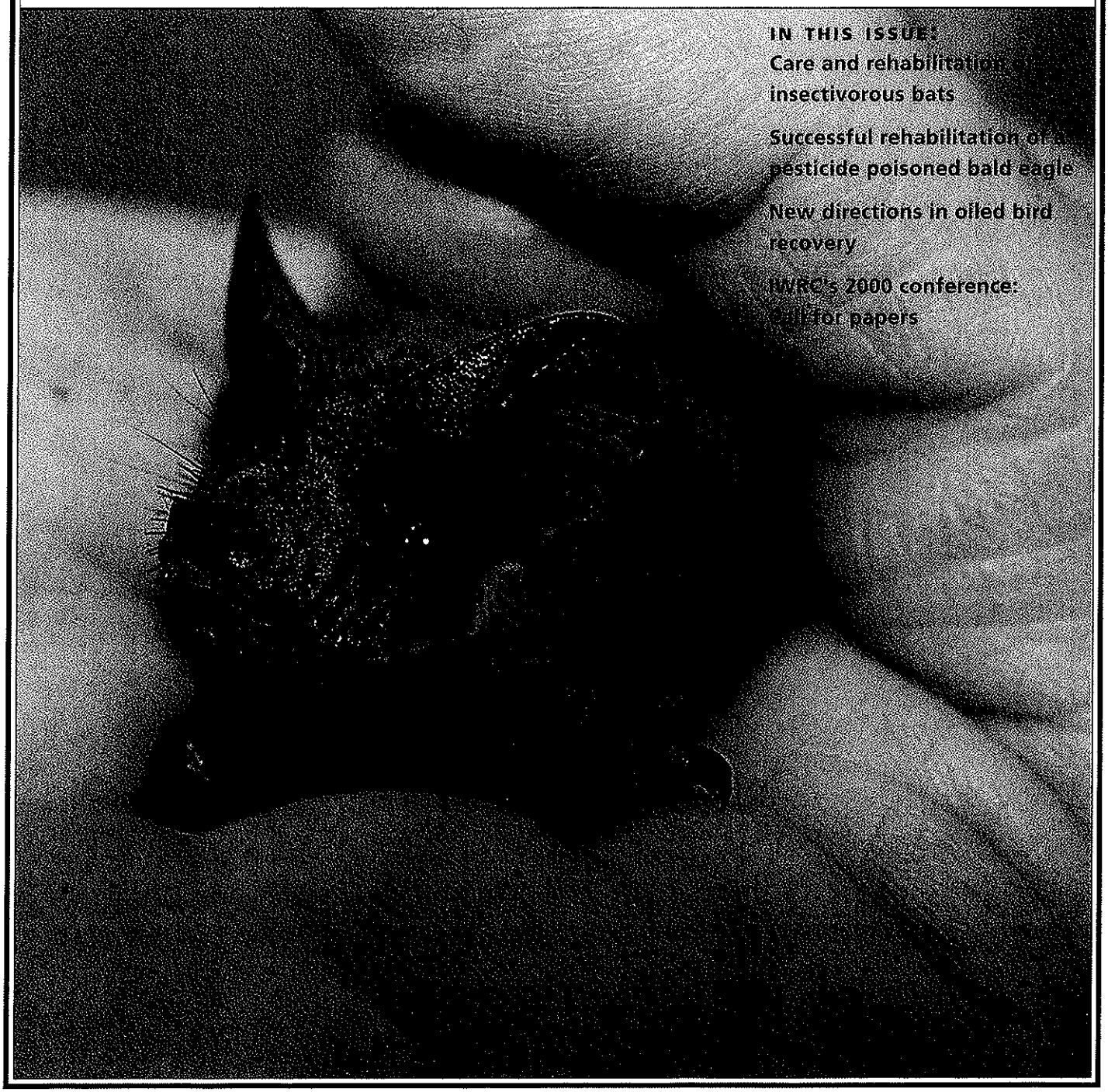
INTERNATIONAL WILDLIFE
REHABILITATION COUNCIL

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JOURNAL OF WILDLIFE REHABILITATION



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Care and rehabilitation of insectivorous bats

Successful rehabilitation of a pesticide poisoned bald eagle

New directions in oiled bird recovery

IWRC's 2000 conference: Call for papers

ABOUT THE JOURNAL . . .

THE JOURNAL OF WILDLIFE REHABILITATION is designed to provide useful information to wildlife rehabilitators and others involved in the care and treatment of native wild species, with the ultimate purpose of returning them to the wild. The *Journal* is published by the International Wildlife Rehabilitation Council (IWRC), which invites your comments on this edition. Through this quarterly publication, rehabilitation courses offered in numerous locations, and an annual conference, IWRC works to disseminate information and improve the quality of care provided to wildlife.

ON THE COVER: CAPTIVE CARE FOR THE REHABILITATION OF INSECTIVOROUS BATS. A young Mexican free-tailed bat (*Tadarida brasiliensis*), a common crevice-dwelling bat. [Photo: Amanda Lollar/Bat World] BELOW: A bald eagle is equipped with a satellite transmitter at the Raptor Center at the University of Minnesota's School of Veterinary Medicine. The transmitter allows tracking of this rehabilitated eagle for three years. [Photo: The Raptor Center]



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WILDLIFE REHABILITATION

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EDITORIAL

Put It Down in Writing

Working as a wildlife rehabilitator is certainly more than a fulltime job—whether you are paid or volunteer. But even as you care for the animals in your facility you know that there are many others in your region, your state, your country, and throughout the world who are in need or in danger.

Many of us have neither the temperament or the time to become fulltime activists. But how about becoming an *armchair* activist? Each year hundreds of wildlife-related bills are introduced by governments throughout the world. There are also campaigns to aid or protect at-risk animal populations, and others that seek to halt practices harmful to animals. By writing the appropriate agency you can play a small but important role in helping wildlife.

You can learn about many of these campaigns—and particularly those in the United States—through a free electronic newsletter published by the Humane Society of the United States and the Fund For Animals. To subscribe to HUMANElines, visit www.hsus.org and click on "Get Involved." Recent issues have discussed ...

- A proposal to allow the hunting and trapping of bobcats in Pennsylvania. Bobcats have been protected in Pennsylvania for more than 30 years.
- A proposed wolf "management" plan in Minnesota that calls for public hunting and trapping of wolves, discretionary wolf killing by landowners, and wolf bounties (with no provisions for public education, habitat protection, or nonlethal methods for discouraging wolf depredation).
- A campaign by a coalition of animal protection and environmental organizations to compel federal and Florida agencies to enforce manatee protection laws.
- News of the Bear Protection Act, S 1109, which would create a single, uniform, national policy prohibiting the import, export, and interstate commerce in bear viscera. (The poaching and trade in bear parts, spurred by their use in traditional Asian medicines, now threatens the existence of all bear species, worldwide.)
- A campaign to stop Montana from allowing the killing of Yellowstone bison who wander outside park boundaries.
- Support for a proposal by Kenya and India to protect elephants from commercial trade.
- Word of a USDA proposal for a massive blackbird poisoning program in North and South Dakota intended to reduce sunflower damage during the birds' southward fall migration. The proposed poison, DRC-1339, provides a slow, inhumane death.

After a brief description of the bill or campaign, HUMANElines tells where to go for more information, and provides names, addresses, fax numbers, and email addresses to send your concerns. HUMANElines is always clear, concise, articulate, and written in the animals' interests.

When you do write, remember to:

- Be brief and to the point.
- When appropriate, use facts and first-hand experience to support your position.
- Be courteous and reasonable.
- Be sure to sign your name.
- Don't wait until the last minute!

Two final notes before you get writing: If you live in the United States, visit www.hsus.org/programs/government/scorecard_shaping.html or call 202/955-3668 for a 1999 Congressional Scorecard for Animals. And don't forget to visit the IWRC's website at www.iwrc-online.org. The "Wildlife News" section features short articles about bills and programs concerning wildlife around the world. Current issues include fox hunting in Britain, the California condor, tigers, and a crack-down on the illegal trade in endangered animals in China.

Marianne Skoczek
Editor

Bald Eagle Z-083: A Case Report That Keeps on Reporting

By Lori Arent, MS, and Jalila Abu, DVM

ABSTRACT: On 19 March 1999 the Raptor Center at the University of Minnesota admitted a 3-year-old bald eagle from Wabasha, Minnesota. She was presented in sternal recumbency, with rigidity of both legs and periodic whole-body seizures. The bird was diagnosed with pesticide poisoning and treated with atropine sulfate, Valium, activated charcoal, and supportive fluid therapy. After 10 days of treatment, clinical and physiological signs of poisoning were eliminated. The bird was provided cage rest for an additional week, exercised on a creance for 3 weeks, and released with a backpack satellite transmitter near Morton, Minnesota, on 4 May 1999.

KEY WORDS: Bald Eagle, Pesticide Poisoning, Atropine Sulfate, Satellite Transmitter

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Introduction

The Raptor Center (TRC) at the College of Veterinary Medicine at the University of Minnesota admits approximately 100 bald eagles annually. On average, 25% of these are admitted with symptoms of toxin exposure, primarily lead. However, about 3% of the eagles are diagnosed with pesticide poisoning (organophosphate or carbamate compounds). Typical symptoms of pesticide poisoning include posterior rigidity (legs are extended and rigid), clenched feet, tremors of the head and/or whole-body seizures, and bradycardia (a suppressed heart rate, from a normal range of 150–200 beats per minute (bpm) to as low as 60–80 bpm [Dr. Pat Redig, pers. comm.]). (Ritchie, et. al. 1994).

Case number BAEA Z-083 was a 3-year-old female bald eagle admitted from Wabasha, Minnesota, at 4 pm on 19 March 1999. She was presented in good flesh (4.08 kg) with several of the symptoms listed above, i.e., sternal recumbancy with extensor rigidity of the legs, clenched feet, and a heartrate of 130 bpm. She exhibited head tremors and began full-body seizures shortly after her admission exam was begun. Lab samples were quickly taken and the bird treated with 2 mg atropine sulfate (0.5 mg/kg) IV (Carpenter, et. al. 1996), followed a short time later with 4 mg Valium (1 mg/kg) IV to control the seizuring. She was also treated for possible lead poisoning with 200 mg calcium disodium EDTA (Meta-Dote), 50 mg/kg in 60 cc warm LRS IV. The bird was then placed in a padded patient cage in a dark room to minimize stimulation.

Diagnosis

The diagnosis of pesticide poisoning is often based on a depressed level of cholinesterase in the plasma. Specifically, organophosphate and carbamate compounds bind to and inactivate acetylcholinesterase, an enzyme responsible for breaking down acetylcholine, a neural transmitter at cholinergic nerve endings. The result is a build-up of acetylcholine and continual stimulation of some organs and muscles (Porter 1993), resulting in head tremors and seizures. Initial lab results on BAEA Z-083 are reported in Table 1 (see page 4).

The lab data indicated that the blood level of cholinesterase was definitely lower than normal for healthy eagles. Some literature suggests that a blood cholinesterase level of 50% below normal is diagnostic for pesticide poisoning (Ritchie, et. al. 1994). Based on this information, the bird's clinical symptoms, and the ability to rule out head or back trauma (through visual examination and radiographs), we concluded that the eagle was suffering from pesticide poisoning.

A treatment plan was then established based on this diagnosis. For the first three days this plan consisted of IV injections of atropine sulfate twice per day as needed. Due to the whole-body seizures that appeared to follow handling in the early days of treatment, the decision was made to minimize handling until the symptoms subsided. Atropine can be administered every 3 to 4 hrs if needed (Ritchie, et. al. 1994). Due to a reduction in the parasympathetic neurological signs observed as treatments progressed, the dose of atropine was tapered to prevent a decrease in gut mobility, which is often observed with atropine treatments (Ritchie, et. al. 1994). In addition, Valium (1 mg/kg) was given as needed to control whole-body seizures, as was supportive care in the form of warm fluids. Since the blood lead level

TABLE 1. BLOOD RESULTS FROM ADMISSION SAMPLES OF BAEA Z-083.

The CBC was performed at TRC; all other samples were run by veterinary labs at the University of Minnesota.

PARAMETER	VALUE	NORMAL RANGE
Packed Cell Volume	45%	37–45%
Total Solids	3.0 g/dl	3.5–4.5 g/dl
WBC Estimate	21,600, normal morphology	10,000–12,000
AST (SGOT)	556 U/L	153–370 U/L
ALP	60 U/L	30–82 U/L
CK	6167 U/L	0–1018 U/L
Calcium	9.0 mg/dl	8.2–10.6 mg/dl
Glucose	422 mg/dl	285–400 mg/dl
Uric Acid	5.2 mg/dl	5.5–14.8 mg/dl
Blood Lead	0.16 ppm	<0.1–0.2 ppm
Blood Acetylcholinesterase	0.08 delta pH units/hr	0.12–0.2 delta pH units/hr

**Normal range for plasma cholinesterase was determined by sampling healthy captive eagles at TRC who were on a known diet for a minimum of 4 years. Cholinesterase activity was measured at the University of Minnesota diagnostic lab using the Modified Michells Method (Stahr 1977).*

was negative, CaEdta treatments were discontinued.

Treatment

Day 2: On the morning of Day 2, the eagle was standing, but slight head tremors still existed. Her heartrate was normal (205 bpm). A dose of 1 mg atropine sulfate (0.25 mg/kg) IV was administered, along with 90 cc warm LRS. Two hours after treatment, the bird went into a whole-body seizure and 4 mg Valium (1 mg/kg) IV was administered. She was then kept in a dark area and monitored throughout the day. At 8 pm the bird was standing but wobbly. Head tremors were present and her heartrate was 152 bpm. Treatment included 90 cc warm LRS and 0.5 mg atropine sulfate (0.125 mg/kg) IV. No additional whole-body seizures were observed this day.

Day 3: The eagle was standing in her cage and head tremors were still present. Her heartrate was 172 bpm. Treatment consisted of 60 cc LRS SQ, 50 cc of an oral electrolyte solution gavaged, and 0.25 mg atropine sulfate (0.062 mg/kg) IV. When the bird was returned to her cage she began seizuring, and 4 mg Valium (1 mg/kg) IV was administered. At 4:30 pm the bird was standing, alert, with no head tremors noted. Treatment consisted of leaving her 50 g cut-up chicken breast soaked in an electrolyte solution.

Day 4: The eagle was standing and calm, with no tremors present. She did not eat the food offered the night before. Treatment consisted of 70 cc LRS SQ, 2 mg Valium (0.5 mg/kg) oral (the injectable form was not available) and 10 cc activated charcoal (1 g/10 cc water) gavaged to bind to toxins remaining in the gastrointestinal tract. In retrospect, this charcoal treatment should have been initiated at the time of admission. In addition, a course of itraconazole, an aspergillosis preventative, was initiated. Itraconazole (Sporanox) was given at a dose of 7 mg/kg orally twice daily for 5 days, and then once a day for 12 additional days. In the evening, the bird was standing and aggressive. No tremors were noted. The eagle was given 10 cc activated charcoal orally and was hand-fed 100 g clean meat (no fur or bones). A blood sample for cholinesterase was taken and the level was reported at 0.1 delta pH units/hour.

Day 5: The bird was standing, bright, alert and responsive. Fluids (70 cc LRS SQ) and itraconazole (30 mg) were given and 125 g of clean meat was left.

The eagle did not demonstrate any tremors or seizuring after Day 3. She continued to improve in attitude, but refused to eat on her own. For the next 5 days, fluids, hand-feeding, and itraconazole were maintained to support the bird. On Day 10, blood samples were drawn to assess the overall condition of the eagle. The results are in Table 2.

Food (150 g rat) was left for the bird after the sampling was completed. This was the first meal the eagle ate on her own, and she continued to do so from this day forward. The bird remained in a convalescent cage for 1 week and was then transferred to a small, indoor, free-loft room 3.7 m x 3.7 m x 3 m (12 ft x12 ft x10 ft).

Prerelease Conditioning

After one week in the small indoor room, the eagle was transferred to an

TABLE 2. BLOOD RESULTS FROM POST-TREATMENT SAMPLES

Acetylcholinesterase: 0.13 cc delta pH units/hr

PCV: 48%

TP: 33.2 g/dl

WBC estimate: 16,600

outdoor pen (18.3 m x 3.7 m x 3.7 m [60 ft x 12 ft x 12 ft]) and exercise on a creance was begun. The bird was flown on a 91.4 m (300 ft) parachute cord creance. During the first session, the bird was wobbly in flight, flew low to the ground, and made eight 91.44 m (300 ft) flights. The bird was very tired during the session and required rests of 1–2 minutes between flights. Over the course of the next 3 weeks, the bird was exercised 3 times per week, at the end of which time her flight was graceful, her mechanics solid, and a height greater than 3 m (10 ft) was maintained. The eagle easily flew 10 flights of 91.44 m (300 ft) without tiring. With her release imminent, a backpack satellite transmitter (sponsored by Nationwide Insurance) was attached (Kenward 1987) and the bird federally banded. The eagle was released on the Minnesota River in Morton, Minnesota on 12 May 1999.

Postrelease Monitoring

The satellite transmitter attached to this eagle allows postrelease monitoring for a span of 3 years. A variable schedule of signal emissions was established to originally get a location reading every 4 days for the first 2 months following release, then every 10 days for about 4 months before returning to a 4–day cycle. This information will help us evaluate the rehabilitation methods used and determine if there are long-term effects of the pesticide poisoning on this eagle's survival and reproductive success.

During the first 8 days following release, the eagle flew about 215 km (133 mi), moving primarily in an east/west direction along the Minnesota River. She began moving north about 2 weeks after release, staying in an area for about 2 weeks at a time before continuing north to a new location. The eagle flew approximately 916.5 km (569 mi) during the first 2 months postrelease, moved into Ontario, Canada, in September 1999, and has since settled in Lake County (boundary waters) in northern Minnesota. During the 7 months since her release, Z-083 has traveled 1272 km (790 mi), and has unsystematically moved from different locations throughout the state of Minnesota.

This eagle's past and future movements can be followed on TRC's web page, under eagle tracking, Z-083 (www.raptor.cvm.umn.edu).

Conclusion

Poisoning in raptors, primarily bald eagles, appears to be a continual cause of illness in the upper Midwest. About 25% of the bald eagles admitted to TRC have lead poisoning, and about 3% suffer from toxicity due to the ingestion of pesticide compounds. Prescribed treatment protocols for pesticide poisoning can be effective provided the exposure is relatively acute. Pesticide toxins result in hyperactivity of skeletal muscles, muscle fatigue, and paralysis; thus, chronic exposure and the resulting debilitation often result in death.

Bald eagle Z-083 was successfully treated for her acute poisoning with a regime of atropine sulfate, Valium, and supportive care. This treatment has previously been reported to be successful in bald eagles (Shimmel, et. al. 1999). The bird was effectively reconditioned using the creance technique (Chaplin 1989), as indicated in the extent of her postrelease movements identified by a satellite transmitter. What remains to be seen is the potential for long-term effects of the pesticide on this eagle's reproduction and survival. The transmitter attached to the eagle will provide us with information on her movements and activity for 3 years following release.

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ABBREVIATIONS USED IN THIS STUDY

LRS	Lactated Ringers
SRS SQ	Lactated Ringers administered subcutaneously
CBC	Complete Blood Count
g/dl	grams per deciliter
U/L	units per liter
CaEdta	Calcium disodium versenate
AST	aspartate aminotransferase
ALP	alkaline phosphatase
CK	creatinine phosphokinase

SUCCESS STORY

New Breakthroughs in Oiled Bird Rehabilitation

By Jay Holcomb and Mark Russell
International Bird Rescue Research Center, Berkeley, California

Species and Natural History

The mallard, American widgeon, and gadwall are all considered common in North America. They are dabbling ducks who spend most of their lives in freshwater lakes, ponds, and rivers feeding on plants and insects. The three species are heavily hunted species whose populations are considered stable.

