dry off and master their long legs. Once this is accomplished, usually in 24 hours, they are off and running. The young will hide in the reeds or flatten themselves to the ground at the first sign of danger. The nestling period usually lasts 28-32 days. The young grow rapidly, and when not feeding, they will often rest with tarsi flat on the ground, until their bones are well formed (Bent 1962). First flights begin around 27 days. It is common for family groups to stay together long after the young become self-sufficient.

CASE HISTORY

On June 17, 1986, 18 Black-necked Stilt eggs were rescued from the San Jose-Santa Clara Water Pollution Plant (SJ/SC WPCP). The eggs were laid in a sludge lagoon (a large rectangular pit bordered by levees and used to store sludge until it dries.) Rainwater had collected on top of the 24 inches of sludge in this particular lagoon, providing a suitable environment for the growth of patches of bulrushes. The stilts used the bulrushes to construct their nests on top of the sludge. When it became necessary for the SJ/SC WPCP to bulldoze the sludge, the threatened eggs were removed by Kathy Hobson and Jean Young of the San Francisco Bay Bird Observatory at the request of SJ/SC WPCP personnel. Eggs from several clutches were rescued hence the large number of eggs and the varied hatching dates.

The eggs were transferred to a Marsh Farms Hov-a-bator type incubator at the Palo Alto Junior Museum. Humidity was kept at approximately 70% and temperature was kept at 101°F. The eggs were placed large end up on a screen and numbered for identification. An automatic egg rotator was used to turn the eggs ¼ turn four times a day, to equal one full rotation a day. The eggs needed to be removed from the rotator one or two days before hatching or the chicks could be injured by the rotator. As it was not known how long the eggs had been incubated before they were rescued, they were removed early on from the rotator. The eggs were then hand turned until they were ready to hatch.

One egg was damaged when rescued and, as expected, it failed to hatch. Of the 17 that hatched, one died from unknown causes and one was euthanized due to a leg deformity. The chicks were raised by personnel of the Palo Alto Junior Museum, the Youth Science Institute, and Wildlife Rescue, Inc. The first egg hatched on June 26th; the last eggs hatched on July 14.

HOUSING

Hatching To Two Weeks of Age:

Various brooders were used to house the chicks. The brooders ranged from large plastic laundry baskets wrapped in aluminum foil and warmed with a heat lamp to a more traditional Marsh Farms Home Hobby brooder box, and a Marsh Farms Brooder Top placed over a 10 or 15 gallon aquarium and covered with aluminum foil. All brooder variations were used successfully to house the young stilts.

The temperature was kept at 95°F for at least the first week. Some of the birds preferred the higher temperature into the second week. Bedding included newspaper covered with paper towels and mealworm bags covered with high quality paper towels. Uncovered newspaper is not recommended because the ink will soil the bird's feathers and impair their ability to repel water.

Two To Three Weeks Of Age

By the end of the second week, the stilts outgrew their initial housing because of the rapid development of their long legs. The birds were transferred into large cardboard boxes, wooden boxes or Vari-Kennels (plastic airline kennels) at this time. The average size cage used at this stage was 4 ft. long x 3 ft. tall. Brooder tops were placed on 1 foot high cinder blocks in the cage or hung from wood covers over the boxes. The ambient temperature was kept at approximately 80-85°F.

Three To Four Weeks Of Age

The chicks were still living in the housing described above. The birds raised in San Jose were completely off heat at three weeks of age. This is because average summer temperatures in San Jose are normally 10 - 15 degrees higher than the average temperatures of the cities in the northern parts of the county. Those raised further north were off heat during the day and on heat at night.

Stilts, although normally vocal, will huddle under the heat source, peep loudly and fluff out their feathers when they are cold. Contented stilts are very active, and inquisitive, moving all over the enclosure, but will occasionally rest on their tarsi. Use a combination of current weather conditions (i.e. high ambient temperatures) and the stilt's behavior to determine when to lower the temperature in the cage or discontinue using artificial heat.

Four Weeks To Release

At four weeks of age, most of the stilts were moved to outside enclosures. The average enclosure was 4 ft x 4 ft x 8 ft. The walls were made of ½ inch hardware cloth or welded wire. The flooring was sand, grass, or un-

printed newsprint. One enclosure was a 3 ft. high by 6 ft. wide circular wire animal pen that was placed on grass and covered with fiberglass paneling. Be sure to cover wire with sheets, cardboard, etc. to protect against feather damage and injuries. About this time, the stilts started making whirling "helicopter" flights.

The stilts at YSI shared an outside aviary with an American Avocet and an immature Green-Backed Heron with no difficulties. The heron spent most of the time on perches at the top of the aviary.