How the Birds Were Obtained

The 6 birds discussed in this article were oiled in an inland oil spill that occurred in early February 1991 when a pipeline carrying San Joaquin heavy crude oil ruptured in Valencia, California, spilling oil into the Santa Clara River and traveling downstream for approximately 9.7 km (6 mi). The Santa Clara is a small, inconspicuous river, 16 km (10 mi) long, that is located in the mountains northeast of Los Angeles. The river is just 4.5 m (15 ft) wide at its widest and only gets about 1 m (3 ft) deep in certain areas where pools are created. There are many shallow areas that are thick with aquatic vegetation and that attract many ducks and wading birds. At approximately the 15.5 km (9 mi) mark, the river becomes very shallow. At this point, it widens into sand and gravel beds and gently sinks under ground.

The International Bird Rescue Research Center (IBRRC) began field collection of oiled birds within 24 hr of the pipeline rupture. The 6 ducks in this article represent 6 of the 166 live birds captured and admitted to the rehabilitation facility.

Problems and Concerns

Most of the birds captured in this oil spill were severely oiled with San Joaquin heavy crude oil. This is a highly viscous oil that contains a high percentage of sulfur and selenium. Petroleum can have serious short- and long-term effects on birds.

Comments on Rehabilitation

Within 24 hr of the pipeline break, IBRRC mobilized and began collecting birds in the field. Those who were oil affected were beginning to succumb to hypothermia as daytime temperatures reached 80–90°F. Hypothermia at night was another concern and was evident in birds captured early in the day as night-time temperatures dipped to 30–40°F.

Predation by raptors and carnivorous mammals was evident, and many oiled birds were found scavenged. Golden eagles, coopers hawks, barn owls, and turkey vultures were seen feeding on oiled ducks.

The facility used to rehabilitate these birds was located in Long Beach, California, at Terminal Island. This was about 80 km (50 mi) from the capture site. The facility was a prefabricated structure within a large warehouse used to repair boats. It had recently been used for another spill and was ready to house birds within 48 hr of activation. Water for an immediate rehabilitation response was available due to pre-existing water heaters and water softeners. Outdoor pens suitable for dabbling species were constructed and functional as soon as the birds were ready to be placed out-of-doors.

The stress faced by oiled wildlife during the rehabilitation process can be



PHOTO: IBRRC

Birds caught in the February 1991 Santa Clara oil spill were heavily oiled by San Joaquin crude oil, a highly viscous crude oil that contains a high percentage of sulfur and selenium.

great. In considering the 6 recovered-banded birds, it is important to note the amount of stress they underwent during their rehabilitation. San Joaquin oil is particularly viscous and covered the birds entirely. In some cases, the oil even impinged movement of wings and/or feet. Though these 6 birds had normal blood values upon admittance, it was important—and is IBRRC's policy—to stabilize all birds prior to washing. When birds are admitted, it is essential to first get their core temperature and to note their general condition. If, and only if, the temperature reads 100°F or greater is a thorough examination given and a blood sample taken. Fluids and Toxiban are administered following the exam. Toxiban is an activated charcoal suspension, used to absorb and move the toxins through the intestinal tract. The bird is then placed in a warm ventilated pen.

The degree of oiling should be noted, as well as the volatiles given off by the oil. Should the amount of oil impinge movement or pose a respiratory threat to the bird, a "quick" wash should be considered, but only if the bird has normal blood values, temperature, and body weight. A quick wash means washing the bird quickly, usually within 5 min, to remove just enough oil so the bird can be properly stabilized. By no means is a quick wash meant to rid the bird of all the oil. The bird is given a diet based on the results of the initial exam and re-evaluated in 24 hr. If all the blood parameters look adequate and the bird's behavior is normal after 24 hours they are regarded as stable and may be considered for the washing process.

To be considered for washing, a bird must meet certain criteria. The bird should be at a normal weight or be gaining weight and close to normal; the blood parameters should be PCV of 25 or greater, BC of 2 or less, and TP of 3–5 gm/dl. Any exceptions require the veterinarian's approval prior to washing. It has been found that an unacceptably high mortality will result when birds are washed at substandard levels.

Each of the 6 birds under discussion was subjected to an average of 3 washing sessions during the rehabilitation process. A washing session consists of putting the oiled bird through a series of warm water baths with a 1% Dawn (dishwashing liquid) solution. As many as 15 baths may be used per session. The wash session is concluded with a thorough, hot water rinse, using a high-pressured water nozzle. It is highly unusual to need more than 1 washing session. However, some residual oil remained after the birds had completed their first washing session; this is attributed to the viscosity of San Joaquin heavy crude oil, which is extremely difficult to wash out of bird feathers using conventional methods. Because the washing process is one of the most stressful procedures that a bird undergoes during rehabilitation, it was decided that the birds would benefit most by 2 or more regular washing sessions as opposed to one very lengthy one.

Despite being severely oiled and enduring multiple baths and stressful procedures throughout the rehabilitation process, upon release, these birds survived re-introduction to the wild and migrated out of the area before falling prey to hunters. An immediate response, professional care, and good facilities all contributed to this success.

The birds were periodically weighed and blood was taken every 3 to 5 days. They were housed indoors until they were completely clean and water repellent. They were then placed with conspecifics in outdoor caging. IBRRC's criteria for release is based on the overall health and behavior of each bird. A thorough physical exam is given and a blood sample is taken. The blood parameters for release vary from species to species. For dabbling ducks, the PCV should be close to 40%, the BC should be trace to 1.5%, and the TP should be 4–6 gm/dl. The bird should show normal behavior in an outdoor setting and be well fleshed.

However, there are some circumstances under which birds may be released at substandard blood values. The staff veterinarian, in conjunction with the wildlife rehabilitation staff, evaluates the condition of each bird. They may determine that it is in the bird's best interest to be released at slightly lower values than to remain at the center, where they will be subjected to captive stresses. These stresses may result in secondary care problems such as keel lesions, swollen joints, or bumble foot. This was the situation for bird 896-32541. Although his PCV was low (30%), the decision to release the bird was determined to be in his best interest. All 6 birds were released within 24 hr of the final evaluation.

At the time of the 1991 spill it was not always possible to take a blood sample on initial intake due to the number of birds admitted, availability of qualified staff at time of admittance, and other complicating factors. However, most birds had a sample of blood taken within the first 48 hr.

The Release and the Encounters:

Most of the birds were released in an estuary in the Long Beach area but a few long-term cases were released in Berkeley, California, near IBRRC's rehabilitation center (where they had received more intensive care). The

time that the 6 released birds were in the wild was 1006 days or 2.8 years. The number of days varied from 256 to 1417 days in the wild. All birds were shot and killed by duck hunters. Their bands were reported to the National Bird Banding Lab.

Table 1 lists the days in the rehabilitation center; the approximate distances that birds traveled from the release site to the location where they were shot; years in the wild; and the site and date released; and the site and date recovered.

Comments

The length of time that these birds survived after being oiled and rehabilitated is of particular interest. These birds were very heavily oiled and endured multiple baths and stressful procedures throughout the rehabilitation process. The birds' bloodwork during the rehabilitation process did not deviate significantly from the normals for their species. Upon release, these birds were able to survive reintroduction to the wild and migrate out of the area before falling prey to hunters. An immediate response, professional care, and good facilities all contributed to this success. Because all 6 birds were eventually shot and killed by hunters, it is not possible to know how long their natural lives may have been.

Oiled wildlife rehabilitation remains controversial, primarily because in the past many oiled wildlife rehabilitation efforts were hindered by limited funding, poor or (often) nonexistent facilities, and a general misunderstanding of the rehabilitation aspects of oiled wildlife rehabilitation. Many oiled wildlife rehabilitation programs resulted in low survival rates, prompting the questions: Is it worth it attempting rehabilitation of oiled wildlife? and Do the ones who make it to release survive?

These band returns are important indicators that let us know that some oiled birds do survive rehabilitation. However, many questions remain unanswered. Did these birds breed and were they successful? What were their movements before being shot? What were the long-term internal effects, if any, of the oil? Based on the band return data, we do know that all of the birds had successfully migrated at least once before they being killed. These band returns are very promising and point to the need for further postrelease studies of oiled wildlife. There are currently a number of studies following rehabilitated oiled birds that will hopefully help to answer these questions.

For more information on the rehabilitation of aquatic birds, contact IBRRC at 510/841-9087 or by email at ibrrcdir@aol.com.

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TABLE 1.

FED. BAND # & SPECIES	DAYS IN CAPTIVITY	YRS IN WILD	APPROX DIST FR RELEASE	RELEASE DATE & LOCATION	RECOVERY DATE & LOCATION
896-32088 (mallard)	17	3.7	105 km (65 mi)	19FEB91 Long Beach, CA	10OCT94 Castaic Lake, CA
896-32038 (mallard)	13	3.9	145 km (90 mi)	15FEB91 Long Beach, CA	3JAN95 Desert Hot Springs, CA
896-32042 (widgeon)	14	0.7	607 km (377 mi)	17FEB91 Long Beach, CA	31OCT91 Sacramento, CA
896-32049 (gadwall)	15	2.9	690 km (429 mi)	18FEB91 Long Beach, CA	5JAN94 Gridley, CA
896-32117 (widgeon)	19	1.8	692 km (430 mi)	24FEB91 Long Beach, CA	30DEC92 Lambertville, CA
896-32541 (widgeon)	53	3.5	1014 km (630 mi)	29MAR91 Berkeley, CA	12OCT94 Bear River Refuge, UT
Averages	22	2.8	542 km (337 mi)		

BOOK EXCERPT:

Captive Care and Medical Reference for the Rehabilitation of Insectivorous Bats

Amanda Lollar and Barbara Schmidt-French

I. Introduction

NATURAL HISTORY OF BATS

Bats are among the most beautiful and agile creatures to grace our night skies, yet they remain one of the most mythical and secretive animals on Earth. They are the only flying mammals in the world, and there are approximately 940 different species worldwide (Nowak 1994). They are represented symbolically in ancient cultures, ranging from Egyptian and Mayan ruins to Chinese art. Although revered in some cultures, they are feared in others, and many species have been persecuted to the brink of extinction. In fact, bats are both an ecologically and economically valuable component of our natural environment.

Approximately 20% of all bats worldwide are frugivorous (i.e., fruit and nectar feeders). They are important pollinators and seed dispersers and play a major role in the reforestation of the world's rain forests. Most of the remaining 80% are insectivorous (i.e., insect feeders). These bats are important in controlling insect populations, including many significant agricultural pests. A few species are specialized for catching and eating fish (i.e., piscivorous). About 1% of all bats are carnivorous, feeding on small vertebrates such as frogs, lizards, birds, or other bats. There are also three species of vampire bats. These bats are sanguivorous (i.e., feed on the blood of birds or mammals) and are found only in Mexico, and Central and South America.

Forty-one of the 45 bat species found in the United States and Canada are insect eaters, feeding on a tremendous diversity of insects, including beetles, moths, crickets, flies, leafhoppers, ants, mosquitoes, and true bugs, to name a few. Some, such as big brown bats (*Eptesicus fuscus*), are beetle specialists (Whitaker 1993, 1995); others, such as Mexican free-tailed bats (*Tadarida brasiliensis*), feed mainly on moths (Whitaker, et al. 1996). Because beetles and moths lay eggs that develop into larvae that feed on vegetation, including many domestic crops, bats are critical to agricultural interests in the United States and Canada. For example, Mexican free-tailed bats feed on the moth *Helicoverpa zea*. The larva of this moth is the corn earworm, a serious pest that causes significant damage to a wide range of crops.

Bats have also been observed feeding directly on insect larvae in the wild. Patricia Winters of Ross, California, observed pallid bats (*Antrozous pallidus*) feeding on tomato hornworms (*Manduca quinquemaculata*) that they had carried back to their roost (Winters 1997). Tomato hornworms are a serious pest on tomato, tobacco, potato, and other solanaceous (i.e., belonging to the family Solanaceae) crops in the southern United States. Only three species of bats in the United States feed on nectar or pollen. These three species are the long-nosed bat (*L. nivalis*), the lesser long-nosed bat (*Leptonycteris curasoae*), and the Mexican long-tongued bat (*Choeronycteris mexicana*). All three of these species are found in the Southwest and are important pollinators of cacti and agave. They also feed on fruit, as does the Jamaican fruit bat (*Artibeus jamaicensis*), which ranges north into the Florida Keys.

The *Journal of Wildlife Rehabilitation* is pleased to feature three chapters from *Captive Care and Medical Rehabiltation of Insectivorous Bats*, a comprehensive 329-page reference by Amanda Lollar and Barbara Schmidt-French.

KEY WORDS: Bats, Species Identification, Rabies, Hand-Feeding, Self-Feeding, Mealworm Media, Infant Formulas

AMANDA LOLLAR is president of the Beneficial Animal Teaching Society (BATS) and founder and executive director of the Bat World Sanctuary and Educational Center in Mineral World, Texas. This facility provides sanctuary to more than 100 nonreleasable bats from around the world; the organization also maintains a wild sanctuary that is used throughout the year by up to 20,000 Mexican free-tailed bats. Lollar has studied the behavior of the Mexican free-tailed bat extensively, in both the wild and captivity. She has authored or coauthored both popular and scientific literature on this species, documenting reproductive behavior and clinical signs of pesticide poisoning and rabies.

BARBARA SCHMIDT-FRENCH is conservation information specialist with Bat Conservation International in Austin, Texas. She has studied the reproductive behavior of the Mexican free-tailed bat and maintains a nonreleasable captive study colony of more than 60 individuals. She also specializes in the care of the red bat, and has studied the birthing and captive parenting behavior of this species. She has authored articles on bats for Microsoft Encarta Encyclopedia.

Bats are nocturnal (i.e., nighttime) feeders, roosting during the day in a variety of places. Some are crevice-dwellers and roost in places like caves, abandoned mines, rock crevices, old cisterns or wells, hollow trees, under bridges, or beneath tree bark. Crevice-dwellers may also roost during the day in human dwellings like the attics of homes or old buildings, under shingles, or behind window shutters. Others, like the Lasiurine bats (i.e., yellow, red, seminole, and hoary bats), roost more openly in the foliage of trees or other vegetation. Many of these bats resemble dried leaves and are well camouflaged in this setting.

Most bats in the United States and Canada either migrate to warmer climates during the colder winter months, or hibernate in caves or underground networks like mines, where warmer and more stable temperatures prevail. Only a few species remain active year-round. Some species, such as big brown bats (*E. fuscus*), occasionally hibernate in the attics of homes. Although hibernating bats may arouse during the winter, each arousal leads to the expenditure of almost 30 days of body fat when the bat is in deep torpor. Unnecessary arousals could therefore cause a bat to use up so much of its fat reserves that it might not survive the remainder of the winter. Hibernating bats are thus extremely sensitive to disturbance during this period of time. In addition, because some species of bats come from multistate regions and congregate in great numbers (as many as tens or even hundreds of thousands of individuals) at a small number of hibernation sites, disruption or disturbance of any kind to these areas may result in the loss of large numbers of bats that impact agricultural interests in many states or provinces. This manual focuses on the rehabilitation and captive care of the 41 species of insectivorous bats found in the United States and Canada. The range of many of these species also extends into Mexico, and some into Central and South America. Wildlife rehabilitation of these animals cannot begin, however, without first obtaining the appropriate permits and vaccinations.

WILDLIFE REHABILITATION PERMITS

Regulations regarding the captive care of indigenous bat species vary significantly from state to state. These regulations are typically determined by state wildlife agencies or fish and game divisions. To determine the specific requirements that apply to you, begin by contacting your state Department of Natural Resources. Some bats are federally listed as threatened or endangered by the United States Department of Agriculture (USDA), Fish and Wildlife Service. Currently, the endangered species include the gray bat (*Myotis grisescens*), the Indiana bat (Myotis sodalis), the long-nosed bat (*Leptonycteris nivalis*), the lesser long-nosed bat (*Leptonycteris curasoae*), the Hawaiian hoary bat (*Lasiurus cinereus semotus*), the Ozark big-eared bat (*Corynorhinus = Plecotus townsendii ingens*), and the Virginia big-eared bat (*Corynorhinus = Plecotus townsendii virginianus*). These animals are federally protected and require special permits for handling and emergency care.

Even a veterinarian is required to hold valid permits for the care of endangered species, except under certain circumstances. For example, a veterinarian can render emergency first aid for an endangered species as needed, but must turn it over to the appropriate authorities once stabilized. In addition, each state has its own list of threatened or endangered animals. These animals are protected regionally and may also come under special permitting requirements. You can obtain this listing and requirements from your state wildlife agency or natural resources department.

RABIES, HISTOPLASMOSIS, AND SPECIAL HEALTH PRECAUTIONS

Bats, like all mammals, are susceptible to the rabies virus. Although they are not considered to be asymptomatic carriers of the disease, some species are natural reservoirs of the virus. The rabies virus can be transmitted to a susceptible host through contact of saliva or nervous tissue of a rabid animal with deep body tissues (as takes place with a bite) or with mucous membranes (e.g., nose, eyes, and mouth). All rehabilitators considering the captive care of mammals should receive pre-exposure rabies vaccinations. This involves a series of injections (usually three) given over a period of several weeks. Bats should not be cared for until you have received the entire series of vaccinations. Although relatively painless, the vaccine is often costly. It can, however, be obtained relatively inexpensively if a group of people receive immunizations at the same time. Members of wildlife rescue organizations or employees of zoos or veterinary clinics often organize vaccination schedules with a local physician so that all members receive immunizations from the same multidose vial of vaccine, thereby decreasing the cost of each injection because the remaining doses are not discarded.

Known (or suspected) contact of a pre-exposure immunized person with a rabid animal (or a suspect animal that is unavailable or unsuitable for testing) should be reported to your personal physician and will generally be treated with a series of two postexposure vaccinations, one administered on day zero (the day of your first visit with your physician—always contact your physician the day contact takes place), and the second one on day three. Your personal physician should be informed of the dates of your last vaccinations so he/she can determine the appropriate vaccination program for your particular case. Bat rehabilitators should have their rabies titer checked annually. This is a measure of the level of rabies antibodies in your blood. If it drops below an acceptable level (1:5 as recommended by the United States Centers for Disease Control and Prevention), a booster vaccination will be recommended.

It is important to understand that animals suspected of having human contact should be considered for testing of the brain tissue. (Note: Not all

contact with rabid animals results in the transmission of virus.) It is not possible to reliably exclude rabies infection in an animal, regardless of the lack of any clinical signs of disease or even the length of time it has been in captivity, without euthanizing it and submitting it to your state or provincial health department for testing. If you have concerns about potential contact with an infected animal, you should contact your personal physician or local health department immediately. If you are unable to reach local authorities, you should contact the Viral and Rickettsial Diseases Division of the United States Centers for Disease Control and Prevention (CDC) in Atlanta, Georgia, at 404/639-1075 during working hours, or at 404/639-2888 nights, weekends, and holidays.

Contact between humans and potentially rabid animals causes great concern among public health officials. Rabies is, after all, a lethal disease. Such incidents also frequently receive sensationalized coverage by the media, which, unfortunately, sometimes leads to hysteria and the needless destruction of wild bat populations. Although the frequency of infection in wild bat populations remains relatively low, about one half of one percent (anywhere from 1 in 200 to 1 in 1000 depending on the species [Constantine 1988]), those taken in by rehabilitators are the sick and injured, and are therefore rabies suspects. There is likely to be a higher rate of infection in these animals, as is the case with animals submitted to public health departments, which frequently only accept animals for testing that have had contact with humans or domestic pets. It is, therefore, important to use extreme care, not only when handling, but in determining whether or not to accept an animal for rehabilitation.