DIET

Hatching To Two Weeks Of Age:

The first food offered to newly hatched chicks should be live prey to induce feeding behavior. Tubifex worms were the first food items offered in this case. Brine shrimp can also be used. The stilts should start pecking at the live food almost immediately, and be eating well by the end of the first day. Ground Feline Science Maintenance Diet (dry kibble not canned) and Purina Trout Chow were also offered. Once the stilts began eating non-living food, the birds seemed to prefer the Feline Science Diet to Trout Chow. Purina Gamebird Startina (crumbles) can be offered in addition to the above mentioned foods. Water should be offered in a chick water dish (a narrow trough that is screwed onto a quart jar). Avitron vitamins (or other water soluble vitamins suitable for birds) can be added to the water, or SuperPreen (a powdered avian vitamin/mineral supplement) can be mixed with the dry food. It has been suggested that SuperPreen can be offered in the water as well. Food can be offered in jar lids or other small dishes.

When the chicks were one week old, mealworms were added to their diet. This proved to be a popular food with the stilts. A powdered calcium supplement and other supplements can be added to the mealworms, to improve their calcium/phosphorus ratio and nutritional value.

Remember that mealworms are high in phosphorus and low in calcium and that a diet that consists mainly of mealworms may cause severe health problems such as metabolic bone disease, poor feather growth, stunted development and death. For healthy growth and development, the chicks should be offered a wide variety of foods every day!

Two Weeks Of Age And Older:

Finely chopped baby smelt (white bait) was added to the diet at this time. This was not as popular a food, but would be consumed after all other choices were eaten. One group of chicks was fed waxworms as well.

Brine shrimp was introduced anywhere from the first week of age to one week before release. It seems that stilts took some time to become comfortable with the shrimp. as this is an important part of their natural diet, brine shrimp should be offered early on, so that the birds will become accustomed to eating them. At this time, young stilts will to show interest in chasing flies around their enclosure.

"Recreational" water can be offered at this time so that the birds can bathe. Pie tins make good starter wader pools. Make sure that the surface of the pie tin is not too slippery, as the chicks may slip and injure themselves. The bottom of the pan can be lined with sand. Food should be offered in narrow weighted dishes with slightly higer sides than the bathing dish, as stilts will step into anything that gets in their way.

This feeding regime can also be followed when the birds move into the outside aviary. But a larger diameter 1.5-2 inch deep wading pan can be placed in the aviary Be sure to place dry food away from the bathing area. In a couple of cases, the dry food was removed altogether because birds were tracking water into the pellets and transforming them into "cement patties."

RELEASE

Birds should be acclimatized, possess normal flight capability and be waterproof before release. Selection of the release site is important. Choose locations where there are other stilts such as mudflats, salt ponds, etc. Stilts should be released during morning hours and preferably on days with little wind.

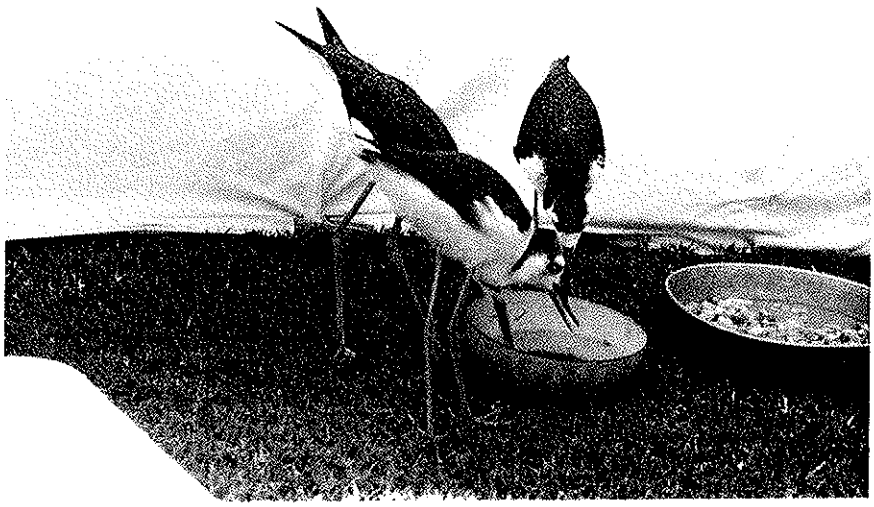
If the stilts were raised in relatively small enclosures, as was the case at YSI, an effort should be made to place the birds in a larger flight aviary for a few days prior to release.

Our stilts were released at six to nine weeks of age. Release weights ranged from 131 g to 167 g. The average weight at release was 149 g.

Salt Water Tolerance

If the stilts are to be released near salt water, they should be physically prepared to process the salt water they will be drinking upon release. At least three or four days before release begin introducing the birds to salt water. Use salt water in food and water dishes as follows:

On the first day use a solution that is approximately 1 percent salt. Mix it by adding 10 g of table salt (equal to a volume of approximately 10 ml) to a liter of water. If you can't weigh the salt or measure in metric measurements, you can approximate this strength solution by adding 1½ teas-



Frances Johnson

Stilts in outdoor housing.

poons of salt to one quart of water. The most accurate way to make the solution is to weigh the salt, the next best way is to measure it in mls and the least accurate way is to use teaspoons and quarts, but any of these methods will work.

On the second day, use a 2 percent salt solution. Mix it by adding 20 g of salt (approximately 20 ml) to one liter of water. Again, if necessary, you can mix one tablespoon of salt per quart of water.

On the third day, use a 3 percent salt solution. Add 30 g of salt (approximately 30 ml) to one liter of water or mix 1½ tablespoons of salt in one quart of water.

Seawater is approximately 3-3.5 percent salt, water in the San Francisco Bay is approximately 2.5-3 percent salt, and water in salt evaporation ponds is many times as salty as bay water.

As these birds dislike being handled, and their legs and beaks may be injured during tubing, we decided to put the salt solution in all dishes and pans that normally held water. When introducing birds to salt water, watch them closely for signs of dehydration, including lethargy, loss of appetite and convulsions. Should this occur, give no more salt solution that day. Tube fresh water until bird recovers and start slowly reintroducing them to salt the next day. None of the stilts that we raised had any problems adjusting to saltwater.

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1986 EAST COAST SUMMER GRADUATES OF THE BASIC WILDLIFE REHABILITATION 1AB CLASS

On August 23-24, 1986 IWRC conducted the Basic Wildlife Rehabilitation skills seminar in Bowie, Maryland. Hosted by the Chesapeake Wildlife Sanctuary, IWRC offered the class to eastern rehabilitators. All individuals listed below have received a passing score of 70% or better on the IWRC Basic Wildlife Care Exam and have completed homework on nutrition and calculating drug dosages. Additionally, all participants have successfully demonstrated adequate skills in wildlife care and handling in a supervised lab session. These individuals are certified in Basic Wildlife Rehabilitation.