Incidents involving animals that have had contact with humans should be reported immediately to public health officials. Some rehabilitators retrieve animals involved in such incidents and submit them to the proper authorities (i.e., health department). Others, for liability purposes, prefer not to retrieve these animals, but to notify the caller that they must report the incident immediately to the proper authorities themselves. If you accept an animal involved in such an incident, it is important to record the name, phone number, and address of all individuals involved so that this information can be relayed to authorities when the animal is submitted for testing (and the animal should be submitted immediately). If you choose not to accept such an animal, it is essential to keep a log and document the name, address, and phone number of the caller, along with the recommendation that the incident be reported immediately to the proper authorities.

Although it has been the authors' experience that infected animals demonstrating signs of disease usually die within a few days to a few weeks after they are taken in, this would not be the case with animals still incubating the virus. In fact, for bats, one incubation period reported in the literature involved a bat taken into captivity that lived 209 days before the onset of clinical signs (Moore and Raymond 1970 [cited by Brass 1994]). Other researchers (Kaplan 1969 [cited by Brass 1994]; Trimarchi 1978 [cited by Brass 1994]) have mentioned instances in which an incubation period of over one year was reported. Remember, contact with rabid animals must be treated immediately; it is therefore inappropriate and possibly life-threatening to accept an animal that has had human contact, care for it until it dies, and then submit it for testing.

The authors also strongly recommend that rehabilitators not give medical advice to individuals having contact with potentially infected animals. Instead, instruct them to seek medical attention from health professionals (i.e., personal physicians, local health departments, or the Viral and Rickettsial Diseases Division of the United States Centers for Disease Control and Prevention). Medical professionals will make recommendations based on the specifics of individual cases.

Histoplasmosis is a disease of man and other mammals. It is a fungal disease which frequently manifests with respiratory symptoms and is contracted by inhalation of spores of *Histoplasma capsulatum*, a naturally occurring soil fungus. Like most fungi, it requires warm, humid conditions for growth and is known primarily from the Mississippi and Ohio River Valleys and adjacent areas, especially in bird droppings. Conditions necessary for growth are also found in some cave environments where the fungus may be found in bat droppings. For this reason, caves containing large bat populations should be avoided (for the protection of the bats as well as humans). Such conditions may also exist in buildings in warm, humid climates. The fungus can be found in these areas in bird or bat droppings. Disturbance of dry fecal deposits causes the aerosolization of spores which may then be inhaled by humans. The seriousness of the infection may depend on the time spent in the vicinity, the quantity of spores inhaled, and any pre-existing health conditions of the victim. A small percentage of people may become seriously, even fatally, ill with this disease. It is therefore important that pest-control operators who clean up bird or bat droppings after animals have left (or have been safely excluded from) human buildings wear organic respirators capable of filtering out small fungal spores (i.e., particles as small as 2μ in diameter) in order to minimize the potential of contracting the disease. (Simple dust masks are insufficient for this purpose.) It is also recommended that deposits be dampened with water before removal in order to minimize aerosolization of fungal spores.

We strongly recommend giving careful consideration to the above health issues before beginning the rehabilitation of bats or any other mammalian wildlife. Not only your own life, but the lives of others, will depend on the knowledge you have beforehand, the procedures you use, and the kind of referrals you make.



II. Identification

SPECIES

It is important that rehabilitators are able to identify bat species. This information is valuable in that many state wildlife divisions use information obtained from annual reports that permitted rehabilitators are required to submit to assist them in monitoring species of special concern. In addition, because different species of bats have different natural histories, captive care routines including caging, feeding, and even grooming may vary significantly from one species to another. Differences in roosting habits in particular will determine the manner in which a bat is initially rescued, temporarily or permanently housed, and subsequently released.

Bat species are identified by a variety of characteristics including the length of various body parts (e.g., forearm, ear, foot, tail), presence or absence of various anatomical features (e.g., keeled calcar, nose-leaf, tail), the shape of various anatomical structures (e.g., ears, tragus), varying degrees of fur on different parts of the body (e.g., the tail), weight, and coloring, as well as various dental and skeletal (e.g., cranial) characteristics. Measurements often vary by only millimeters from one species to another. The color of the fur frequently varies from one individual to another, making it impossible to identify most species on the basis of color alone. In truth, most species will look very much alike to an inexperienced caretaker. The authors frequently receive requests for assistance in species identification. The individual specimen in question is oftentimes described as "very small with pointed ears and brown fur" (the infamous LBJ's, or "little brown jobs"), a description that could be used for most of the North American insectivorous bats!

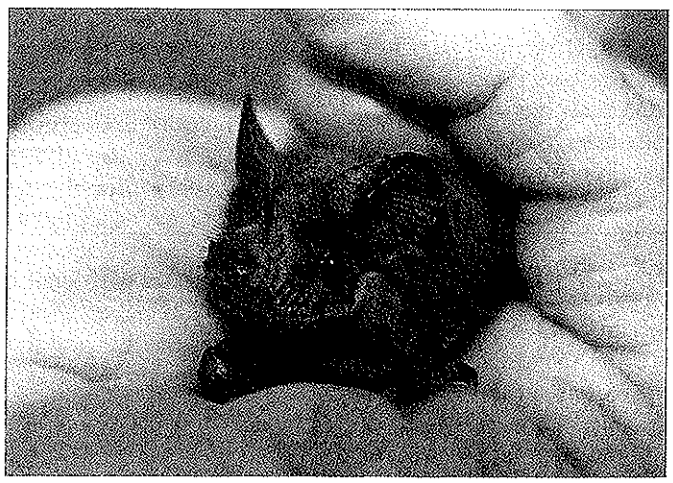
There are many keys available for identifying North American bat species (Barbour and Davis 1969; Jones, Armstrong, and Choate 1985; Harvey 1992; Nagorsen and Brigham 1993; Kunz, in press). When possible; use one specific to your area. The length of the forearm is one of the most critical measurements for identifying bats. You will need a ruler scaled in millimeters to take these measurements.

AGE

You must be able to distinguish an infant from an adult in order to determine an appropriate diet. Due to the small size of North American insectivorous bats, inexperienced handlers sometimes mistake adults for infants. (See Anthony, 1988, for detailed information on age determination in bats.) Most pups are born naked. Their skin is pink for at least the first few days to the first couple of weeks of life. The skin darkens as the fur begins to grow.

Young of some species, such as Mexican free-tailed bats (*T. brasiliensis*), are born with their eyes open (Lollar 1994). Others are born with eyes closed. French has found that red bats (*L. borealis*) do not begin to open their eyes until day 10 or 11. Most species will be fully furred within a few weeks at most. Red bats (*L. borealis*) are well furred dorsally by one week of age, although the abdomen is still naked at this time. They are fully furred within 15 days of birth. Mexican free-tailed bats (*T. brasiliensis*) develop more slowly and are not fully furred until about four weeks of age, although their pink skin begins to darken after the first or second week.

Birth weights and growth rates vary, but many of the medium-sized bat species (adult weight of 10 g to 20 g) weigh just under 2 g at birth. French has observed several dozen red bats (*L. borealis*) from birth and found that they typically weigh 1.75 g within 24 hours. Females weigh about 4 g at one week, 6 g at two weeks, and 8.5 g at three weeks. Males are smaller and weigh about 3.5 g at one week, 5 g at two weeks, and 7 g at three weeks. Both sexes reach adult weight by four to six weeks of age. Smaller-sized bat species (adult weight of under 10 g) may weigh less than a gram at birth. Most





pups will attain adult weight by six to eight weeks of age.

Nursing pups have tiny, sharp-hooked milk teeth which they use to cling to the mother's teat or fur. Young bats also have elongated, cartilaginous areas of the finger (phalanges) and hand (metacarpals) joints for the first several months of life; these joints in adults are no longer cartilaginous, but round, bony knobs.

ROOSTING BEHAVIOR

Crevice-dwelling and foliage-roosting species are identified as follows.

Crevice-Dwelling Bats

These bats roost during the day in a variety of places, including caves, abandoned mines, rock crevices, old cisterns or wells, hollow trees, under bridges, behind signs, or beneath tree bark. Crevice-dwellers may also roost during the day in human dwellings in the attics of homes or old buildings, in barns, under shingles, or behind window shutters. Some species roost openly within natural or man-made structures (e.g., *Corynorhinus*), while others roost within small cracks or crevices (e.g., *Tadarida*). Crevice-dwelling bats are often shades of gray, tan, brown, or black—colors that blend well with their roosts. We categorize the following genera as crevice-dwelling bats: *Mormoops*, *Eumops*, *Nyctinomops*, *Tadarida*, *Antrozous*, *Euderma*, *Eptesicus*, *Idionycteris*, *Myotis*, *Nycticeius*, *Lasionycteris*, and *Corynorhinus* (= *Plecotus*). The pipistrelles (*Pipistrellus subflavus* and *Pipistrellus hesperus*) may roost in either crevices or foliage (these bats sometimes roost in clumps of Spanish moss in trees), and may therefore be cared for as either crevice-dwelling or foliage-roosting bats.

Foliage-Roosting Bats

Unlike the crevice-dwelling species that roost inside a variety of natural or man-made structures, these bats commonly roost in the open among the foliage of trees or other vegetation. They often hang by one foot attached to a small twig or stem of a leaf. Although they roost in the open, these species are well camouflaged, ranging in shades from yellows and oranges to bright reds, colors that resemble dried leaves or fruit. We include the following as foliage-roosting species: the red bat (*Lasiurus borealis*), the desert red bat (*Lasiurus blossivilli*), the seminole bat (*Lasiurus seminolus*), the northern yellow bat (*Lasiurus intermedius*), the southern yellow bat (*Lasiurus ega*), the Arizona or western yellow bat (*Lasiurus xanthinus*), and the hoary bat(*Lasiurus cinereus*).

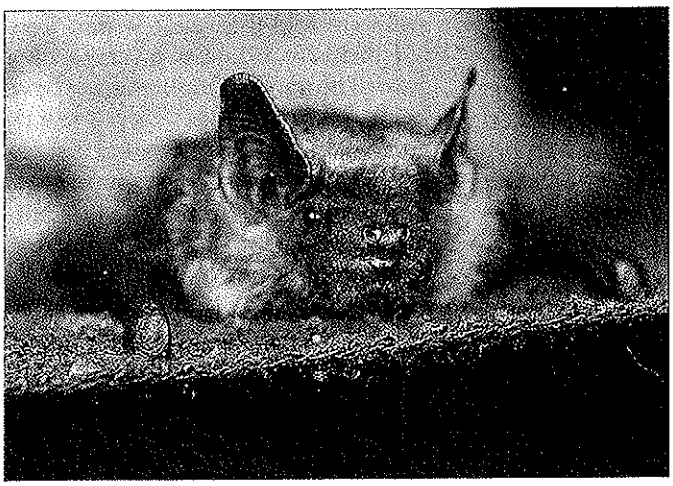
(Far left) The Mexican fruit-tailed bat (*Tadakida brasiliensis*), a common crevice-dwelling bat, is born pink and hairless. The ears are laid flat against the head at birth and the umbilical cord and dried afterbirth remain attached to the pup for up to 48 hours. By two to three weeks of age, the skin begins to darken as fur grows.

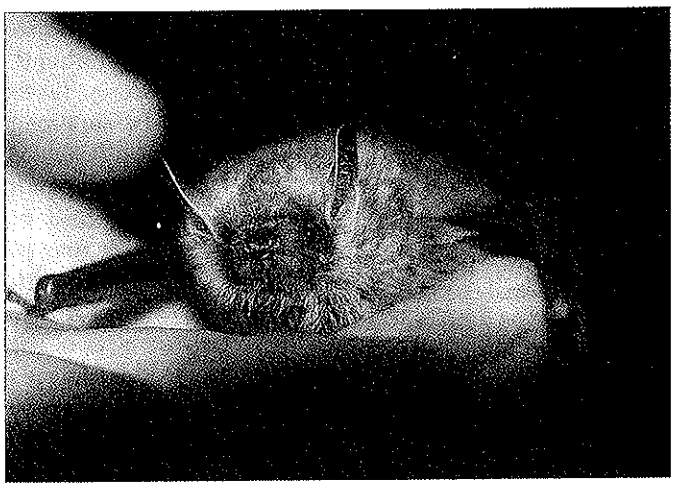
(Second from left) The red bat (*Lasiurus borealis*) is a common foliage-roosting bat. Pups have patches of peach-like fuzz within 24 hours of birth. By 5 days of age, males and females are easily distinguishable: males develop a reddish-yellow pelage; the females are commonly grey. By 15 days of age, pups are fully furred and the eyes are open.

(Second from right) The big brown bat (*Eptesicus fuscus*) is a common crevice-dwelling bat.

(Far right, below) The pipistrelle bat (*Pipistrellus subflavus*) roosts in both crevices and foliage.

ALL PHOTOS: AMANDA LOLLAR/BAT WORLD





III. Feeding

FEEDING ADULT BATS

Bats, like other mammals, must be sufficiently rehydrated before being fed. The nutritional content of insects varies from one species to another, although insects are generally a high-protein food item. Specific caloric requirements for many bat species are unknown. However, one proposed energy budget for a fringed myotis, (*M. thysanodes*) in early pregnancy (weighing 9 g) is 3341 calories (0.37 kcal/g/day) during nocturnal activity (flight and night roost maintenance), and 1410 calories (0.16 kcal/g/day) for day maintenance at 68° Fahrenheit (20° Celsius) for a regulating female, or 690 calories (0.08 kcal/g/day) for a nonregulating female. The total energy budget would thus be 4751 calories (0.53 kcal/g/day) for a regulating female or 4031 calories (0.45 kcal/g/day) for a nonregulating female (Hill and Smith 1984). (By entering torpor and allowing their body temperature to drop to levels below the lower thermal critical, that is, below the ambient temperature at which an animal must expend metabolic energy to maintain a constant warm internal temperature or homeostasis, many microchiropteran bats are able to decrease energy demands.)

Insectivorous bats probably obtain some of their vitamin and mineral requirements from green plant material present in the digestive tracts of some insects. A detailed discussion of the success and nutritional composition of various captive diets for insectivorous bats can be found in Rasweiler, 1977. This work includes valuable information about mammalian protein, carbohydrate, fat, vitamin, and mineral requirements and discusses conditions that can result from excesses as well as deficiencies. Further information about nutritional requirements of captive bats can be found in Wilson, 1988.

The authors do not presume to know the specific nutritional requirements of bats in the wild. We do know, however, what nutritional regimens will promote long-term maintenance of many species of bats in captivity. For example, although they have modified their diets somewhat over the years, both authors have maintained reproductive colonies of Mexican free-tailed bats (*T. brasiliensis*) on the same basic diets described in this chapter, and a number of individuals have survived for up to eight years to date. Many of the young conceived in these colonies have survived for four years to date and currently participate in mating activity. In addition, species considered particularly difficult to maintain in captivity, such as the red bat (*L. borealis*), have been successfully raised from birth to three years of age on these diets (French 1997).

The standard diet for most insectivorous bats in captivity consists of live mealworms. The authors feel that a mealworm diet should be supplemented with blended food mixtures and Linatone (a daily food supplement that contains both linoleic and linolenic acids). Bats should be given 1 drop (0.05 ml) of Linatone orally once a week. Alternatively, it can be added to the bats' drinking water (Barnard 1995). Other vitamin and mineral supplements are also necessary and can be added to blended food mixtures or drinking water. Do not exceed the authors' recommendations for vitamin or mineral supplements, however, as overdoses can result in serious disorders.

Mealworms are not actually worms at all but rather the larvae of the darkling beetle, *Tenebrio molitor*. This diet may be supplemented with crickets (*Acheta domestica*) for some bat species such as pallid bats (*Antrozous pallidus*) and big brown bats (*E. fuscus*). It may not be a good idea to feed wax moth larvae (*Galleria mellonella*) to bats because the outer tissue layer of the larvae can become embedded in the gums of a bat's teeth, resulting in gum infections and subsequent tooth loss over a period of time (Lollar 1994). The authors do not feed captive bats net sweepings (live insects caught out-of-doors) because of the possibility that some of them may contain pesticide residues that could adversely affect bats that are already sick or injured.

Although mealworms and crickets are often available at pet stores, they may be purchased in greater quantities for greatly reduced prices from several commercial distributors. Mealworms may be purchased in small, medium, large, and giant sizes. The smaller worms are best for bats weighing 3 g to 6 g, like the pipistrelles and smaller myotis bats. Medium and large-sized mealworms may be the best for species weighing more than 6 g. The large worms are simpler for the rehabilitator when feeding a bat that is only able to eat the viscera (i.e., insides) of the mealworm. This might be the case for a bat with very worn teeth or a gum infection that makes it impossible for the animal to chew the harder, chitinous outer portion of the worm. Bats do need the chitin in their diet, however, and you can provide this by blending the worms with other foods as described below.

Some rehabilitators question the wisdom of feeding giant-sized mealworms to bats. Many wildlife rehabilitators have expressed concern about substances fed to these worms that result in their larger size. The authors contacted two commercial insect supply companies regarding this issue, and were told that these worms were fed either a vitamin mixture or a growth hormone. Other animals have been known to develop growth anomalies due to pituitary imbalances that can result from growth hormones. Because it is impossible for a rehabilitator to determine specifically what these worms are fed, it is not recommended that they be fed to bats. In order to guard against dietary deficiency, it is critical that regular-sized mealworms be kept in a nutritionally enriched medium for several days before feeding them to bats. Researchers have

used a variety of vitamin and mineral supplements for this purpose. (Rasweiler 1977; Wilson 1988). Those preferred by the authors are listed below.

Mealworm Media

Mealworms commonly arrive packaged in a cloth bag filled with crumpled or shredded newspaper. Upon arrival, they should be placed immediately in one of the nutritional mealworm media listed in the box below.

Use finely ground powders and/or cereals so that the mealworms can be easily sifted out of the mixture with a large-holed strainer. The use of a sifter greatly reduces the time-consuming process of removing larvae (mealworms) from the culture at feeding time. It is important that no grains adhere to larvae fed to the bats because bats sometimes inhale small grain particles and suffer respiratory distress.

Mealworms fed to bats should not feel soft to the touch; choose mealworms that feel firm when rolled between your fingers. Adding bone meal to your medium results in firmer mealworms, which may promote healthy teeth and gums in bats. Mealworms frequently take on the coloration of the fruits, grains, and vegetables on which they have fed and are subsequently likely to taste different and add variety to a bat's diet.

Keep the mealworms and medium in a shallow, plastic storage container (at least 18" x 24"—46 cm x 61 cm—for 5000 worms). Either drill holes into the lid of the container, or cut out a large section of the lid and replace it with nylon window screen (simply hot-glue the screen onto the lid). This allows necessary air flow to keep the worms alive.

When you first receive a shipment of mealworms, keep them in one of the media listed above. Allow them to remain at room temperature for at least four days before placing the container in the refrigerator. This initial period allows the worms to "plump up" considerably. Stir the medium a bit each day to prevent it from becoming too tightly packed and molding.

Mealworms develop into pupae, which subsequently develop into beetles. It is recommended that you remove any beetles that develop in the medium containing adult worms because quinone secretions (from beetles) consumed by the mealworms may have toxic effects on bats (Barnard 1995). Many rehabbers have fed mealworm pupae to bats without detrimental effects. The beetles lay eggs in the apple or potato skins, and larvae (mealworms) hatch from the eggs. Refrigeration slows down this process and helps to prevent contamination of the medium by parasites such as grain mites.