Mike & Maryel Schein
Bowie, MD

Katherine Mierke
Fort Washington, MD

Sue Parker
Hyattsville, MD

Christine Montuori
Potomac, MD

Jan Westervelt
Takoma Park, MD

Donna Mezzullo
Crofton, MD

Christine Weickert
Bowie, MD

Schantz Basir
Owings, MD

Tina Rhea
Greenbelt, MD

Kirsten Bulhack
Rockville, MD

Judith Greene
Fort Washington, MD

Anne Hiss
Arlington, VA

Ginger Cotten
Cambridge, MD

Denise O'Donnell
Millersville, MD

Pam Getson
Vincentown, NJ

Sandra & Deborah Carr
Davidsonville, MD

Lenore Kohlman
Columbia, MD

George & Dee Brandon
Norfolk, VA

Carole Nicholson, DVM
Lorton, VA

Kathi Shea
Silver Spring, MD

Karen Weatherbee
Accokeek, MD

Wendy Simms Swiss
Wheaton, MD

Tasha Hyatt
Derwood, MD

Carol Lebowitz
Baltimore, MD

John Hadigan
Silver Spring, MD

Becky Murchie
Edgewater, MD

Sharon Becker
Haymarket, VA

Diana Conger
Bethesda, MD

Barbara (Bobbie) Stadler
Manokin, MD

Davis Vaughan
Bowie, MD

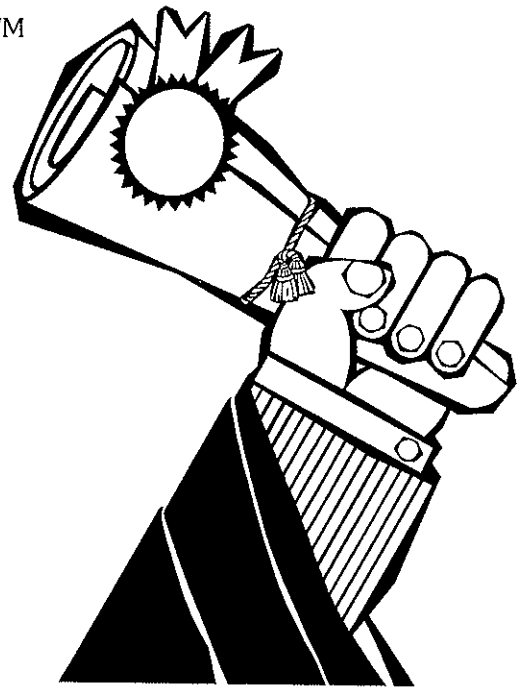
Kathryn Brown
Hampstead, MD

Virginia Rutter
Midland, VA

Fabienne McCormack
Prince Frederick, MD

Carol Embrey
Upper Marlboro, MD

Maria Lum
Palmyea, VA



Roger Hansen
Millersville, MD

Gerri Michael
Baltimore, MD

Marie C. L'Etoile
Sterling, VA

Howard Buck
Waldorf, MD

Jane Zuke
Adelphi, MD

Emerson Jackson
Port Deposit, MD

Sharon Sparks
Wheaton, MD

Jackie Freitag
Manassas, VA

Pat Brown Barbosa
Scotch Plains, NJ

Wendy Ernst
Silver Spring, MD

Barbara Williams
Seville, OH

Paula Bohashka
Solomons, MD

Paul Graninger
Hyatt, MD

Elizabeth C. Thompson
Bowie, MD

Stan Fetter
Fort Washington, MD

Billie Sue Durham
Chase, MD

Kelly Anne Campbell
Laurel, MD

Wanda Hopwood
Purcellville, VA

Hazel A. Spencer
Baltimore, MD

JoAnn Balliet
Tabernacle, NJ

Deborah Lieb
Washington, DC

Advanced classes are now scheduled for Maryland on April 4-5, 1987, They are: Clinical Pathology for the Wildlife 1987. Rehabilitator, Fluid Therapy, and Wound Management. For further information on registration, contact Diane Pearce at 17308 Queen Anne Bridge Rd., Bowie MD. 20716

Basic Wildlife Rehabilitation 1AB has additionally been scheduled for Long Island, New York on March 28-29, 1987. For further information on that class or to set up a class in your area contact Lisa Hoefler at P.O. Box 943, Carmel Valley, CA 93924.

continued from page 6

Attention should be paid to the photoperiod so that the rehabilitating raptor may not suffer the additional physiologic stressor of disruption of the day/night cycle.

PRE-ANESTHESIA

The best candidate for anesthesia is the patient in stable physical condition who is well hydrated, of normal or near-normal weight, and accustomed to its surroundings. Once the patient is stabilized, it is time to evaluate any abnormalities identified in the initial physical exam. The more that is known about the patient before the anesthesia is administered, the more likely the patient is to come through the anesthesia successfully. Basic examination of the stool for parasites should be performed routinely on debilitated raptors. Radiographs, preferably survey films of the whole body, can help identify other problem areas (gunshot wounds, other injuries old and new, respiratory disease, etc.) as well as defining the nature of the condition that requires treatment under anesthesia. A gram stain of smears from the choanal slit, cloaca, or wound site can be helpful in identifying major imbalance of gastrointestinal flora and aid in identification of infectious organisms. The gram stain is a simple test that can be performed by anyone with experience in basic microscopy. Gram stains can help the veterinarian determine appropriate antimicrobial therapy pending results of culture and sensitivities (which may require 72 hours or more to obtain). A complete blood count (CBC) can easily be performed with a few drops of blood. The CBC provides information of value, including the raptor's white blood cell count, red blood cell count, plasma proteins and may identify raptors who are infected with blood-borne parasites.

For birds considered anesthetic risks or when prolonged or strenuous anesthetic procedures are contemplated, blood chemistries should be performed prior to anesthesia. Modern laboratories can perform complete multi-channel blood chemistries on as little as 1 ml of serum. Basic chemistry profiles including tests for plasma proteins, kidney function, liver function, electrolytes and glucose (blood sugar) determinations can be performed on even less serum so that routine blood screenings can be possible with the smallest raptor patients. Blood clotting times are often performed especially when procedures that risk blood loss are planned.

If facilities or economics preclude blood chemistry evaluations or if more routine anesthetic procedures are planned for healthier birds, minimum blood tests should include PCV, plasma protein and blood glucose determinations. These basic and easily performed tests can be done with only a few drops of blood obtained from clipping a nail or cannulating a superficial vein such as the ulnar (wing) vein. These three tests speak volumes about the state of health of the bird. Comparisons of values from the patient to established normals for the species provide information the veterinarian may use for the anesthesia protocol. Raptors with resting blood sugar levels below 200 mg/dl are candidates for carbohydrate supplementation before and during anesthesia to avoid the dangerous situations that may raise in the hypoglycemic state (extreme depression, prolongation of anesthesia or convulsions). Birds that have decreased packed cell volumes or plasma proteins may not be as healthy as they appear. Decreased PCV and PP values from blood loss or malnutrition may also effect the raptor's ability to withstand anesthesia. Transfusions are often required to provide better odds for success in the anesthetic procedure and post-anesthetic recovery. Non-homologous species transfusions are often performed in avian medicine with good success so that the anemic raptor may still receive blood even without an ideal matched donor.