Mealworm media should be discarded and replaced with fresh preparations every two to four weeks. It is particularly important that you do not keep mealworms in a warm, moist environment like a laundry room. An exceptionally humid environment will speed up the life cycle (i.e., development into the beetle) and will frequently result in the development of mold or fungal contamination which could be fatal if fed to bats. French replaces fruit in refrigerated mealworm media daily. Lollar removes any fruit or potatoes from mealworm media before refrigeration. Some rehabilitators prefer to culture mealworms themselves so that they need not continue purchasing them from supply houses. Place potato or apple peels in a container filled with one of the mealworm media given on this page. Mealworms from your supply can be placed in this container and allowed to pupate. Beetles that develop from the pupae will lay eggs in the peelings. Eggs will develop into tiny mealworms. Mealworms can be removed from the mixture and placed in a separate container of fresh medium before being fed to bats. Some of the mealworms should be left in the container with the beetles. These mealworms will pupate, and so the cycle begins again. The entire cycle takes a couple of months. It should be noted, however, that rehabilitators who care for many bats will probably not be able to culture a sufficient supply of mealworms on their own. The authors each use 15,000 mealworms weekly and so they must depend on supply houses to fill their requirements.

MEALWORM MEDIA Add up to 5000 mealworms to one of the following:

(Lollar's recipe)

- 1 cup (240 ml) Spiru-Tein (human vitamin/mineral supplement)
- 1 cup (240 ml) sterilized bone meal powder
- 2 8-oz (454 g) boxes mixed-grain baby cereal
- 1 12-oz (340 g) box wheat germ
- 1 thinly sliced apple (small)
- 1 thinly sliced sweet potato (small)
- 1 to 2 small, peeled carrots
- 1 fresh ear of corn
- 1 cup (240 ml) greens (organic)

(French's recipe)

- 2 cups (480 ml) wheat germ
- 2 cups (480 ml) oat bran
- 1/2 cup (120 ml) sterilized bone meal powder
- 1 cup (240 ml) C-Biotic (canine vitamin/mineral supplement)
- 1/2 cup (120 ml) UltraCare powder (avian vitamin/mineral supplement)
- 1 slice apple
- 1 slice sweet potato
- 1/4 cup (60 ml) greens (organic)

Hand Feeding Adult Bats

Most wild bats can be tempted to eat when first received by offering them a live mealworm. (Do not feed dead mealworms to bats because they decay quickly. Only if worms are offered to bats immediately after being killed should you do so.) Allow the bat to hang head down on the side of its cage or to rest horizontally inside a roosting pouch, and, using a forceps, gently run a mealworm along the bat's jaw line. This usually causes the bat to open its mouth, allowing you to place the mealworm inside. A bat will often clamp down on the worm, recognize it as food, and continue to eat. If the bat will not eat a mealworm whole, you should try offering it a

decapitated one. Pinch or cut off either the head (Lollar) or the last segment of the tail (French) of the mealworm, and, using forceps, squeeze out a small amount of the viscera (i.e., insides) onto the bat's mouth.

Some bats will immediately open their mouths and bare their teeth, inadvertently tasting the viscera and giving you the opportunity to again place the live worm in their mouth. (Mealworm viscera stains the fingertips. These stains can be removed by scrubbing the fingers with the green side of a Double-Action Polident tablet.) However, some bats will shy away from food offered in this manner and must be held gently but firmly in the hand with the head somewhat lower than the rest of the body. Mealworm viscera can then be offered as described above. If a bat still does not open its mouth, one of the blended food mixtures given here should be offered.

Although you may generally feed a bat as many worms as it will accept at each feeding twice a day for the first couple of days, it is important that you check the distention of the abdominal area periodically as it feeds. The area on the underside of the bat just between the legs should not be sunken in. (This is a sign that the bat has gone without food for too long.) Nor should you allow this area to become overdistended while feeding.

The number of mealworms a bat will consume in captivity will depend upon a variety of factors, including the species, the general health and condition of the individual animal, its nightly activity (i.e., is it flying), the season, and the ambient temperature. Although the authors prefer not to specify a given number of mealworms as appropriate for every feeding case, we are asked for this kind of information repeatedly by beginning rehabilitators. Generally speaking, bats should be fed twice a day, once in the morning before dawn and again in the evening after dark. We have typically found that species weighing between 10 g to 20 g may eat anywhere from 10 to 20 1-inch mealworms at each feeding, while smaller species weighing between 3 g to 10 g may only eat 5 to 10 mealworms at each feeding. The important thing to remember is to check the distention of the abdomen. (If a bat starts looking like it has a golf ball in its stomach, you've fed it too much!) Don't let the bat lose weight; a bat that comes in from the wild will never be overweight. Try keeping it within the appropriate range for the species, although the authors prefer that bats remain closer to the upper end of these ranges. You don't want to allow a bat to exceed the upper end of this weight range during the summer (i.e., active) months, however, because this will interfere with its flight capabilities. (And never release a bat that exceeds the upper end of the weight range for its species, as it will undoubtedly be caught by a predator!) It should be noted, however, that hibernating and migrating bats may increase their body weight by as much as 30% above their summer weight. Keep this in mind when dealing with bats that are to be released just prior to fall migration (i.e., movement to winter sites).

A variety of blended food mixtures have been used to maintain bats in captivity (Rasweiler 1977; Wilson 1988). These mixtures, often referred to as bat glop, are generally prepared in bulk, frozen, and then defrosted in small quantities and fed to bats. Use of blended mixtures was generally intended to minimize the time and/or expense of caring for multiple bats used in research studies. However, bats fed blended mixtures only (i.e., without live insect supplements) often did not fare well (Wilson 1988).

The authors have found that severely debilitated bats, and even some relatively healthy bats, will frequently refuse to eat mealworms or mealworm viscera when first taken into captivity. However, these bats can usually be encouraged to eat blended food mixtures that include veal (Lollar 1994). In addition, the authors feel that even bats that do accept live mealworms benefit from diets supplemented daily with 0.5 ml to 1.0 ml of one of the following blended mixtures. French uses a mixture of mealworms, veal baby food, banana baby food, and eggs. These ingredients are thoroughly mixed in a blender. The mixture is prepared as follows:

Preparing a Blended Mealworm Mixture

Blend:

2 cups (480 ml) mealworms (either frozen or fresh)
5 oz (142 g) veal baby food
2.5 oz (71 g) banana baby food
1 raw egg

(The following to be added after mixture has been blended.) To every 75 ml of blended mixture add:

3 drops (0.15 ml) Avimin liquid mineral supplement
3 drops (0.15 ml) Avitron liquid vitamin supplement

Although the mixture can be prepared with either fresh or frozen mealworms, it is less likely to overheat during blending if frozen mealworms are used. Remove dead worms. Even one bad mealworm will spoil the entire mixture. To prepare, place 2 cups (480 ml) of frozen mealworms in a blender, add 5 oz (142 g) of veal baby food, 2.5 oz (71g) of banana baby food (with no sugar or preservatives), and one raw egg. Set the blender on a relatively high (e.g., "grind") setting and use the "pulse" cycle until the mixture is ground up, and then set it on a lower (e.g., "blend" or "beat") setting until it attains the creamy consistency of a thick ice cream malt. The recipe makes between 12 and 15 fluid ounces (360 ml to 450 ml) of blended food.

If not blended for a sufficient period of time, the mixture may have a grainy consistency and still have small chunks of worms or softened worm skins that can get caught between a bat's teeth. In addition, many bats will not accept the mixture if it is at all "lumpy." The mixture should flow freely through the tip of a 3 ml syringe. Check to make certain it is thoroughly blended by drawing the mixture into a 3 ml syringe *while it is still in the blender*. If the entire 3 ml does not flow

freely back out of the syringe when the plunger is pushed in (i.e., it clogs the syringe), continue blending until it does. Check the temperature of the sides of the blender throughout the blending process. They should remain cool. If overheated, the substance will spoil and a bat will not eat it, which is why it is best to use frozen mealworms.

The blended mixture may be stored in a number of different ways. It can be poured into small baby-food jars and placed in the freezer until ready for use. The top layer of this mixture will turn a darker color. When needed, the frozen mixture may be placed in the microwave on "defrost" for just a minute or until thawed, and then heated on low for a few more seconds until warm. Then add 3 drops (0.15 ml) of Avimin, 3 drops of Avitron (0.15 ml) to each 2.5 oz (75 ml) of the blended food preparation. The mixture should be stirred thoroughly and the temperature tested before feeding it to a bat. It should be lukewarm (never hot). It is very important that you do not overheat the mixture when warming it prior to feeding. If overheated, moisture will be drawn out of the mixture and it will take on a lumpy consistency that most bats will not accept. In emergency situations when veal and banana baby foods are used without mealworms because mealworms are not readily available, be especially careful not to overheat the baby foods. The mixture should have a thick consistency. Overheating will turn this mixture into a liquid that bats will usually not accept.

Lollar uses another preparation that involves the blending of frozen mealworms. Mealworms should be free of medium and dead worms before freezing.

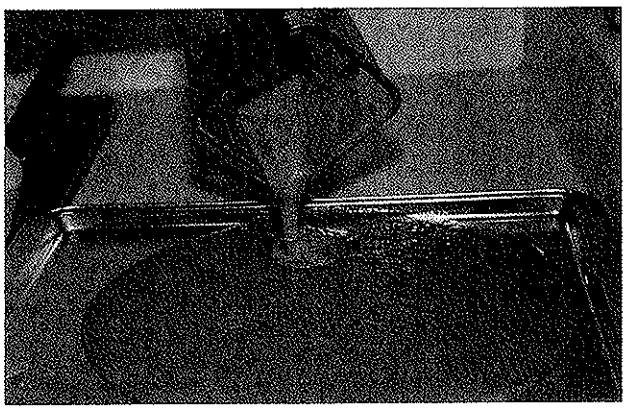
2.5 to 3 cups (600 ml–720 ml) frozen mealworms (i.e., 2000 large meal worms)

1 tablespoon (15 ml) Mother's Helper Puppy Formula (powder)

1 tablespoon (15 ml) Pro Balance, vitamin/mineral supplement

5 oz (142 g) veal baby food

1 hard-boiled egg



Blended food mixture made from mealworms and veal and banana baby foods.

Blend the ingredients on the liquefy setting until the mixture has the smooth consistency of a milkshake. After blending, pour a thin layer (1" or 2.54 cm thick) into a shallow, nonstick baking pan, and place it in the freezer until frozen solid.

Once frozen, the mixture can be broken into small pieces, placed in freezer bags or a freezer container, and then put in the freezer for storage. When ready to use, a few pieces of the frozen, blended mealworms are placed in a small bowl and allowed to partially thaw at room temperature. "Beginner" baby foods, including banana and one other bland flavor like sweet potato, applesauce, or pear, can be mashed into the partially thawed mealworms until smooth. The ratio for this mixture should be 3/4 blended worms to 1/4 baby-food fruit. Offering a flavor variety may be an option for bats in permanent captivity that must remain on blended food diets due to worn teeth. Add 3 drops (0.15 ml) of Avimin, and 3 drops (0.15 ml) of Avitron, to each 2.5 oz (75 ml) of the blended food preparation. For bats that appear particularly weak or undernourished, you may add Nutri-Cal or Stat dietary supplements to the blended mixture for initial feedings. Add 0.5 ml of Nutri-Cal or Stat nutritional supplements to every 3 ml of blended food for a period of two to five days.

Some bats, including *Pipistrellus*, *Nycticeius*, and species of *Myotis* may prefer daily supplements of mixtures that contain only veal and banana baby foods with Nutri-Cal.

By hand, mix the following:

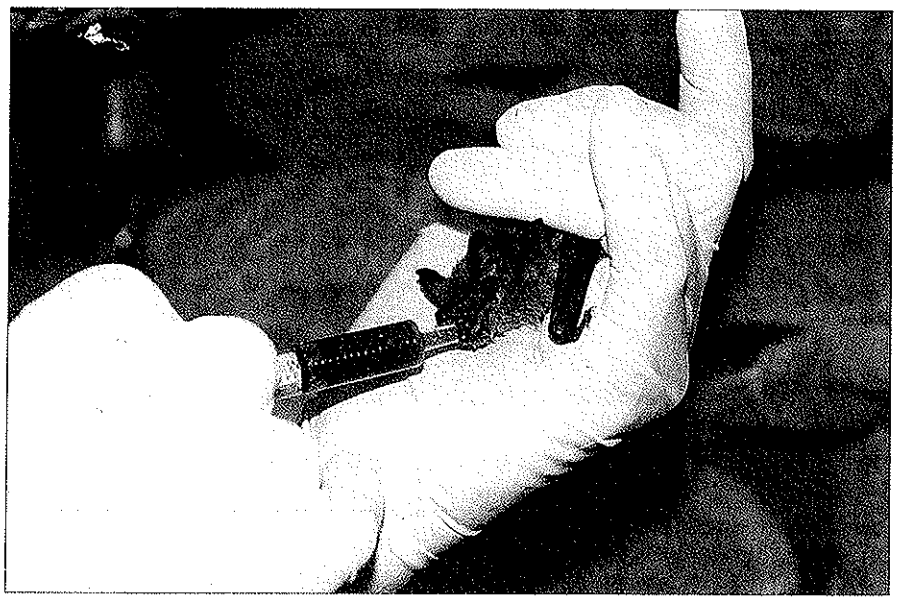
2.0 ml Nutri-Cal, a vitamin/mineral paste

2.5 oz (71 g) veal baby food

0.6 oz (17 g) banana baby food

The mixture must remain thick. Bats will refuse this mixture if heated until it turns into a liquid. Bats fed this mixture should also receive live mealworms or mealworm viscera on a daily basis. Bats that receive Nutri-Cal on a daily basis should not be given weekly supplements of Linatone. *It may be beneficial for all bat species to be given this veal, banana, and Nutri-Cal mixture once or twice a week.*

Use 3 ml syringes to feed blended mixtures to a bat. Monoject syringes are perhaps the best choice of syringe (others tend to clog easily). Again, mixtures that have not been thoroughly blended will also clog syringes, so be certain your mealworms are well-blended. Monoject syringes can be obtained from pharmacies or veterinary clinics. (They can also be ordered at reduced prices in boxes of 100.) Once the syringe is filled with the blended mixture, it should be placed in a bowl of warm water. Before feeding, the temperature of the mixture should be tested by placing a small amount on the inside of your wrist as you would do to test the temperature of a bottle of milk before feeding a baby. Again, the mixture should only be warm, not hot. If a bat is arousing from daily torpor, it will shiver as it warms up. It needs to be sufficiently warmed before feeding so that it can digest its food. A bat is also more likely



Gently forcing the lower jaw downward with the tip of a feeding syringe so that blended food can be placed in the mouth of this crevice-dwelling bat.

to aspirate or choke on food if forced to feed while it is shivering. You can speed up the arousal process by holding the bat in your warmed hand. When it has stopped shivering, hold it so that the head is slightly lower than the rest of its body, and place a small drop of the blended mixture inside the bat's mouth.

Many bats will immediately begin to eat the mixture as you dispense it through the syringe. When feeding species of the family Vespertilionidae, it is helpful to point the syringe towards the chin or to one side of the mouth to avoid getting blended food in the bat's nose. Others, such as the molossid bats, may require more prompting to get them to begin eating. Although many weak bats cannot be forced to eat live mealworms or even mealworm viscera, most can be coaxed into eating a blended mixture. Gently force the lower jaw downward with the tip of the syringe, and then place a small amount of food directly in its mouth.

It may take a few minutes for the bat to begin eating. Bats that are very weak will swallow the food placed in their mouths very slowly, and you will need to be very patient, allowing them to take their time with each mouthful. Even for bats that readily accept this food, it is important to wait a few seconds between mouthfuls to make certain they have swallowed all of the food before offering more. Some bats tend to eat too fast and will aspirate if not forced to wait a few seconds between mouthfuls.

Feed blended mixtures twice a day to bats unable to eat live mealworms, once in the early morning and again in the evening. Bats weighing 10 g to 20 g will typically eat 1.0 ml to 2.0 ml of a blended food mixture at each feeding. Bats weighing 20 g to 35 g may eat as much as 3.0 ml per feeding. Smaller species may only eat 0.5 ml to 0.75 ml per feeding. Even the smallest of bat species in the United States (i.e., a 3 g bat) should be fed a total of at least 1.0 ml per day (0.5 ml per feeding). It is not recommended that you exceed these quantities when feeding bats blended food mixtures. Bats that also eat live mealworms only need to be fed a blended food mixture once a day. Although they can be stored in a freezer for several weeks, these mixtures should not be stored in the refrigerator once thawed for more than one or two days, and blended food that has already been thawed should not be refrozen. It takes approximately 30 minutes to prepare enough blended food to feed 50 to 100 bats for a period of two weeks. About 25 healthy bats can be hand fed blended food mixtures per hour; however, it takes longer to fed debilitated bats.

Live mealworms should be fed to bats as soon as they will accept them, although the authors continue to offer blended mealworm mixtures (or the veal/banana/Nutri-Cal mixture) to all bats as a daily supplement. Permanent captives that have lost teeth or have extremely worn teeth can remain on a blended food diet for the rest of their lives. These permanent captives should also be offered the viscera from live mealworms daily in addition to the blended food.

Occasionally, an extremely debilitated bat will refuse even blended food mixtures. You may be able to coax such a bat into eating by feeding it warmed Nutri-Cal from a syringe. Heat a small amount of Nutri-Cal in the microwave for just a few seconds, and add just enough water to turn it into a liquid. Draw the warmed liquid into a feeding syringe and test the temperature on your wrist. It should be lukewarm and can be fed to the bat by gently lowering the jaw as described above, and putting a small amount (0.1 ml to 0.2 ml) of warmed liquid in the bat's mouth. Again, it is important that the bat is held with its head tilted lower than the rest of its body while being fed. Once the bat begins to swallow the liquefied Nutri-Cal, try inserting a small amount of one of the blended mealworm mixtures in its mouth and feed as described above.

The authors find it of interest that red bats (*L. borealis*) taken in from the wild will feed avidly on slices of avocado without prompting. French feeds avocado to adult red bats (*L. borealis*) twice a week. Cut a small wedge of the fruit and allow the bat to chew along the thin edge and eat as much as it likes.

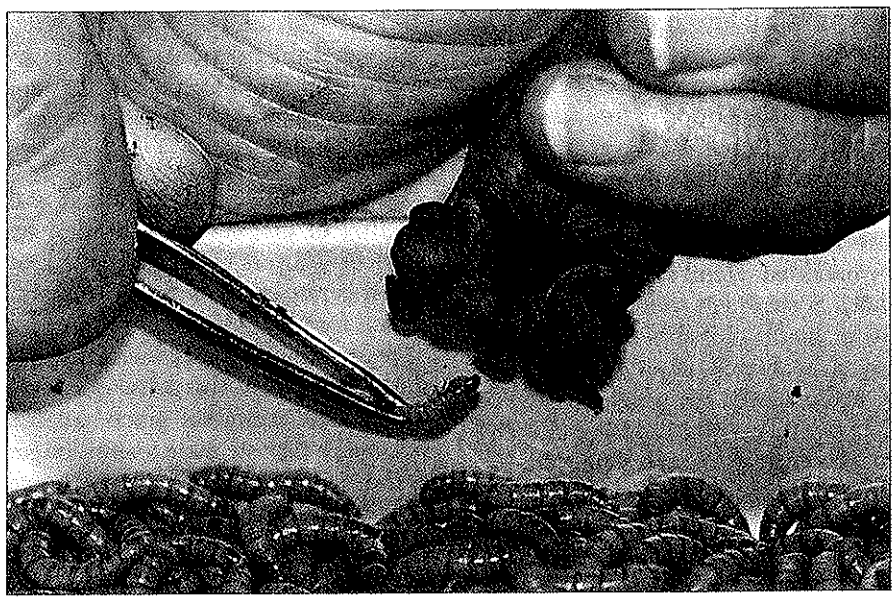
Teaching Adult Bats to Self-Feed

Some bats can eventually be taught to self-feed. (Others may do so immediately without being taught.) The length of time it takes depends upon the species and the individual. Bat species that glean insects from surfaces or forage for insects on the ground may

learn to eat from a tray placed on the bottom of a cage more quickly than species that mainly feed in flight. Molossid bats can be difficult to teach to self-feed. The facial structure of these bats appears to make it difficult for them to grasp a moving insect from a flat surface, although many can be taught to self-feed over time (Lollar 1994).

When first training a bat to self-feed, hold the animal over a small tray of mealworms. The tray should be shallow but large enough for the bat to climb in and out without allowing the mealworms to escape and heavy enough so that the bat can't tip it over. Mealworms can easily climb on roughened surfaces, so use a tray with a smooth surface. The authors have used plastic storage containers and small aluminum baking pans made specifically for children (Lollar), and small kitten trays (French) to hold mealworms. Remove mealworms from the tray with a blunt forceps, and offer them to the bat one by one. Hold the bat close to the tray so it can watch as you pick up each mealworm. Hand-feed it several mealworms until it begins reaching into the tray on its own.

Place the tray (with an ample supply of clean mealworms—enough to completely cover the bottom of the tray) into the corner of the bat's cage against the walls at night. Placement of trays against the walls allows a bat to crawl down the side of the cage and feed from a tray while still hanging upside down. Be sure that mealworms placed in trays for self-feeding are free of medium, shed skin, pieces of worms, and dead worms. Some bats will begin eating from a dish within a few nights. Remove the dish from the cage in the morning, remove guano, dead worms, and mealworm feces (the powdery substance you will find on the bottom of the tray), and place the remaining live worms back into the mealworm medium in the refrigerator. For smaller bat species only, some rehabilitators partially immobilize mealworms by pinching the neck just prior to placing them in a tray for bats that self-feed (P. Winters, pers. comm.). This



To teach crevice-dwelling bats to self-feed, remove mealworms from a tray on the cage bottom with forceps and offer them to the bat as it watches.

effectively immobilizes the mealworms but does not kill them for a few hours. However, once the mealworms have died, they should be removed. Bats should not be fed dead mealworms. French does not immobilize mealworms for the smaller bats, but instead feeds them just the smallest-sized mealworms while still mobile. All dishes and water containers should be emptied and washed in an antibacterial detergent daily. Rinse thoroughly with hot water.

Foliage-roosting species should not be fed from trays left on cage floors. Instead, they can be taught to feed by hanging small coop cups filled with live mealworms on the cage netting directly below the bat where it hangs from the side or top of the cage. Begin by removing worms from the coop cup with forceps and handing them to the bat. Hang the cup of mealworms as close to the bat as possible and feed the bat this way twice a day. Hang cups close enough to pups that their nose actually touches the mealworms, and leave them hanging like this for at least 5 to 10 minutes before feeding them worms with the forceps. Many adult foliage-roosting bats will quickly learn to feed from coop cups. It may take several months for some pups to learn to self-feed, however. Although red bats (*L. borealis*) may be taught to self-feed in this manner, they may be less likely to sustain injury if they are hand fed during their entire stay in captivity.

Mealworms should never be left in cages with severely debilitated bats or with bats that have sustained back or other injuries that seriously impede terrestrial movement because such bats can be overcome and eaten by mealworms.

Upon first learning to self-feed, some bats will overeat. This is true of big brown bats (*E. fuscus*) in particular. However, the authors have found that bats of several different species, including big brown bats (*E. fuscus*), Mexican free-tailed bats (*T. brasiliensis*), cave bats (*M. velifer*), evening bats (*Nycticeius humeralis*), and pallid bats (*Antrozous pallidus*) will in fact regulate their consumption within a period of approximately two to three months and remain within appropriate range for the species, despite initial (and sometimes dramatic) weight gains.

Although it can take some long-term captives a longer period of time (up to six months) to regulate their own weight, all of the 150 self-feeding bats maintained in permanent captivity at both the French and Bat World facilities, including a captive

colony of big brown bats, have regulated their consumption and remained within appropriate ranges for the species over a period of five years. It should be noted, however, that because some bats initially become obese when given unlimited access to food, bats intended for release may need to be hand-fed. Hand feed the bat twice daily. A small bat weighing less than 10 g should probably be fed five to seven mealworms at each feeding and a medium-sized bat weighing 10 g to 20 g should get 10 to 12 large (1 inch) mealworms at each feeding. If a bat intended for release was allowed to self-feed until it exceeded the upper end of the weight range for the species, its diet should be carefully adjusted by reducing each feeding by only two or three mealworms until the desired weight is attained. (Remember, many captive bats will also increase their consumption and body weight naturally in the fall in preparation for migration or hibernation.)

Although some bats may never learn to self-feed on live mealworms, they may feed from shallow trays filled with blended mealworm mixtures (Krutzsch and Sulkin 1958). Bats that do not learn to self-feed will need to be hand-fed for the duration of their captive lives. Rehabilitators who are not prepared to care for a bat in this manner should place it with a rehabilitator who can.

Feeding Crickets

Although many bat species will refuse to eat crickets in captivity, the common field cricket (*Acheta domestica*) can be fed to some of the larger bats as a supplement to mealworm diets. Crickets constitute a part of the normal diet in the wild for some bat species. Pallid bats (*A. pallidus*), big brown bats (*E. fuscus*), and other bats that sometimes glean insects from vegetation may enjoy an occasional cricket.

Field crickets are available from several insect supply houses. These insects are actually less nutritious than the mealworm diets because crickets are more selective in their feeding habits and cannot be provided with supplemental nutrients as easily as mealworms can (which is why they are only used as a supplement to traditional diets).

Information regarding the care and feeding of crickets is provided by Patricia Winters as follows. It is best to refrigerate crickets immediately on arrival for about 30 minutes in order to reduce their activity level, making the transfer from the delivery carton to a storage container much simpler. (Do not refrigerate them for any longer as they will die within just a few hours of initial refrigeration.)

Crickets should be maintained in a large aquarium or plastic storage container covered with a screened top. You can cut the center out of the lid of a plastic container and replace it with aluminum screening or hardware cloth. (Do not use nylon window screen. Crickets will quickly chew through it.) This screened lid will be placed over the top to keep the crickets from jumping out, but will allow sufficient ventilation. Place rabbit-food pellets, greens, and carrots on the bottom of the storage container and add a small dish of water filled with small pebbles (to prevent the crickets from drowning). Then place the crickets in the container and cover the screened lid.

Keep the storage container with crickets in a dry environment at about 70° Fahrenheit (21° Celsius). Dampness is a constant problem when housing crickets. Check the container carefully each day for any mold or fungal growth either in the container or on the food, and dispose of the entire contents, including the crickets, if you detect any contaminated materials. Thoroughly clean the container with a disinfectant and rinse well before adding fresh medium. All water and any remaining food must be removed daily and replaced with fresh supply.

When providing crickets as a supplement for self-feeding bats, Winters recommends that the insects be disabled by either removing the last pair of legs or by cracking the neck on the ventral side just above the thoracic shield. (The insect will still be alive, but will be unable to climb out of the dish.)

Water Routines

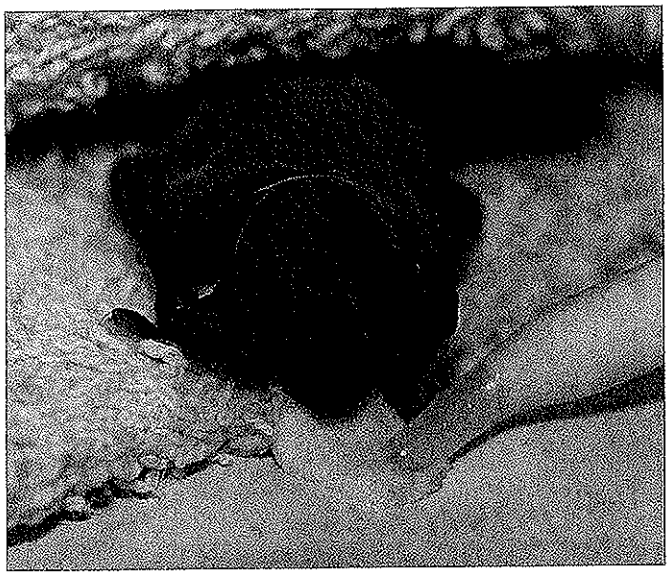
Fresh water (at room temperature) should be available to bats at all times. Because water dishes will become soiled with urine or feces, they should be washed and refilled twice a day—in the early morning and again in the evening. It is best to provide more than one water dish per cage. Water dishes for insectivorous bats should be very shallow, as these bats have been know to drown in trays filled with as little as 0.5 inches (1.25 cm) of water. Stainless steel (not galvanized metal or brass) automotive freeze plugs can be placed on the bottom or roosting cages and used as water dishes for crevice-dwelling bats. Bats can easily find these water sources as they walk on the cage bottom without danger of drowning. Do not leave containers on cage bottoms with enough water that a sick bat could become soaked, exacerbating its condition. Foliage-roosting species will not eat or drink from containers placed on the bottom of cages. Small water containers can be made from film canisters or similarly sized plastic containers and hung throughout roosting cages for crevice-dwelling species. Simply cut the canister (or container) and staple a piece of Velcro (the soft side) of the canister. Small strips of Velcro (the rough side) can then be stapled to the inside of the cage so that the film canisters can be attached to various places throughout the cage.

Notice that these containers are small enough that the bat's entire body could not fit inside. These water dishes may also be attached by Velcro to the upper frame of small netted cages for foliage-roosting bats, and to the upper frame of flight cages for both crevice-dwelling and foliage-roosting species.

Small bird cage trays (i.e., coop cups) may also be hung from the netting of flight cages for foliage-roosting species. These trays should be filled

with marbles, however, before adding water. This precaution ensures that bats will not fall into dishes and inadvertently aspirate water. Some bats may need to have water dishes held to their mouths before and after being fed because they do not seem to drink from water trays on their own. This is sometimes the case with red bats (*L. borealis*). Simply hold the water tray beneath them as they hang from the cage top.

Avimin and Avitron are liquid mineral and vitamin supplements that should be added to the drinking water for adults, unless these supplements are fed to bats in the blended food mixtures described on previous pages, in which case no supplements need to be added to the drinking water. Because these supplements can leave sticky residues in water dishes, it is preferable that they be added to food mixtures. (Water dishes should be cleaned with soap and water daily.) Bats that are fed blended food mixtures only once or twice a week should have Avimin and Avitron added to their drinking water only on the days that they do not receive the blended foods. These water mixtures should be prepared fresh each day. Water dishes should be cleaned and refilled twice a day. The vitamin and mineral supplements should be mixed in the drinking water each day, and then the same batch of mixture used to fill water dishes that morning, and again that evening. To prepare the mixture, add 1 drop (0.05 ml) of Avitron and 2 drops (0.1 ml) of Avimin to every 25 ml of water (Barnard 1995). In addition, Linatone, a fatty acid supplement, can be added to the drinking water once a week (Barnard 1995) unless given orally as described above. Add 1 drop (0.05 ml) of Linatone to every 25 ml of water (once a week only), although it is preferable that Linatone is given orally (see above) as this supplement also leaves a sticky residue in water dishes. Caution should be exercised in the use of vitamin and mineral supplements (i.e., do not exceed recommended dosages given in this manual). Overdoses have resulted in a variety of medical conditions, including bone and skin conditions.



During feedings, bat pups should be placed on a heating pad that has been covered with two layers of cloth (i.e., there should be two layers of cloth between the pup and the heating pad. (pictured: *Tadarida brasiliensis*)

FEEDING INFANTS AND JUVENILES

Feeding Orphaned Pups

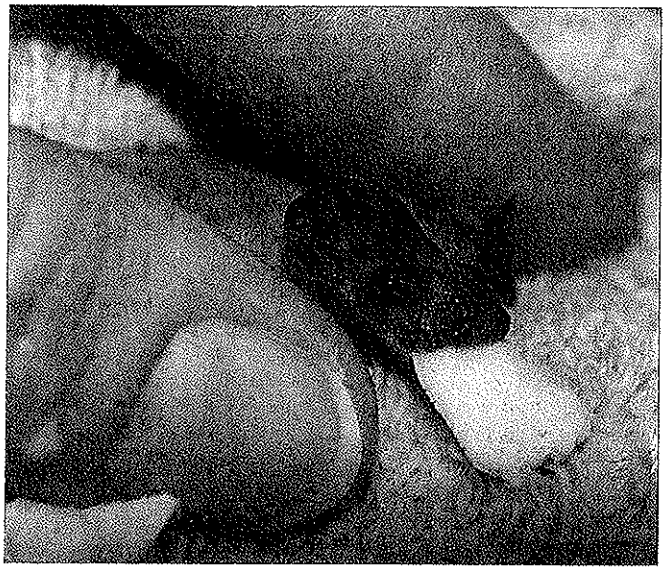
Pups that are naked (furless), or that have tiny, sharp-hooked milk teeth should be fed milk formulas. Pups that are fully furred but that have elongated joints in the wings are juveniles. Juveniles that are underweight should be fed the intermediate mealworm diet given below. Juveniles that are not underweight should be feed the mealworm diets for adult bats, which includes both live mealworms and blended mixtures. Depending on the species, most bats should no longer be fed milk formulas by the time they're flying (four to six weeks of age, although some species are flying by three weeks of age). In general, adults should not be fed milk formulas, although milk formulas may be added to blended mealworm mixtures.

Orphans that have just arrived at your facility should be thoroughly examined and weighed. Bat pups need to be kept warm (typically between 90° to 100° Fahrenheit—32.2° to 37.8° Celsius) and so should be placed on a warm, clean cloth on top of a heating pad during examinations. Emaciated infants should be rehydrated and fed as quickly as possible. Because orphans are typically dehydrated when received, they may need to be injected with Lactated Ringers solution for at least the first two to three days.

The natural composition of a mother's milk varies from species to species. Within a species, the composition may also vary throughout the course of lactation. Many commercial milk replacers developed for other animals do not appear to provide the necessary nutritional requirements for bat pups. The authors specifically recommend powdered puppy formulas, either Mother's Helper or Zoologic 33/40. Powdered formulas allow a caretaker to prepare small amounts at a time. Liquid formulas will spoil long before they can be used up. (Do not use milk formulas intended for kittens. Bat pups seldom survive on these formulas.) Although pups may survive on milk formulas other than those recommended above, they commonly suffer vitamin deficiencies that result in developmental abnormalities. Add 1 drop (0.05 ml) of Avitron liquid vitamin supplement and 2 drops (0.1 ml) of Avimin liquid mineral supplement to every 35 ml of mixed formula. A coffee warmer can be used for keeping formula warm during feeding sessions, although formula will scald if the container is placed directly on the warmer. Instead, place the container of formula in a shallow bowl of warm water first and then place the bowl of water on the coffee warmer. You must constantly check the temperature of the formula by dropping a few drops on to your inner wrist. Bat pups typically prefer milk at a temperature that feels

(Right) To encourage troublesome pups to accept applicator tips for nursing, immobilize them by placing an index finger on one side of the face, and a thumb on the opposite side. (Pictured: *Tadarida brasiliensis*)

(Below) Emaciated orphans.





somewhat warmer (115° to 120° Fahrenheit—46.1° to 48.3° Celsius) on the wrist than that considered appropriate for human infants. Infant bats will usually refuse liquids that are not warm enough. If the formula becomes too hot, remove it from the heated water for a few moments until it cools sufficiently. (Feeding implements that become clogged with thickened formula during feeding can easily be rinsed clean in the warmed water.)

Various implements have been used to feed bat pups, including syringes, medicine-droppers, rubber catheters, and foam sponges. Some of the species in the family Vespertilionidae will "lap" fluids and so may accept a "drop-by-drop" method of feeding. Begin by holding the pup upside down. This is extremely important because infants are likely to aspirate fluid into the lungs when learning to lap formula if the head is not kept lower than the rest of the body. Placing a small amount of warm formula onto the pup's mouth typically induces a lapping response. Continue to dispense the formula drop-by-drop as the pup drinks.

Bat species in the family Molossidae will not lap liquids. It is therefore critical that milk is offered to these pups through soft, high-density foam sponges. White foam eyeshadow applicator tips (removed from the wand) are generally used for this purpose (Lollar 1994). Again, hold the pup upside down when feeding. Use a pair of tweezers or forceps to dip the foam sponge into warmed formula. Allow the tip to remain in the formula for several seconds to absorb heat as well, then dab the excess off on a paper towel. Test the temperature of the tip against the inside of your wrist. Gently stroke the pup's mouth with the warmed applicator tip, clicking your tongue on the roof of the mouth as you do so. These soft clicking sounds mimic the directional calls some mother bats use to identify their pups. Occasionally, a pup will immediately make a clicking sound in return, grab the sponge with its mouth and start sucking. You will no longer need to hold onto the sponge at this point because the pup will keep it clamped between its teeth. When the pup begins sucking, dispense warmed formula one drop at a time onto the foam sponge with a syringe or medicine dropper. Be careful to drop the formula onto the portion of the applicator tip that is furthest from the pup's face, allowing any excess milk to drip away from the infant.

Pups should be kept warm during the entire feeding process. (Never expose a pup to drafts from fans or air conditioners.) Foliage-roosting pups should be fed formula while hand held. Once crevice-dwelling pups have started drinking formula while being hand held, they can be placed abdomen down on a heated surface and fed as shown on page 21. Heating pads can be utilized for this purpose. The medium setting appears to be the most comfortable for crevice-dwelling pups. (There should be two layers of cloth between the pup and the heating pad.) Fold a clean, absorbent cloth on top of the pad to absorb excess formula during each feeding. Allow the cloth to warm before placing the pup into the folds for feeding. Cover the pup's body with the cloth, leaving only the head exposed. Use a clean, dry cloth for each feeding.

Some pups will need coaxing to nurse from applicator tips. Place the

pup horizontally within the folds of a warmed cloth on a heating pad so that only its head is exposed. Position your index finger on one side of the pup's face, and your thumb on the opposite side. While immobilizing the pup's head with one hand, use your other hand to dip an applicator tip into warmed formula. Click to the pup and stroke its mouth in a back-and-forth motion with the tip several times. Then gently push the tip into the pup's mouth and maneuver it so that a portion of the tip is anchored between the pup's teeth. Continue to hold the head immobile to prevent the pup from shaking its head and spitting out the tip. Slowly dispense warmed formula onto the tip with the syringe or medicine dropper so that a small amount trickles inside the pup's mouth. Make clicking sounds as described above during this procedure. Continue dispensing warmed formula to the applicator tip. Most pups will either begin nursing (i.e., sucking), or at least swallow some of the formula. The tragus will move up and down slightly each time the pup swallows. If the pup begins nursing, stop dispensing formula until the tip is less saturated. As the pup continues to nurse, warm formula can be added to the tip one drop at a time. Some pups are difficult to feed and will manage to spit the applicator tip out of their mouth. Some will swallow a few drops of formula but will not begin sucking. These pups will eventually start nursing once they overcome initial fear of the handler. If you become frustrated, put the pup back in the incubator and try again once you feel more relaxed.