ANESTHESIA

The rehabilitator is often called upon to assist the veterinarian during anesthesia procedures on wildlife patients. Often (especially with captive raptor patients) the rehabilitator can provide invaluable information not only about the immediate medical history of the patient, but about the raptor itself and the natural history of the species. This information can be of special significance in the pre-anesthetic, anesthetic and post-anesthetic handling of the bird. The rehabilitator versed in various anesthesia types and their administration as well as proper anesthesia monitoring techniques will be able to provide the greatest assistance during the procedure.

Except for procedures calling for surgical manipulation of the gastrointestional tract, prolonged fasting is not usually done with raptorial patients. Smaller raptors may not be able to withstand fasts of 12 hours or more without becoming hypoglycemic. These birds are often fed shortly before the anesthetic procedure and supplemented with additional carbohydrates during and after anesthesia until they have recovered fully. Fasted raptors (especially smaller ones with high metabolic rates) may suffer from depleted glycogen stores in their livers. Low glycogen levels may deprive the anesthetized rap-



Checking the injured left eye.

tor of the ability to metabolize anesthetics and prolong recovery times. Many raptors have a tendency to regurgitate under anesthesia and must be watched very carefully for this occurence to avoid aspiration of crop or stomach contents.

There are a number of anesthetic regimens that have been successfully used in the treatment of raptors. Often the choice of anesthesia rests with the one(s) the veterinarian feels more comfortable with. The best anesthetics are the ones that provide the widest margin of safety, fastest recovery times, and best analgesic capabilities.

To avoid complications from excess secretions in the respiratory tract during anesthesia, many veterinarians will administer atropine at the dose of 0.04-0.1 mg/kg by injection prior to anesthesia. There is some controversy regarding efficacy of atropine in raptorial patients.

The two main routes of anesthesia used in raptors are injectable (parenteral) general anesthetics and inhalation anesthetics. In the United States, the chief parenteral anesthetic is ketamine. Safest when used intravenously in low doses for short anesthetic procedures, ketamine alone or in combination with other drugs can provide anesthesia sufficent for shorter procedures and allow restraint of the patient for endotracheal intubation and maintenance with inhalation anesthetics during longer anesthetic procedures.

Ketamine hydrochloride, when administered at the rate of 30 mg/kg intravenously or intramuscularly, is sufficient to induce and maintain anesthesia for short periods of time in most raptors. Owls may be somewhat more susceptible (as are nocturnal raptors) and may require lower initial dosages (Haigh, 1980). Ketamine hydrochloride alone has relatively poor analgesic qualities and can adversely affect the bird's ability to thermoregulate and (especially in higher intramuscular doses) may have the disadvantage of prolonged recovery periods, and possibly induction of cardiac electrical abnormalities and violent recoveries. Ketamine is often combined with xylazine hydrochloride at the dosage of 0.5-1.0 mg/kg to provide increased analgesia and smoother recovery. Another drug often mixed with ketamine to speed induction, aid in muscle relaxation and smooth recovery is diazepam (at the dose of 1.0-1.5 mg/kg). For induction of anesthesia and endotracheal intubation prior to maintenance with inhalation anesthetics ketamine is safest used in low doses (2.5-5.0 mg/kg) in combination with xylazine (0.2-0.5 mg/kg). Lower dosages results in the fewest cardiovascular disturbances and the speediest recoveries. All parenteral anesthetics require intact renal and hepatic systems and should be used with caution in raptors who are compromised.

Inhalation anesthesias are generally felt to be the safest anesthesias in all avian patients. They have the advantages of providing the ability to alter anesthetic doses (and therefore blood concentrations) continuously during anesthesia, as well as providing good analgesia and speedy recoveries. Some (especially metofane) have the fewest cardiovascular effects of all anesthetics and require, in general, less energy to metabolize and remove from the system since much of



It is equally important to check the "normal" looking right eye.

the volatile anesthetic leaves through the respiratory tract in expired air. Induction of anesthesia through a face mask is usually easily achieved with inhalation anesthesias. Induction of anesthesia in excitable, debilitated birds with inhalation drugs may have a greater risk than the smoother, quicker inductions with low-dose parental anesthetics, however.