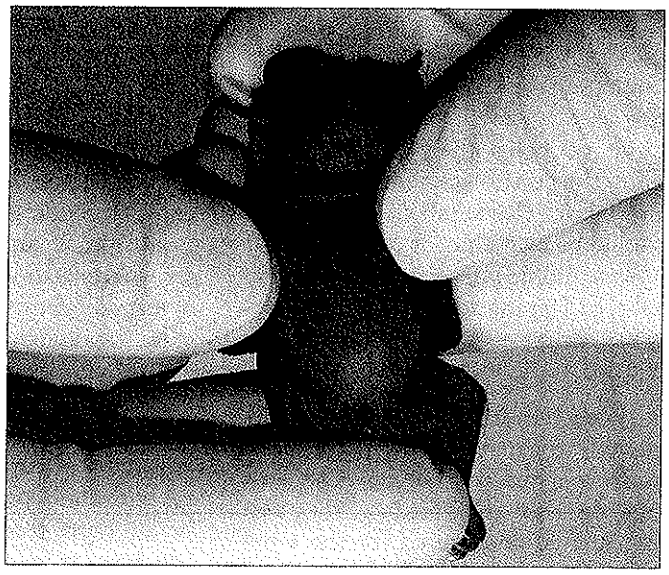
Some pups will discard the foam applicator tip as soon as they are full; others will hang onto the tip even after they have stopped drinking milk. Do not try to remove the tip from a pup's mouth. Any remaining formula in the tip may be accidentally forced down the pup's throat, causing it to aspirate. These pups will eventually discard the tip after being returned to the incubator. It is important to note that when some molossid pups are first rescued, they will immediately grasp the cloth or fabric that encloses them with their milk teeth, refusing to let go. Don't attempt to remove the pup's teeth from the fabric. Instead, carefully dispense warmed formula onto the cloth just below the pup's mouth and allow the pup to suck the formula from the fabric. (Remember, clean cloths should be available for each feeding.) Vespertilionid pups that do not readily take to lapping formulas, will sometimes accept milk from foam applicator tips as the molossids do.

Although it is preferable that disposable foam nursing tips (i.e., eye shadow applicators) be replaced with new ones for each feeding, they can be cleaned and reused for the same pup. Use tweezers or forceps to hold the tip under hot running water, and then place it in a small glass jar with a tight-fitting lid. Fill the jar with hot water and then add only a couple drops of a mild detergent; close the lid and shake vigorously for several seconds. Thoroughly rinse the tip again by holding it with tweezers or forceps under running water until suds no longer appear. Rinse the tip several more times (five or six—use clean water each time), using the "shaker" method described above. Place the tips on a paper towel to air dry. (Keep sink drainers in place while rinsing so you don't lose tips down the drain!)

Pups can aspirate fluids or choke on food while being hand fed. A bat may be choking if it suddenly gags and/or appears to gasp for breath during hand feeding. If this happens, *keep the bat upside down* with the abdomen against the palm of your hand and gently but firmly tap your finger on its back just below the shoulder blades in an attempt to force the substance from the airway. (Use no more pressure than you would use if you were drumming your fingertips on a tabletop.) Normal respiration can sometimes be restored immediately. If liquids or food particles actually enter the lungs, the bat is likely to develop pneumonia and will subsequently exhibit signs of respiratory distress, making soft squeaky, wheezy, or rattling sounds with each breath, often audible even without a stethoscope.

Because some infants are naked or sparsely furred for the first few weeks of life, milk can clearly be seen through the skin of the abdomen as it fills the stomach. It takes only a small amount of milk to fill the stomachs of most pups. Depending on the size, age, and condition of an infant, it will take anywhere from a few drops to 1.0 ml of formula per feeding. (Pups of most species should not be allowed to drink more than 1.0 ml per feeding.) Pups that have not yet attained adult weight should not be fed fewer than three to four times per day. Pups accepting 1.0 ml of formula per feeding should not be fed more than four times per day (once every six hours). Because it is not possible to measure exactly the amount of formula a pup has consumed when using foam applicator tips for feeding, it is necessary to rely on visual or tactile examination. The stomach should be slightly rounded, but not bulging with milk; a distended abdomen may indicate you have allowed the pup to drink too much.

Regardless of the species or the age of an infant, feeding schedules should be determined by the amount of time it takes for an individual pup's stomach to partially empty. To determine an infant's feeding schedule, first familiarize yourself with the look and feel of the pup's abdomen both before and after its first feeding. Take note of the visual appearance (milk in stomach) and how the abdomen feels when you gently palpate it with your finger (tactile examination). Note the time and check the pup each hour thereafter. Do not let the stomach empty completely. Keep enough formula in the pup at all times to keep the abdomen slightly rounded. Feed the pup again when the milk is nearly gone (i.e., when only a small amount is visible in its stomach). If the pup is already furred, milk will not be visible through the abdomen and you will need to rely solely on tactile examination. Note the time it took for the stomach to partially empty so that you can estimate when the next feeding will be due. Feeding infants too often may result in bloat (i.e., distention of



Milk is visible in a pup's stomach through the skin of the abdomen. This pup's stomach is full.

the stomach and severe abdominal distress), which commonly leads to death. Most pups will initially require feedings every four to six hours (four to six times a day) depending on their condition, although some pups' stomachs may empty in as little as two hours. Younger pups and pups that are emaciated will need to be fed more often than older pups that are healthy. In other words, as pups grow older and become healthier, the number of feedings per day will decrease.

For severely emaciated pups, add 1.0 ml of Stat™ High Calorie Liquid Diet (5085 kcal/kg) to each 9 ml of formula for every other feeding for three to five days. (It should be mixed fresh for each feeding.) This supplement dramatically increases the weight of starving pups within five to seven days.

French has found that red bat pups (*L. borealis*) are not likely to survive on milk formulas alone. Instead, they must be fed blended food mixtures consisting of mealworms and banana and veal baby foods. This mixture must be particularly smooth (i.e., no lumps) for pups because they do not have the kind of teeth needed for chewing. For pups weighting 5 g or less, the blended mealworm mixture can be mixed with an equal volume of reconstituted milk formula (Mother's Helper). Mix one part powdered milk with at least two parts water. Mix this with an equal volume of the blended mealworm/veal/banana preparation and warm it gently in a bowl of water. Do not overheat or the mixture will clump. Pups should be offered reconstituted milk formula after being fed the blended food. Older red bat pups weighing nearly 7 g or more should be fed the veal baby food, banana baby food, and Nutri-Cal preparation described on page 17. These pups should also be offered mealworm viscera (or whole mealworms if they will accept them). Offer these larger pups reconstituted milk formula after feedings and then offer them water. Attach water trays made from film canisters (as described above) in cages so that the water is at head level for the older pups. Do not put water trays in cages with red bat pups weighing less than 5 g.

French feeds red bat pups blended food mixtures with a 1 ml syringe and the milk formula with a 1 ml syringe attached to a soft, open end (not funnel connector) red rubber tubing called a "feeding tube and urethral catheter for cats." The one made by Sovereign with the open end (not a funnel tip) works well and should be purchased in a "size 8 French." This brand wears well and bats readily accept the soft tip as a feeding instrument. Measure off approximately 3" (7 cm) from the end of the tubing with the largest diameter and cut. Attach this 3" section to the tip of a syringe. Mix the powered milk formula as described above. Warm the formula and then offer it to the pup through the tip of the catheter.

Red bat pups generally require feedings once every four hours until they reach a weight of 8 g or more, although the 2 g to 5 g pups may require feedings once every two to three hours. Pups that are consuming as much as 1 ml of blended food can be fed once every six hours (two- or three-month old juveniles can thus eat as much as 4 ml per day). Only when they are eating whole mealworms live can they be reduced to twice a day feedings. Red bat pups should be stimulated after each feeding as described on the following page to initiate urination/defecation. If not stimulated, these pups tend to urinate and defecate on themselves. If not cleaned and dried immediately, they are likely to become hypothermic and die.

Remember, infants should always be positioned so that they are lying on their abdomens and tilted so that the head is slightly lower than the rest of the body to prevent aspiration of fluids. Pups will either lap the formula one drop at a time from the tip, or will take the tip directly inside the mouth and chew on it as the formula is dispensed one drop at a time. Rubber catheters used for feeding pups should be rinsed in a mild detergent, submersed in a pan of boiling water for 15 minutes, and then placed on a paper towel and allowed to air dry before being reused. Keep feeding tubes clean and warmed—they become stiff and less flexible if chilled. Some pups will lap formulas from tiny bird trays.

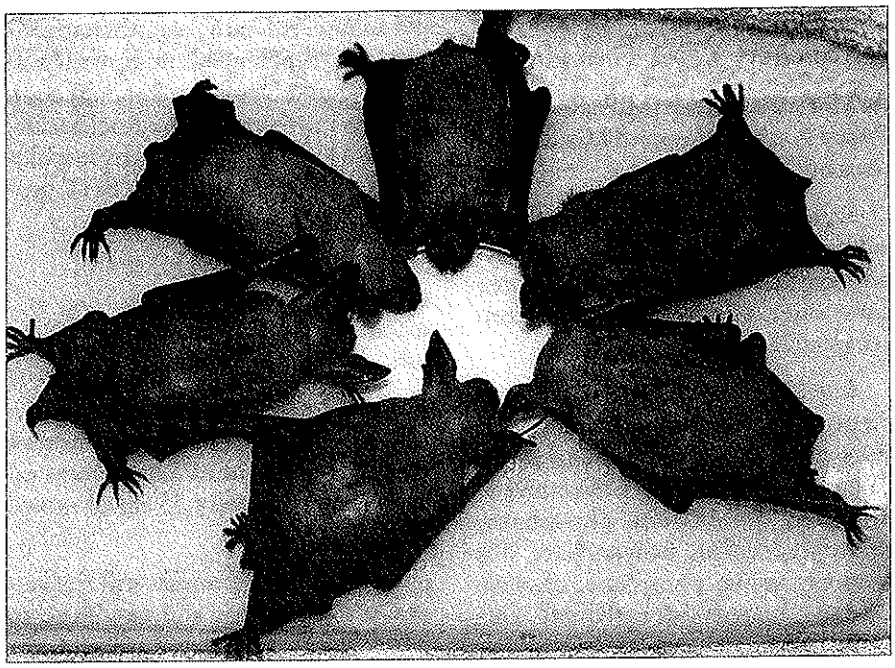
Rehabilitators sometimes receive large numbers of pups for various reasons, including exclusions of maternity colonies from buildings. Caring for large numbers of pups requires some modifications in care procedures. Over the course of several days, some pups can be conditioned to being fed in groups. Pups that lap formula can be taught to drink from shallow dishes. Hold a pup over a shallow dish of

warmed formula while feeding it from a catheter as described above. While the pup is lapping, move the catheter away and carefully lower the pup until it drinks from the shallow dish. Be careful not to immerse the bat's nose in the formula. Once the entire group of pups has been individually taught to lap formula from a shallow dish, they can be fed in groups on top of a heating pad. You will need a thin sponge measuring approximately 4" by 6" (10 cm x 15 cm). Place a stainless steel automotive freeze plug (see Water Routines section) in the center of the sponge, trace around its edges, and cut out the circle. (Place an absorbent cloth on top of a heating pad (on medium setting). Then place the sponge on top of the cloth and put the freeze plug into the hole. Fill the plug with warmed formula. Each pup should be placed on the sponge one at a time, covered, and should be drinking formula before the next pup is put in position.

Molossid pups can also be fed from applicator tips (as described above) in groups. Each pup should be nursing from its foam tip and covered before the next pup is put into position.

After feedings, the pup's face and chin should be gently wiped clean with a foam sponge dipped in warm water. Some pups, such as red bats (*L. borealis*), will also need to be stimulated to initiate urination/defecation. Gently dab at the pup's anus and genitals with a soft, high-density foam make-up sponge or eye-make-up remover pad that has been dipped in warm water (cotton swabs are too rough for this purpose). Dab repeatedly until the pup urinates or defecates. Although newborn foliage-roosting species require stimulation, some of the crevice-dwelling species such as Mexican free-tailed bats (*T. brasiliensis*) do not.

You can begin weaning insectivorous pups off of milk formulas, depending upon the species, at three to six weeks of age. At this time, you may begin offering pups the intermediate level formula given below. The mixture should be fed to the pup through the tip of a 3 ml syringe. Again, infants should be positioned so that they are lying on their stomachs and tilted so that the head is slightly lower than the rest of the body.



Big brown bat (*Eptesicus fuscus*) pups are fed formula in a group. The pups and warmed formula are placed on a heating pad covered with two layers of cloth.

Infants may be fed 0.5 ml to 1.0 ml of the blended mixture per feeding and should be fed every six to eight hours (three to four times per day) at the most. At three to six weeks of age pups can typically be allowed to eat as much of the blended mealworm mixture as they will accept at each feeding, although small species (adult weight less than 10 g) should not be allowed to eat more than 0.75 ml per feeding, and medium-sized species (adult weight 10 g to 20 g) should not be allowed to eat more than 1.0 ml per feeding. Milk formulas can be offered after each feeding of blended mealworm mixtures. Although we caution against allowing significant abdominal distention in newborn pups, pups at this age will normally have somewhat distended abdomens following each feeding. Water can be offered after the pup has been offered milk formula. Infants should also be offered live mealworms (or mealworm viscera by pinching off one end of the worm and squeezing the insides into the pup's mouth) each day at this time until they begin accepting them. Pups that refuse blended mealworm mixtures are not ready to be weaned. Wait another week or two and try again. Pups readily accept blended food when they have reached the appropriate developmental stage.

Older bat pups (i.e., those that no longer require milk formulas) can also be raised successfully on the mixture of Nutri-Cal and veal and banana baby foods given on page 17. (Red bat pups [*L. borealis*] weighing 7 g or more thrive on this mixture.) Pups fed this mixture should also be fed live mealworms or mealworm viscera each day. Nutri-Cal should not be added to milk formulas because some nursing infants appear to be unable to digest this substance.

Rehabilitators should be aware of the fact that pups that are transferred from one caretaker to another (even for just a brief period of time) often do not survive.

NOTE: As a note of interest, French has received reports from individuals who have kept bat pups alive for a few days to several weeks (until they were passed onto bat rehabilitators) on human milk, including a big brown bat (*E. fuscus*) and a red bat (*L. borealis*).

Infant Bat Formulas

Introductory Level Formula

A. Mother's Helper (Milk Replacer) Puppy Formula Powered Mix distributed by Lambert Kay

Formulas should be mixed in ratios varying from about one to two parts powdered milk replacer to two to three parts water. Mix fresh daily and keep refrigerated.

Ingredients include: partially hydrogenated soybean oil, casein, dried skim milk, maltodextrin, egg yolks, calcium carbonate, d-methionine, l-arginine, dipotassium phosphate, monoglycerides, polysorbate 60, monopotassium phosphate, choline chloride, magnesium carbonate, calcium hydroxide, sodium hydroxide, magnesium sulfate, zinc sulfate, vitamin A palmitate, ferrous sulfate, manganese gluconate, vitamin E, copper sulfate, niacinamide, calcium pantothenate, cyanocobalamin, thiamine hydrochloride, riboflavin, pyridoxine hydrochloride, potassium iodide, folic acid, vitamin D_3.

crude protein:	min. 33.0%
crude fat:	min. 40.0%;
moisture:	max. 5.0%;
ash:	max. 6.0%

Add 1 drop (0.05 ml) of Avitron liquid vitamin supplement and 2 drops (0.1 ml) of Avimin, liquid mineral supplement to every 35 ml of mixed formula.

B. Zoologic 33/40

Formulas should be mixed in ratios varying from about one to two parts powdered milk replacer to two to three parts water. Mix fresh daily and keep refrigerated. Ingredients include: casein, dried skimmed milk, egg yolk, l-arginine, dimethionine, calcium carbonate precipitated, potassium phosphate monobasic, dried corn syrup, calcium hydroxide, salt, lecithin, monocalcium phosphate, sodium hydroxide, choline chloride, potassium chloride, magnesium carbonate, magnesium sulfate, vitamin A supplement, zinc sulfate, vitamin E supplement, iron sulfate, niacin supplement, copper sulfate, calcium pantothenate, vitamin B_{12} supplement, manganese sulfate, vitamin D supplement, folic acid, riboflavin, thiamine hydrochloride, calcium iodate, pyridoxina hydrochloride, (preserved with BHA, BHT, propyl gallate and citric acid).

crude protein:	min. 33%
crude fat:	min. 40%
moisture:	max. 5.0%
ash:	max. 6.0%

Add 1 drop (0.05 ml) of Avitron liquid vitamin supplement and 2 drops (0.1 ml) of Avimin liquid mineral supplement to every 35 ml of mixed formula.

NOTE: Milk formulas will thicken under refrigeration and may need to have water added for subsequent feedings. Add just enough water so that the mixture returns to its original consistency.

Intermediate Level Formula (Blended Mealworm Mixture)

2 cups (480 ml) frozen mealworms (*Tenebrio molitor* larvae)

5 oz (142 g) veal baby food

2.5 oz (71 g) banana baby food (no sugars/preservatives)

1 raw egg

Blend together in heavy-duty blender until mixture is smooth (consistency of an ice cream malt). The blended mixture may be poured into small baby-food jars and frozen. Defrost and warm mixture. Add 3 drops (0.15 ml) of Avimin, 3 drops (0.15 ml) of Avitron, and 1 drop (0.05 ml) of Linatone to each 2.5 oz (75 ml) of blended food. Do not overheat the mixture or it will lose moisture and its smooth consistency. Add two tablespoons (30 ml) of reconstituted Mother's Helper Puppy Formula to every 2.5 oz (75 ml) of blended food. (The powdered milk formula should be reconstituted by mixing two parts water to one part powdered milk formula.) Decrease the amount of reconstituted milk added to the mixture each day until none is being added after a period of one week. Do not reuse the mixture for more than two days after it has been defrosted.

Complications

Bloat

Overfeeding is one of the most common causes of death in bat pups. Bloat can result from overfeeding or from the absence of beneficial organisms that normally reside in a healthy pup's intestinal tract. Absence of beneficial flora may allow other organisms to grow out of control and result in a build-up of intestinal gas. Bloat is often a fatal condition. White or grayish feces often precede severe abdominal distention. Small bubbles are commonly visible in the stomach through the skin of the abdomen.

To treat this condition, Lollar administers 0.01 ml of simethicone for every gram of body weight every two hours (to relieve gas) and adds 0.02 ml of BeneBac gel (*Lactobacillus acidophilus*) preparation to each feeding until abdominal distention is relieved. (*L. acidophilus* may encourage growth of beneficial bacteria.) Patricia Winters treats the condition with Neomycin (Winters 1995). However, the authors recommend that bats treated with Neomycin not be released due to the potential for hearing loss.

Metabolic Bone Disease

Metabolic bone disease resulting from vitamin D deficiency is called rickets in young individuals and osteomalacia in adults. Metabolic bone diseases can also be caused by an inadequate intake or absorption of calcium. Fat is an important dietary component because it facilitates calcium uptake. On the other hand, an excess of fatty acids in other animals can interfere with calcium absorption by the intestine because of the formation of insoluble calcium soaps (Rasweiler 1977). An excess of phosphorous can also interfere with intestinal absorption of calcium. However, the results of some studies suggest that bats may function optimally at low levels of vitamin D and that mineral homeostasis in bats may not be regulated at the level of the intestine, but instead by a

highly efficient method of manipulating bone and teeth reservoirs (Kwiecinski, Chen, and Holick 1998).

The authors often observe signs of nutritional deficiency in bats. Orphaned pups that have survived for a number of days without sustenance or pups fed inappropriate diets are likely to develop growth abnormalities. Pups fed Mother's Helper or Zoologic 33/40 milk formulas that have had Avitron and Avimin liquid vitamin and mineral supplements added are unlikely to develop such abnormalities. Rapidly growing bones like the long bones of the arm are often affected by nutritional deficiencies. Common clinical signs are similar to those seen in other animals experiencing vitamin D or calcium deficiencies and can include swelling or curvature of the long bones of the wing, enlargement of the joints, muscular weakness, and neuro-muscular hyperirritability often progressing to tetanic spasms and sometimes convulsions (Rasweiler 1997). This condition is painful. In severe cases, a bat will rest with its wings slightly extended at all times and may experience intermittent spasms of the wings in particular. Bats with this condition will sometimes cry out when touched.

Treatment consists of use of appropriate formulas and vitamin and mineral supplements (including sterilized bone meal) as described in the feeding routines given in this chapter. While mild cases may be reversible in early stages, they cannot be reversed in later stages. For pups exhibiting these signs, add 1/4 teaspoon (1.25 ml) of sterilized bone meal powder to every 3 tablespoons (45 ml) of reconstituted milk formula until there is noticeable improvement, usually within one week. Juveniles and adults exhibiting these signs should not have sterilized bone meal powder added to their food. Instead, supplement feedings of the intermediate level formula with a thick paste of Mother's Helper milk formula. Allow the bat to eat as much of the paste as it will consume three times a day.