In the United States, the two most commonly used inhalation anesthetics are metofane and halothane. A third, newer, inhalation anesthetic, isoflurane, shows promise as well due to its speedy uptake and excretion through respiratory membranes and residual anesthetic effects. This paper will focus on the use of the first two inhalation agents. Halothane, the more volatile of the two anesthetics, has the advantage of faster absorption time and excretion rates in the avian system (and therefore most rapid induction and recovery). The stage (depth) of anesthesia can be adjusted quickly with halothane, thus providing an extra margin of safety with this drug. Halothane has the disadvantage of requiring a special vaporizer to control its concentration at the point of administration and it tends to depress body temperature so that thermal support becomes critical during and after anesthesia. Halothane has potentially more cardiovascular effects than metofane and is more likely to lead to bradycardias (lowering of the heart rate) and rhythm disturbances. Induction of anesthesia with halothane in raptors is achieved with vaporizer concentrations of 2.5-3.5% (in inspired gas) and maintenance can be achieved with concentrations of 0.5-1.5%. Metofane's advantages over halothane are that it does not strictly require a vaporizer for induction or maintenance (although it is best that one is used). Induction and recovery are slower with metofane but it has fewer of the cardiovascular effects that halothane has. A greater degree of muscle relaxation and analgesia is obtained with

metofane as well. Induction rates with metofane are 3.5-4% and maintenance levels are achieved with concentrations of 1.5-2.0%.

Any anesthetized avian patient should have an endotracheal tube placed as soon as induction of anesthesia has been achieved. Standard cuffed endotracheal tubes (those that can be inflated at their tips after insertion to provide a good seal against leakage of air or fluids in the trachea) can be used on any but the smallest raptors. Smaller raptors can still be intubated with appropriately sized catheters that are fitted with adaptors to the anesthesia machine. Common anesthetic emergencies such as apnea (cessation of breathing) and regurgitation (that might otherwise lead to aspiration) can be best handled in birds who are intubated. Endotracheal tubes must be watched carefully for obstruction with respiratory secretions, however.

Often used in conjunction with inhalation anesthetics is another anesthetic gas, nitrous oxide. Nitrous oxide has minimal anesthetic properties on its own, but when used with halothane or metofane anesthesia may be induced and maintained with less cardiopulmonary depression and with greater analgesia. Nitrous oxide may be mixed with oxygen and delivered at a 50:50 percentage rate. Anesthetized raptors require a few minutes of pure oxyen after anesthesia is completed to allow the nitrous oxide to leave the bloodstream.

The respiratory system of avian species is unlike that of mammalian species. Understanding the differences is important when administering anesthesia through inhalation routes. Avian species are provided with air sacs in their thoracic and abdominal cavities. These air sacs receive inspired air and serve as reservoirs of oxygen- (or anesthetic-) laden air. In flight they may provide necessary bouyancy and they appear to serve an important role in evaporative cooling (this can be deleterious in anesthetized patients). Avian total respiratory capacity (air sacs included) may be three times greater than in mammals, although their lung volumes are smaller. Air movement in avians is primarily via bellows action of the ribs, sternum, coracoids and furcula since birds have no diphragm. Movement of these anatomic structures should not be hindered during anesthesia. Most oxygen (and anesthesia) exchange occurs during *expiration* in avian patients and any volume of air received by the trachea requires two full cycles of respiration before it leaves the system again. The avian vascular system within the lungs is far more effective in absorbing even minute quantities of gases than the mammalian system. Their highly efficient gas exchange system allows avians to fly at high altitudes where oxygen tension is quite low but it makes monitoring and administration of volatile inhalation anesthetics challenging and accounts for the avian patient's dramatic response to even small adjustments in anesthetic concentrations.

Monitoring of anesthesia in raptorial birds is essential to the success of any anesthetic procedure. There are generally recognized planes of anesthesia in avian patients (light, surgical (or medium), and deep). Assessment of the plane of anesthetic a patient is in can be difficult at any given time and the bird may seemingly undergo dramatic changes in anesthetic depth within seconds due to physiologic alterations and response to surgical manipulations. The clinical parameters used in measuring anesthetic depth in avian patients are palpebral, corneal, pedal, or cloacal reflexes, heart rate and rhythm and respiratory rates and depth. Under *light* anesthesia the bird responds to touch to the eyelids by fluttering them and responds to touch to the cornea by prolapsing the nictitating membrane (third eyelid). The bird does not respond to audible stimuli; there is no voluntary movement but the bird will respond to light touches to the bottom of the foot or pinches to the skin surrounding the cloaca. Respirations may be fairly rapid but regular. The avian in *surgical* (or medium) planes of anesthesia does not respond to touch of the eyelids (palpebral reflex) and the corneal and pedal or cloacal reflexes are slowed. Respiration is slow, deep and regular and the heart rate slows. Under *deep* planes of anesthesia the avian patient does not respond to stimulation of either the eyelid, cornea, foot or cloacal skin and respiration becomes slow and shallow while the heart rate slows even further. Anesthetic levels where all reflexes are lost are generally considered too deep for safety.

Temperature may drop precipitously during anesthesia in raptorial patients (especially smaller ones). Their normal 104-108°F temperature range may be monitored during anesthesia so that departures significantly below normal may be attended.

Heart rate may be monitored through a stethoscope or an oscilloscope. Electronic monitoring allows observation of both heart rate and electrical rhythm during anesthesia.

Respiratory rate can be monitored visually (especially if clear surgical draping material is used) but also may be monitored with equipment that broadcasts an audible tone with each respiration. Care should be taken to insure that respiratory secretions do not clog the endotracheal tube during anesthesia.

Due to the relatively large capacity of the avian respiratory system and significant evaporation that may occur during anesthesia across respiratory membranes as well as the fact that there is gradual diminishment of cardiovascular function from hypovolemia and lowered blood pressure, avian patients frequently require fluid therapy during or after anesthesia. Many veterinarians will administer 3-5% of total body weight in fluids before anesthetic procedures or supplement with fluids intravenously or in multiple small intramuscular injections during anesthesia. Smaller raptors or hypoglycemic patients may receive 2½% dextrose mixed with half-strength Lactated Ringer's solution intramuscularly or subcutaneously or 5% dextrose (or stronger) solutions in full-strength Lactated Ringer's solution intravenously to aid in the maintenance of adequate blood sugar concentration.

Anesthetic emergencies encountered most often in avian anesthesia are apnea, respiratory arrest and cardiac arrest. Apnea (brief cessation of breathing) occurs most often during induction of anesthesia and recovery. Good oxygen flow and ventilation of the avian patient's respiratory tract generally leads to resumption of normal breathing. Respiratory arrest (where no respiration occurs for 5-10 seconds or more) requires immediate action including cessation of anesthesia delivery, artificial support of respiration (this is where endotracheal intubation is essential) and administration of drugs such

as doxapram (0.01 mg/g) to stimulate respiration. Cardiac arrest may occur with respiratory arrest and is treated by cessation of anesthesia, administration of 100% oxygen, external thoracic massage and injection of cardiac stimulants. Prognosis is generaly poor for any bird in cardiac arrest.

POST-ANESTHESIA

Raptorial patients usually recover quickly from low-dose parenteral anesthetics and inhalation anesthetics. In the case of inhalation anesthetics, cessation of delivery of anesthesia and increases in oxygen concentration result in recovery times in as little as five to ten minutes. Some birds will flutter or react violently during the recovery period and should be restrained to avoid injury to themselves. The best method is to wrap the bird in a loose tube made from a towel so that the wings are restrained without interfering with movement of the chest and respiration. Both ends of the tube are left open and the bird is placed in sternal recumbency until it can extricate itself from the confines of the towel. The bird should be placed in a quiet, darkened, warm environment. Slow recovering birds often are hypothermic and require greater degrees of supportive heat therapy. Soon after the bird has regained its perching ability it may eat. Final rehabilitative care and support begins soon after anesthesia with the goal of early return to function and eventual restoration to the natural state.

Glossary

Apnea:	cessation of breathing.
Ascites:	effusion and accumulation of serous fluid in the abdominal cavity.
Auscult:	to examine by listening, usually to the sounds of the thoracic or abdominal viscera, with or without a stethoscope.
B.I.D.:	Latin abbreviation for twice a day.
Cachexia:	a profound and marked state of constitutional disorder; general ill health and malnutrition.
Cannulating:	inserting a cannula (a tube) into a duct or cavity.
Catabolism:	any destructive process by which complex substances are converted by living cells into more simple compounds.
Choanal slit:	the slit at the top of a bird's mouth; choana is the technical name for the two internal nares which open from the nasal chambers into the roof of the pharynx inside a bird's mouth.
Endotracheal intubation:	insertion of a tube through the trachea to assure a clear airway and prevent entrance of foreign material into the tracheobronchial tree.
Hemorrhage:	the escape of blood from the vessels, bleeding.
Hypocalcemia:	reduction of the blood calcium below normal.
Hypoglycemia:	an abnormally diminished content of glucose in the blood.
Hypovolemia:	abnormally decreased volume of circulating fluid (plasma) in the body.
Sepsis:	the presence in the blood or other tissues of pathogenic microorganisms or their toxins; the condition associated with such presence.
S.I.D.:	Latin abbreviation for once a day.
Sternal recumbency:	lying on keel or sternum.
T.I.D.:	Latin abbreviation for three times a day.

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Photos courtesy of Jan Wild.

MEALWORMS: Their Care and Feeding

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Mealworms are an important, readily available food item in many rehabilitation organizations. They are often the main live food source for several avian species. It is not uncommon for a rehab group with a large number of birds going through its doors to use 30 to 50 thousand worms a week. This becomes expensive, so the need to maintain the worms in a proper environment to reduce losses is important. The following method of care and feeding has been used by the author for several years. It has been very successful in keeping die-offs to a minimum and in keeping this food source nutritional and palatable to the birds.