Do not continue to add sterilized bone meal to a bat's food after swelling has subsided. In other animals, an excess of calcium can cause problems by inhibiting intestinal absorption of phosphorous. In addition, too much calcium can result in excessive calcification of the bones. French added sterilized bone meal powder directly to blended food mixtures fed to 47 Mexican free-tailed bats (*T. brasiliensis*) that were not exhibiting signs of nutritional deficiency, for a period of one week. One half teaspoon (2.5 g) of bone meal was added to 2.5 fluid ounces (75 ml) of blended food. Bats were fed their normal dose of 1.0 ml to 2.0 ml of the mixture daily for the entire seven-day period. At the end of that period, mineralization of feces was apparent for all 47 of the bats (i.e., feces were white), and 9 bats developed bony lumps along the wings and/or legs. The condition appeared to result from an excess of a calcium/phosphorous supplement (i.e., bone meal). When bone meal was eliminated from the blended food mixture, the hard lumps disappeared. (Captive fruit-eating bats fed a diet deficient in protein, calcium, phosphorus, and vitamin D have been known to develop soft lumps on the forearms—Wilson 1988).

The diets recommended in this chapter appear to provide an appropriate calcium to phosphorous ratio and have proven successful in preventing conditions that result from either calcium deficiencies or excesses.

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To Purchase the Book
Send $45 plus shipping and handling ($5 in U.S. priority and via surface, international; $15 airmail) to Bat World Sanctuary at the above address.

NOTE: Book is also available through the IWRC Catalog.

BAT REHABILITATION WORKSHOP

The Bat World Sanctuary offers an annual summer workshop on rehabilitation of insectivorous bats that includes:

- The book, *Captive Care and Medical Reference for the Rehabilitation of Insectivorous Bats,* by Amanda Lollar and Barbara French, with contributions by Patricia Winters.
- Five days of hands-on training in bat rehabilitation.
- Lessons in health precautions, public protocol, caging, and diet including maintaining insect colonies and preparing blended mealworm mixtures.
- Handling and feeding of live crevice-dwelling and foliage-roosting species (both adults and baby bats).
- Lectures on basic anatomy, species identification, examination, troubleshooting, injection techniques and stabilizing both closed and open wing fractures on dead specimens.
- Daily rescues of both injured and orphaned bats that have been found by the public and from our Wild Sanctuary for Mexican free-tailed bats (*Tadarida brasiliensis Mexicana*).
- Rehabilitation and possible release of rescued bats.
- A Certificate of Completion in Basic Bat Rehabilitation.

Four Sessions in 2000:

3 July through 7 July

10 July through 14 July

17 July through 21 July

24 July through 28 July

Each session is limited to seven people. Workshops begin at 1 pm and end around 10 pm. Participants are responsible for their own food and transportation, however, some local motels have rooms with kitchenettes and are offering reduced rates for workshop attendants. Camping is also available at Lake Mineral Wells State Park, which is located about seven miles from Bat World.

Proof of pre-exposure rabies immunization and a signed waiver are required.

Cost:

$350, prepaid and nonrefundable.

$300 if *Captive Care and Medical Reference for the Rehabilitation of Insectivorous Bats* has previously been purchased.

For additional information contact sanctuary@batworld.org.

Bat World is a nonprofit, volunteer-run facility. 100% of the fees paid for our summer workshops go to maintain the sanctuaries and to provide food and medical supplies for the bats.

FROM THE LAST ISSUE...
CASE IX:

Too Much of a Good Thing?

The last Up For Discussion case raised such complex issues as human-wildlife conflicts, wildlife population "management," and habitat loss.

Sylvia Jackson was reflecting on her involvement with wildlife rehabilitation after a phone call with Helen Andrews. Many years earlier, she had helped two fourth-grade students reunite some "rescued" goslings with their mother. The experience had prompted her to call Helen, longtime bird rehabilitator, and begin volunteering. Sylvia was soon spending much of her time helping Helen. By the following spring, Sylvia had her own state and federal rehabilitation permits. She specialized in waterfowl, but accepted a limited number of injured adult songbirds.

Helen had just told her that she would no longer rehabilitate Canada geese. First, Helen said, much of their former local habitat had been developed: there was simply not enough good habitat left for them. Second, many of the geese had concentrated in a few areas and become nonmigratory, causing problems for people as well as for other waterfowl. Third, the state wildlife agency had recently begun trapping "problem" geese and releasing them on a lake ringed with hunters. The agency was reportedly considering taking future trapped geese to a "processing" plant where they would be destroyed. Helen was deeply disturbed by these actions. She decided to concentrate on rehabilitating other birds, for whom she felt she could make more of a difference.

This phone call had caused Sylvia to weigh what *she* should do regarding the Canada geese situation. She recently talked with local Audubon and Sierra Club chapters about the problem of habitat loss and degradation. The conservationists were also very concerned about these issues, as well as the pressure the geese were placing on other, more sensitive species. Sylvia was disturbed by the wildlife agency's actions, but appreciated that they were under pressure to "do something." She also worried about the many people who didn't like the geese droppings on their yard, park or golf course.

QUESTIONS FOR DISCUSSION:

1. What might rehabilitators do about the many species facing serious habitat loss?
2. Human-wildlife conflicts are becoming increasingly common, including impacts from concentrated wildlife populations. What might be done to help resolve such concerns?
3. Some species have adjusted to living around humans. In some cases, this has placed pressure on other, more sensitive species. What are the options for rehabilitators facing this situation?
4. Wildlife agencies have a responsibility to manage wildlife populations. If they feel that a particular population is becoming a threat to wildlife or habitat, drastic action may occur. How can rehabilitators participate in influencing wildlife policy and action?
5. What are some of the options for dealing with rapidly expanding nonmigratory Canada geese populations?

RESPONSES

I.

Scott Diehl
Wildlife Center Manager
Wisconsin Humane Society

The first thing that struck me about Sylvia and Helen's dilemma was sadness that Helen had decided to not continue to rehabilitate Canada geese, after she had clearly had an inspirational encounter with them years earlier and consequently become involved in wildlife rehabilitation. Particularly disappointing was the fact that at least to some degree, her decision may have been prompted by some false or incom-

Wildlife rehabilitators' interests range far beyond the direct care and handling of wildlife. Standards, ethics, wildlife policy, and community involvement are among the many issues that have a direct impact on the work rehabilitators do every day.

Since these issues benefit from broad public discussion, this section of the *Journal* is designed to offer a forum for wildlife rehabilitators to exchange ideas about challenging issues, which are "up for discussion" by you, our readers. Some of the cases may also be discussed at upcoming IWRC conferences.

The following cases will present either hypothetical situations or real-life problems facing active rehabilitators. Each case is followed with several questions for readers, and responses from people involved in wildlife rehabilitation. To encourage a free exchange of ideas, both attributed and anonymous responses are welcome.

Each issue of the *Journal* will publish a synopsis of the original case from the last issue, several responses, and a new case for your consideration. Please see page 29 for this issue's new case, and see the box on page 28 for details about how to participate.

Space may not be available for all responses submitted, and responses may be edited for space and clarity, but we will include as many responses as possible. We welcome your participation. Only with your help can this forum thrive.

NOTE: THE International Wildlife Rehabilitation Council makes no claims as to the accuracy or truth of the following cases, not does it endorse any particular answer or viewpoint.

plete information. For example, the idea that "The [state wildlife] agency had taken the geese and released them on a lake ringed with hunters the day before hunting season" is highly implausible. Canada goose "roundups" take place in the summer, when the birds molt their flight feathers and for a time are flightless and therefore relatively easy to capture. Hunting seasons take place months later when the birds are fully flighted.

Hyperbole is commonplace when "nuisance" wildlife issues are being discussed, and it's important to separate the wheat from the chaff. Now that's not to discount the idea that Canada geese may indeed be overpopulated in this area, and that local wildlife managers may be planning or conducting lethal control methods. But time and again I've seen influential, and often uninformed, people whose lawns are being pooped on or whose flowers are being munched dominate and set the tone for local discussion of these issues. Rumors and half-truths are fostered in emotionally charged conflicts, even philosophical ones.

While Sylvia has gathered some information from local conservationists, I would encourage her and Helen to dig a little deeper to find more solid information about their local goose problems. Are the geese truly *biologically* overpopulated, or "simply" *culturally* overpopulated? Biologically overpopulated means that the birds are in such numbers that they are degrading habitat for themselves and other species, while culturally overpopulated means their presence is causing nuisance problems for humans. A single bird pooping on someone's lawn may qualify the species as being culturally overpopulated in that person's mind. I think it's important to ask concerned parties to list the types and numbers of problems they are aware of: this helps separate emotion from fact and can be helpful in tracking effectiveness if management techniques are implemented.

Of course, we rehabilitators know that we need to be defenders of biodiversity as well as defenders of humane, compassionate treatment for individual creatures. This is not always an easy balancing act. When a species is overpopulated by any measure it may be necessary to implement management strategies to alleviate conflicts and balance their numbers with their habitat and with other species. To my knowledge, there are basically four ways that wildlife managers can affect a wildlife population: modify habitat, increase emigration/decrease immigration, decrease reproduction, and increase mortality. I think it is unfortunate that some people, including some wildlife management agencies, almost automatically select "increase mortality" (i.e., goose "roundup" and slaughter) as their management technique of choice.

I encourage Sylvia and Helen to direct their local officials and wildlife managers to nonlethal methods such as habitat modification, repellents, and scare tactics to alleviate their goose problems. The Humane Society of the United States* is an excellent resource for this information.

If properly implemented, nonlethal methods prove to be ineffective in achieving reasonably set management goals, population control through egg-treatment can be attempted. Egg treatment involves addling ("shaking") eggs or painting them with mineral oil early in their development. (I personally dislike the concept of addling.) Either process stops the development of the embryo. Egg treatment requires a federal permit, and while it is technically a "lethal" technique, it at least spares the lives of sentient birds.

I'd encourage Sylvia to take another look at how her rehabilitation efforts for Canada geese will realistically affect the local goose population and the problems associated with it.

I can empathize with Helen's distress over the idea that a bird or birds she has fought to rehabilitate may end up in a processing plant, but rather than withholding care from individual creatures presented to us, I suggest the better path is for us to the best of our ability become informed about wildlife management issues locally and globally, and become as involved and proactive as we can. If you are able, offer help to your local wildlife managers as a resource for information about, or even deployment of, nonlethal wildlife management efforts. Encourage and support the development of nonlethal population control methods such as immunocontraception for urban wildlife such as deer and geese. Encourage urban renewal to stem the tide of habitat destruction surrounding cities. Advocate the development of urban greenspace to make additional living space for wildlife. Support biodiversity, while at the same time compassionately attending to the needs of individual animals.

**Humane Society of the United States, Wildlife Department, 2100 L Street, NW, Washington, D.C. 20037. www.hsus.org*

RESPONSES

II.

Christopher Panton, PhD
Member, Lindsay Wildlife Museum
Member, Wildcare
Walnut Creek and San Rafael, California

Alas, poor Sylvia. Her admirably well-intentioned rehabiltator's world has been rocked by the intrusion of a major social and political issue. Worse, this particular issue is arguably the most fundamental of all those currently facing wildlife, and therefore, by definition, of greatest importance to rehabilitation. The issue is, of course, the evolutionary changes induced in wildlife by habitat loss due to human depredations. Changes include the extirpation of species that cannot adapt to the presence of humans and their baggage; the adaptation of other species to human presence—a significant competitive advantage over nonadaptive species; the increased interaction between humans and wildlife as a result of successful species adaptations to human's increasingly greedy intrusions. These will all impact Sylvia's workload and her ability to manage her rehabilitation activity.

So what should Sylvia do? She has two options. The better one is to change nothing—to go on rehabilitat-

ing her animals with the same ideals that she started out with, and to make no concessions to those who would urge her to curtail some of her activities. Once she begins to discriminate against species that she would normally help, she becomes a part of the problem, and not a part of its solution. In fact, if she could even marginally expand her activities, then so much the better. Why should she take this approach? It is not simply because she is bound by the powerful moral obligation that she has already established between herself and the community—a community that has come to expect that its wildlife "hospital" will treat a patient with care and compassion, and without being judgmental about the patient's qualifications. Moral obligations notwithstanding, the main reason Sylvia should not change her approach is the reason rehabilitation exists in the first place.

Much has been written on this subject, ranging from the ego-buzz of helping wildlife to the physical satisfaction of seeing a patient recover and released. However, these necessary motivations do not constitute the basic raison d'etre. Like any business or service, rehabilitation exists first and foremost for one true purpose: to serve its primary customer, namely, the public. Those worthy souls and their families, friends, neighbours, and colleagues who probably give wildlife very few second thoughts, yet who will go to remarkable lengths to bring sick, injured, and orphaned animals to rehabilitation, thereby revealing a depth of caring and compassion far, far beyond the expected. Simply put, without the public, there would be no rehabilitation. The role model that Sylvia's work represents is of life-and-death importance because it is the public that must be rallied to support societal efforts to deal sensibly and compassionately with the fundamental issues that are impacting Sylvia in the first place. And if Sylvia begins to discriminate against her former patients, her work will become a regressive role model, teaching the public that they, too, can pick and choose which animals they'll tolerate and that they, too, can eliminate "nuisance" species as the whim takes them.

Staying involved is Sylvia's best option. In this, not only can she implement the salutary standards of the better option, outlined above, but she can also actively take up the causes of habitat loss and the mindless killing of species that do not fit somebody's personal view of the way in which wildlife should be structured and should behave.

This additional activty is a two-edged sword as there are only 24 hours in a typical rehabilitator's day: the added activist role can be a distraction. However, to be effective, Sylvia does not have to lobby in Washington, D.C. She simply has to tell the truth to all those she meets, particularly within the general public. This is not as easy as it sounds. First, the real truth is not always apparent, and second, Sylvia, like many of her volunteer counterparts, was probably not trained in science. So she has only an untrained and vauge awareness of the questions to ask in order to learn the truth. Tempting as it is to use her otherwise invaluable experience in waterfowl rehabilitation as a proxy for broad wildlife expertise, when an analytical truth is beyond her scope, she should remain silent. If she does not, she will only add to the already prolific and counterproductive mythology already concreted into wildlife management today.

Clearly Sylvia's case is a metaphor for all rehabilitators—not just individuals, but also organizations. Again for the sake of the public, the IWRC and the NWRA need to develop a common, consistent policy toward these issues, based on sound science, compassion, and common sense. Possible solutions to habitat loss and possible alternatives to the killing of "irritant" species do exist. Institutionally, a single voice needs to raise them in public debate and to fight for them. Simultaneously, misinformation that is currently being thoughtlessly passed on by unsuspecting rehabilitators needs to be purged by a rigorous internal education program of ethical standards as high as those that the IWRC and the NWRA apply to the treatment of animals.

At the present time, no wildlife organization in the United States has a leadership role on these most fundamental issues that are beginning to impact Sylvia's world. Work is conducted in piecemeal fashion by the many organizations involved; liasons and joint efforts ebb and flow by project. Worse, prominent groups such as the Nature Conservancy and the Audubon Society are tainted with the hypocrisy of their own killing agendas—a stance that must be viewed by the public as condescending at best.

This could be the time when rehabilitation finally comes of age and provides a leadership role on these issues, demonstrating to government agencies, other wildlife groups, and the public a binding commitment to wildlife through word as well as deed. Any alternative is unthinkable. Without a sustained, consistent mitigating counterforce, habitat loss—the root cause of all of wildlife's major problems—will bulldoze on, largely unchecked. In addition, the killing of species deemed undesirable for one flimsy reason or another will inevitably escalate. And in a few decades there will be virtually nothing left to rehabilitate.

JOIN THE CONVERSATION

On the following page, the *Journal* presents a new case. We invite you to read the case and consider the questions at the end. The cases are designed to raise complex issues, leaving plenty of room for varied or even contradictory perspectives. To join the conversation, send your response by 1 May 2000 to Attn: Up for Discussion, IWRC, 4437 Central Place, B-4, Suisun, CA, 94585-1633, USA, or via email to iwrc@inreach.com.

The *Journal* also welcomes suggestions for topics to feature in future issues.

CASE X:

Managing Risks

Emily walked into the house grinning. She'd had a great afternoon at the wildlife rehabilitation center. The dinner that her husband, Dennis, had cooking on the stove smelled great and was almost ready to eat. Their new three-month-old puppy was tugging on an old sock with their twelve-year-old son.

Emily started telling them about her day. As they knew, she had really enjoyed feeding the orphaned baby birds and squirrels the last two summers, but was also glad the numbers of new wild animals arriving had slowed down. Emily got a lot of satisfaction from working with wildlife and had been thinking about applying for her own rehabilitation license.

Today, the staff and volunteers had attended a training on rehabilitating mid-sized mammals and then had a planning meeting. They had discussed some possible changes in next year's approach to rehabilitating foxes, skunks, and raccoons. During the last year, some raccoons and foxes with distemper and parvovirus had been admitted to the center. The center's strict quarantine procedures had prevented a major outbreak among the animals in rehabilitation; all staff and volunteers were committed to following careful procedures to avoid spread of disease.

Because of their desire to further reduce risks of diseases, many at today's meeting had felt it would be better for the mid-sized mammal orphans to be rehabilitated in smaller groups at home-based facilities, rather than keeping them at the rehabilitation center like the other small mammals and birds. The center staff was going to check the budget, but felt that they could provide most of the formula, medical support, and initial caging for young juvenile wildlife. The center would also help people get their sub-permittee rehabilitation permits, offer ongoing training, and refer animals to them.

Although not mandated by any government agency, the wildlife rehabilitation center's internal policy required all skunks, foxes, and raccoons be vaccinated for distemper, rabies, and parvovirus. The center offered to provide these vaccines at no charge to their sub-permittees. Also, since the state wildlife agency required pre-exposure rabies vaccines for anyone working with mid-sized mammals, the center agreed to arrange for sub-permittees to receive these vaccinations on-site so they wouldn't have to travel to the medical clinic.

Dennis said he had been learning about vaccines that day, too. He had taken the puppy for his vaccination after work. While waiting at the veterinarian's office, he had gotten into a discussion with another guardian who had brought his dog in to be treated for a negative reaction to a vaccination.

When Dennis asked the veterinarian about the risks, the veterinarian acknowledged increasing concern about both the short- and long-term effects of vaccines. The veterinarian explained that most of the concern centered on an animal's "automatic" annual vaccinations rather than on a puppy's (or a kitten's) initial series of shots. In addition, he noted, the law required a rabies vaccination for all dogs, although at three months, Dennis's puppy was still too young to be given one. Thinking this over quickly, Dennis consented for the puppy to get his second vaccination in the series. The veterinarian gave Dennis some reading material concerning vaccines and vaccinations.

After dinner, Emily and Dennis read the vet's materials and looked up vaccinations on several Internet sites. The more they read, the more questions and concerns they had about vaccines. At the same time, they were concerned that *not* having vaccines could increase risks. The subject of vaccines was clearly much more complex and controversial than either of them had ever realized.

Emily really wanted to get a sub-permittee rehabilitation license and begin home-based rehabilitation. She thought it would be interesting to work with mid-sized mammal orphans and appreciated that the rehabilitation center would help provide the formula, caging, and so forth to get her started at a minimum cost. However, she was starting to understand why some people didn't like the requirement of vaccinating the mid-sized wild mammals and was not sure she wanted to do it. Maybe she could get the center to consider an alternative policy. She was also somewhat apprehensive about the risks of the pre-exposure rabies vaccine for herself.