Mealworms that are shipped through the mail are usually packaged in newsprint. Although the worms can be used immediately upon receipt, it is recommended that the worms replace lost fluids and eat before being used as food. To do this, remove worms from their packing and place them in a deep plastic dish pan, 15" x 12" x 6" (L x W x H), filled with two inches of one part pulverized dry cat or dog chow (a nutritionally complete food such as Science Diet) and three parts wheat bran. A mixture of 1:1 is also satisfactory. The medium provides the mealworms with a balanced food source. Gently mix the worms into the medium. Add thinly sliced apples, potatoes, or over-ripe bananas sliced in half lengthwise (Fig. 1) to provide a source of fluids. Place a cover made of fiberglass screening and elastic waist band on top of the container to prevent escape. Place in a cool, dry area. This container size will hold up to 5,000 worms.



Fig. 1. Container set up for holding worms.

A container measuring 36" x 24" x 12" (L x W x H) will hold 50,000 worms. Leaving a three to four inch space between the top of the dish pan and the top of the medium usually keeps the worms from escaping.

If the worms come to you packed in wheat bran and it is clean enough to keep the worms in, just add some of the pulverized dry food and sliced fruit. If the bran is dirty with fecal material, (feces looks like slate-gray sand), separate the worms from the dirty bran using a colander, than add to fresh medium as described. It is not necessary to refrigerate mealworms, but it will keep them dormant. If they are refrigerated, keep the temperature at 40° - 55° F. Lower temperatures can kill the worm.

Once the housing is set up, the mealworms must be kept clean. This is important to keep the food source for your birds as healthy as possible. As the feces accumulate, the medium will become foul smelling and should be changed at least once every five days depending on size of container and number of mealworms it is holding. To clean, pour a small amount of worms and medium into a colander and separate worms from debris. Place the mealworms in a freshly prepared container and continue until all the worms have been transferred. If you use the mealworms before the medium is dirtied, it may be re-used. Well maintained worms are plump, active, and do not have a foul odor.

Before using, the mealworms should be room temperature. Sprinkle ¼ teaspoon of Super Preen or Avia, (avian vitamin/mineral supplements) on one pint of worms and stir. This will give a light coating of the supplement on the worms. Be careful, too much powder can kill them. Do not prepare more than you can use in one day.

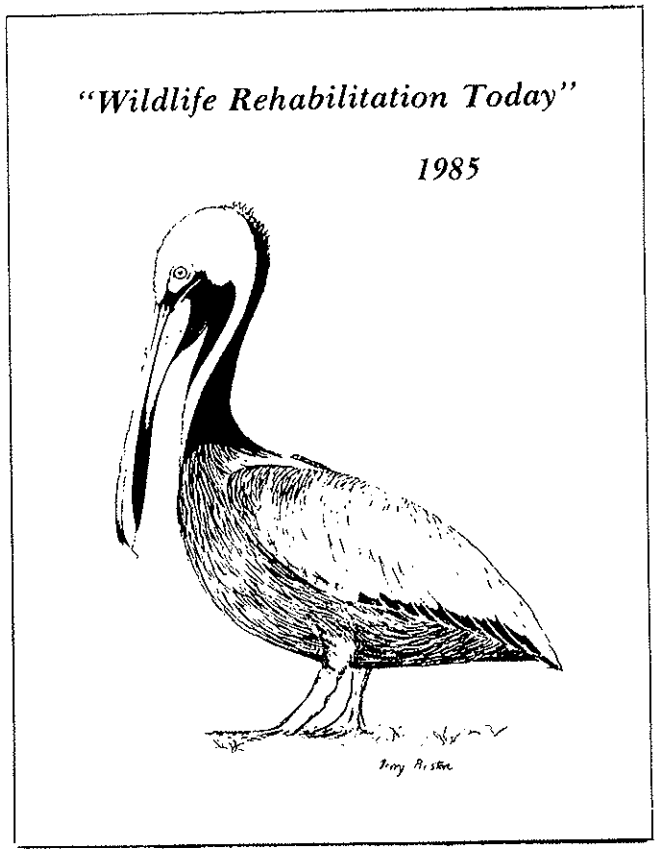
If you purchase large quantities of bran, it is advisable to store it in a well-sealed container to prevent bugs and moths from infesting the bran. To be sure you are not introducing any insects to your home that may be in the bran, spread several scoops of bran in a shallow baking dish and bake at 150° F for one hour. When the bran is *completely* cooled, it can be used in the medium or stored in a tightly sealed container. Do not store while still warm because sweating will occur and the bran will become moldy.

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