Dennis wondered if having wildlife in the house could result in exposure of their son or puppy to diseases or parasites, even if wildlife was generally kept in a separate rehabilitation room in the basement. Dennis questioned if Emily had enough time to rehabilitate wildlife at their home even with her flexible part-time work schedule. They decided to talk more in the next week before any decisions were made and Emily returned to the rehabilitation center.

QUESTIONS FOR DISCUSSION:

1. **How do you feel about the decision to vaccinate wildlife in rehabilitation? Why? What factors do you consider part of the decision?**
2. **Some state agencies as well as rehabilitation facilities require pre-exposure rabies vaccines for people working with rabies vector species. How do you feel about pre-exposure vaccines being required for rehabilitators working with rabies vector species?**
3. **By its nature, wildlife rehabilitation includes some risks. Reducing and managing risks is critical, particularly with species that involve higher risks. What are some decisions (e.g., species to rehabilitate) and practices (e.g., cage location, sanitation) considered to used by home-based rehabilitators to reduce risks?**

CALL FOR PARTICIPANTS

STRAIT OF GIBRALTAR MIGRATION MONITORING SCHEME (PROGRAMA MIGRES)

The Strait of Gibraltar is the most important "bottleneck" for central and western European migratory birds crossing the Mediterranean Sea to and from their wintering quarters in Africa. With a maximum width of just 14 km, the Strait is especially important for soaring birds, which, during the fall migration, concentrate by the hundreds of thousands in the area of Tarifa, waiting for suitable winds to allow their crossing.

In 1997 the Regional Government of Andalucía funded a five-year program to monitor this migration. Between mid-July and mid-October observatories are placed along the Spanish migration front of the Strait. Volunteer monitors conduct bird counts from these watch-sites. At the end of each day, volunteers meet with fieldwork coordinators to check the data and resolve any questions about the birds' behavior.

14-day stays are required from 20 July to 19 October. Depending on the number of participants, volunteers may be able to remain for a second two-week period. Shorter stays (minimum five days) are possible from 10 October through 5 November. This year's observation periods are:

- 20 July - 2 August
- 2–15 August
- 15–28 August
- 28 August–10 September
- 10–23 September
- 23 September–6 October
- 6–19 October

Volunteers will be selected based on their experience, availability, and knowledge of the area (this is not essential). A $40 registration fee, payable upon acceptance into the program, is required. Registration includes 14 days accommodations and full board in a youth hostel; one day of alternative formative activities (e.g., scientific bird ringing); materials and daily transportation to fieldwork; divulgative materials; insurance; certificate of participation; and a report of the 2000 campaign.

To apply, send a completed preregistration form by 15 May 2000 to: Programa Migres, Centro de Visitantes "Huerta Grande," Ctra. N-340. El Pelayo, 11390 Algeciras (Cádiz), Spain; phone/fax: +34 956 67 91 58; email: P.MIGRES@TELELINE.ES

Confirmation, travel directions, and other pertinent information will be sent by June.

. . . . . .

Preregistration Form: PROGRAMA MIGRES

Last name: First name:

Date and place of birth: Passport no.:

Full address:

Phone no: Fax no: Email:

Dates of stay in order of preference:

1. 2. 3.

Profession/Student:

Previous participant in Programa MIGRES campaigns?

Yes: Which one?

No: (Please enclose a brief ornithological curriculum: expertise, skills, etc.)

Why would you like to joint PROGRAMA MIGRES?

I know and accept the conditions for joining MIGRES 2000.

Signature:

Avian Medicine and Surgery

Edited by Robert B. Altman, Susan L. Clubb, Gerry M. Dorrestein, and Kathy Quesenberry

Philadelphia: W. B. Saunders Company, 1997
1,070 pp; $105

Reviewed by Patrice N. Klein

This book serves as a comprehensive reference text in basic aviculture, avian medicine, surgery, nutrition, and clinicopathology. It was written primarily for a veterinary audience, but aviculturists, biologists, and wildlife rehabilitators will find many chapters selectively useful apropos to their specific needs. The book is more than 1,000 pages in length, divided into nine sections and 58 chapters. A color plate section in the front of the book contains an exceptional collection of 121 color plates of figures illustrating topics discussed in sequential chapters. The quality of these plates and the examples provided are by far superior to those seen in other publications. Appendices at the end of the book include useful reference tables of clinical pathology, taxonomic information, and conversion charts.

The first section and its associated chapters discuss basic aviculture and avian biology, and include sections on normal anatomy, physiology, reproduction, embryology (incubation and hatching), husbandry, and nutrition. Chapters describing artificial incubation, caging, and flock health management are particularly pragmatic. In general the references cited at the end of authored chapters are relatively current to the mid- to late 1990s, with the exceptions of the chapters on nutrition and incubation/hatching, which list quite dated reference literature. Considering the advances in nutrition and artificial incubation techniques in the past decade, it would have been prudent to have updated those chapters to more current practice methods.

Sections 2 though 5 include chapters on clinical pathology, radiology, cytology, and other diagnostic techniques; infectious and noninfectious diseases; pathology; and therapeutics. The chapters on diagnostic techniques contain excellent representative figures illustrating examples in hematology, cytology and radiology. The radiology figures are exceptional examples, with well identified anatomic structures in multiple-radiographic views.

Chapters on infectious diseases take a unique approach by describing microbiological pathogens in relation to the affected anatomical systems (respiratory, gastrointestinal, dermatological, etc.). The chapter on chlamydia is a thorough overview of the pathogen and associated diagnostic techniques. While the chapter on zoonotic diseases is a comprehensive, succinct overview, the chapter on differential diagnosis is merely a systematic listing of possible etiologies for a variety of common clinical signs that does not offer further guidance on differentiation, diagnosis, or treatment. The experienced clinician should already be familiar with the majority of these differentials, while the novice avian practitioner or nonveterinarian reader would find little usefulness for this unsubstantiated brevity. The chapters on noninfectious diseases illustrate an excellent systematic approach to understanding pathogenicity. Each disease is described following introductory explanations and illustrations of the normal anatomy and physiology of the system affected. Systems include gastrointestinal, nervous, endocrine, cardiovascular, musculoskeletal, and urogenital.

Sections 6 through 8 contain discussions of traditional veterinary medicine and surgery subjects. These are generally well written by recognized authors, but offer little new information that is not available in other reference publications. However, the chapters on beak injuries and repair are especially comprehensive, very practical, and well-illustrated by figures and diagrams. The use of orthotics in avian beak repair is limited to captive/pet bird situations, and not realistically applicable to wild bird rehabilitation. The anesthesia chapter includes good reference tables for monitoring and assessing anesthetic effects in avian patients. The bandaging chapter includes good reference tables for bandaging and splinting techniques, and well-illustrated diagrams. The emergency and critical care medicine chapter is a superficial review of a subject that has been described more thoroughly in other publications; it does, however, provide a succinct reference source.

Final sections in this book focus on several common classes of birds to include passarins, columbiformes, and raptors. Each dedicated chapter provides good reference tables for species specific diagnostics and therapeutics, but readers seeking more thorough, comprehensive information on each species are encouraged to find alternative published resources. The last two chapters are philosophical treatises on the human-avian bond, and grief and bereavement. Unfortunately, for such interesting topics the chapters are not given sufficient depth. References cited are dated considering the focus in human-animal bonding in the last 10 years.

Reference tables provided in the appendices are very extensive and include species specific reference ranges in hematology and clinical chemistry, but are limited to selected psittacines, ratites, and raptors. A list of scientific names of common psittacines is provided in one appendix. This is of limited use and value, and is obviously directed to pet avian practitioners. A better resource would be the ornithological texts and field guides that should be part of every personal library collection for those working in avian medicine and biology.

Overall, this book is a valuable reference text and should be among the

classic avian medicine literary references in one's personal library. The reader is advised that the book is slanted to pet bird medicine, rather than a more generic approach to avian medicine. Notwithstanding, the basic information provided often may be extrapolated to many classes of birds, both captive and free-ranging.

Patrice N. Klein, DVM, is wildlife veterinarian with the Humane Society of the United States.

First Aid for Birds: An Owner's Guide to a Happy Healthy Pet

By Julie Rach with Gary A. Gallerstein, DVM

New York: Howell Book House, 1999
126 pp; $12.95 U.S., $18.95 Canada

Reviewed by Marjorie J. Gibson

This is a comprehensive book written for the avian pet owner. While not meant specifically for wildlife rehabilitators, it could nonetheless be a great resource for them. Many rehabbers get the first call when a pet bird owner has questions regarding their bird's health or behavior. By recommending *First Aid for Birds*, you can confidently ease yourself out of the picture while giving the caller a sound and useful resource. Even wildlife rehabilitators who help avian species only infrequently will find this book useful.

The book is separated into two main sections, the first dealing with first aid, the second with preventive care. Helpful references to additional avian books and materials, bird-related websites, online discussion groups, relevant organizations, and suggestions for finding an avian veterinarian are included in the final chapter.

The first section begins by naming the parts of a bird. A practical definition and explanation of first aid follows. Questions concerning what constitutes an emergency and when to call your veterinarian are thoroughly discussed. The reader is also told what *not* to do when a bird appears ill. Signs of illness and specific problems are discussed at length; methods of restraining and transporting a bird are also described.

Section Two deals with preventive care. This is especially helpful to those who have no experience with avian species. The book addresses nutrition, as well as such important issues as household hazards. Attention is given to stress factors and the bird's need for time and attention from their handler or guardian. I was happy to see this subject addressed. Many older books ignored the important social needs of parrots and avian species. Observation is given a chapter of its own. This is another important addition and should not be underestimated when dealing with a pet bird. Wing clipping and other grooming tips conclude this section.

First Aid for Birds offers solid information and advice to pet owners. The authors have done a good job of presenting their material in a user-friendly manner. Photos of medical conditions and skills such as restraint methods assist written descriptions in allowing the book to be easily understood even by those not familiar with veterinary medicine. In my opinion, it should be mandatory reading for every potential new guardian before the bird becomes part of a household. This book is an ideal to be recommended with new bird purchases by reputable dealers.

Marjorie J. Gibson is president of the board of directors of the IWRC and director of the Raptor Education Group in Antigo, Wisconsin.

The Red Fox and Wildlife Rehabilitation

by Jeannie Lord

Fredonia, Wisc.: Pine View Wildlife Rescue Center, n.d.
4953 Highway H, Fredonia, WI 5302; pviewrc@aol.com
24 pp; cost unknown

Reviewed by PC Hanes

Jeannie Lord's booklet on red fox rehabilitation is characterized by some very striking photos of foxes at different ages and in different situations. However, the lack of captions leaves the reader at a loss as to where they fit into the text. There are also hand-drawn illustrations of a fox pouncing, presumably at prey; the internal anatomy of a fox; a fox skull; fox scat; and a fox pawprint. Unfortunately, these are so poorly reproduced as to be mostly useless. The author also reproduces radiographs to demonstrate how their use can reveal less obvious injuries, but she does not use any means to point out the injuries depicted. There are also some photos of dissected foxes with no captions or explanations, and photos of foxes who are apparently seriously ill—again without captions.

Lord makes a point in her preface and letter of intent that much of the material is anecdotal, following Jane Goodall's maxim that anecdotal information is important in ethological (behavioral) studies. However, this purports to be a rehabilitation manual, not an ethological study. As a result, Lord fails to distinguish between her personal observations and documented research findings, and uses only one or two citations from sources in the entire booklet. Although she lists references, there are no citations attributed to these sources, and many of the articles referred to are from "soft" publications, such as *Audubon* and *Smithsonian* Magazines. Nothing is cited from a first-hand research source or scientific journal.

There is good information in the booklet, but most of it is hidden among flowery prose. Lord's information on feeding of infants is sketchy at best, and buried in text that emphasizes nipple and bottle size more than feeding amounts and intervals based on weight and age. She does not supply a weight/feeding amount/interval chart where this information would be readily obtainable. Her recommendation for determining how much fluids to initially give a pup who is dehydrated to any degree is to use the calculations given in the NWRA Quick Reference. This is not very informative if one lacks the NWRA booklet. At the same time, someone faced with a seriously dehydrated cub will need the exact information available, and will not have time to make even simple calculations.

Housing recommendations seem out of proportion for active animals such as foxes. The recommended height for a fox cage in only 3–4 feet. All Nearctic foxes climb, and a 3–4 foot ceiling seems a bit low. Lord also recommends feeding kitten kibble as a weaning food, based on one article she read discussing convergent evolution between foxes and cats. She should discover what "convergent evolution" is, before she starts propagating information based on some apparent kinship between foxes and cats. Some rehabilitators use ferret chow for raising canids, to increase their protein intake, and there is evidence to support its use. Cat food, however, is not suitable for canids. Most wild felid rehabbers do not use cat kibble for bobcats and cougars at all.

The book does include good recommendations for enhancing the outdoor enclosure for foxes—e.g., leaves, rocks, tree branches, feathers—but this information has been published several times before. Most of the book's information is derivative, and Lord's lack of distinction between information from other sources, her own observations, and anecdotes makes it difficult to discern what is hard fact and what is her perception.

What Lord needs is a good editor, a spell-checker and a few lessons in technical writing if she wishes to put this publication forward for sale. The typographical errors, spelling and punctuation errors, and many sentences that make no sense at all detract from any valuable information she might be presenting. The publication is poorly organized for someone who needs a bit of specific information quickly, and an amateur taking foxes for the first time would not have a clear starting place to learn about fox rehabilitation other than from an experienced canid rehabber. Lord does suggest that fox rearing requires more than one telephone call or reading a single book. Perhaps she should read a few more books—specifically on technical writing and research—before she offers this publication to rehabilitators.

PC Hanes, MS (zoology), retired from rehabilitation work after 15 years working primarily with mammals. She is executive director of the Central Texas Wildlife Institute, and continues to write on rehabilitation issues.

Manual of Raptors, Pigeons and Waterfowl

General Editor: Peter H. Beynon; Scientific Editors: Neil A. Forbes and Nigel H. Harcourt-Brown

Ames: Iowa State University Press, 1996
364 pp, illus; $89.95

Reviewed by Sandi Leonard, DVM

This clear, well-organized book would be a useful addition to the library of any rehabilitator working with its subject birds. Part One covers general information applicable to many species in addition to the ones intended. Topics include physical examination, emergencies, nursing care, principles of therapy, diagnostics, anesthesia, radiology, postmortem exams, and zoonoses. Diagnostic chapters include cytology, biochemistry, hematology, and sampling techniques appropriate to each.

Parts Two, Three, and Four discuss husbandry, different problems by body system (skin, head, feet/legs, wings, respiratory, etc.); digestive upsets; neurological symptoms; breeding; and neonate husbandry for raptors, pigeons, and waterfowl, respectively. The section on pigeons includes additional information on performance. The appendices include formularies, United States wildlife law, and 1995 accredited laboratories in the United States and Canada.

This book is very well organized with good use of flow-charts, history forms, line drawings, and organizational charts that supplement the text and help clarify explanations. Color photos are used generously and effectively, sometimes even illustrating step-by-step how to perform a given technique. In many places the photos will aid in diagnosis of certain conditions.

Charts include a basic formulary, symptoms by body system, and an explanation of biochemical blood values. The chapter on anesthesia includes a very detailed chart with specific information on many common sedatives and anesthetic drugs. Line drawings of anatomy are included in several chapters and are clearly labeled. Another chart of common postmortem findings organized by body part is concise but quite complete. A glossary of falconry terms and list of falconry clubs in the United Kingdom is included. The waterfowl section features extensive information on the treatment of oiled birds. A few surgical techniques are also illustrated.

Overall, veterinarians serving wildlife rehabilitators and wildlife centers with well-trained staff or vol-

unteers should find this book extremely educational and helpful. It is perhaps more detailed and more technical than some people might wish, as a certain basic understanding of medical terminology and techniques is assumed. However, the information included is so complete that even a private rehabilitator might be pleased to own this book and willing to either use a medical dictionary to clarify terms when needed, or offer it as a reference to their veterinarian.

Sandi Leonard, DVM, is a relief veterinarian in Kansas City, Missouri, and a volunteer veterinarian at the Lakeside Nature Center, also in Kansas City.

BOOKS FOR CHILDREN

EYE TO EYE BOOKS:
Night Creatures
Wild Cats
Birds Of Prey

By Andrea Holden-Boone
Photographs by Simon M. Bell

Toronto: Somerville House Books, 1998
32 pp. (plus photo cards); $11.99 U.S., $14.95 Canada

Reviewed by Marjorie J. Gibson

This series of three attractive books is suggested for children ages eight through 12. They may, however, appeal to a broader range of ages due to the unique qualities in the books' presentation.

The books are each set up in a similar format. They feature attention-grabbing photos filled with animals in action. The pages themselves are backlit with colors that enhance each subject. The ring binder and sturdy glazed paper makes for a book that will withstand rugged use, either in a home or classroom setting.

One factor that distinguishes these books from other nature books is the inclusion of a 3-D viewer. Parents will appreciate the thoughtful addition of a pocket on the inside back cover, in which the viewer can be stored once it has been used.

The information included in each of the books is brief, but factual. The books strike a good balance, offering enough information to whet young minds with interesting facts and natural history tidbits, while stimulating the desire to know more. This approach may encourage further investigation of the subject.

Key words are highlighted in each section. These words are helpful not only as memory triggers for the animals discussed, but function as a vocabulary builder as well. A glossary is included, giving definition of the words to reinforce learning.

Each book includes an index—something not all children's books have. It is another learning tool, and is handy for the youngster who already has that particular study skill, but is looking for specific information.

Night Creatures, *Wild Cats*, and *Birds of Prey* comprise an interesting set of wildlife books with several "hooks" that will get youngsters involved not only in the wildlife topics, but in reading as well. The excellent use of visual aids, color accents, and borders that highlight material of special interest will stimulate even youngsters who find learning a chore. Any one of this series would make a good gift for a child. The stereo viewer adds an additional, special dimension.

The Icky Sticky Frog

By Dawn Bentley
Illustrated by Salina Yoon

Santa Monica, Calif.: Piggy Toes Publishers, 1999
16 pp; $9.95

Reviewed by Marjorie J. Gibson

This is a book for youngsters aged three to eight that is sure to become a favorite for children and environmental teachers.

The cover sets the tone with an attention-grabbing realistic, bright, large frog. He has movable eyes and a real 10-inch-long, pink, sticky tongue with a fly on the end. The tongue, made of a durable stretchy, tacky material, sticks to the book jacket, table, or wherever desired.

The large print settles into well-designed, full-page illustrations. Simple, effective words point out the *blue water* and *brown log*, while the *quiet green frog* suddenly *slurps* various insects with his long, sticky tongue. Action words encourage children to participate in the reading. In the surprise ending, a large fish eats the frog himself. Although slightly disconcerting, this is actually a valuable first lesson in the balance of predator and prey.

My first experience reading this book to a group of prekindergarteners was exciting for all of us, even those children with short attention spans. The children were smiling and laughing for hours afterwards, as they snapped up imaginary flies. I suspect this is one of the books that will remain etched in their memory as they get older.

This sturdy book is sure to be a mainstay for environmental programs of the future.

IWRC
YEAR-END REPORT
January through December 1999

Ordinary Income/Expense	
Income	
Conference	33,325.00
Contributions Income	2,926.00
Grants	9,000.00
Journal Ads	275.00
Literature	20,829.26
Membership Dues	65,951.00
Miscellaneous Income	160.84
Reimbursed Expenses	82.38
Skills Seminars	58,686.85
Total Income	**191,236.33**
Expense	
Accounting Fees	1,187.49
Bank Service Charges	3,469.10
Conference Costs	13,912.71
Dues and Subscriptions	40.00
Equipment Rental/Lease	4,920.69
Honorarium	90.00
Ind. Contractor Fees	16,075.00
Insurance	3,041.51
Office Expenses	5,432.90
Office Supplies	51.27
Payroll Taxes	13,899.11
Postage and Delivery	15,072.21
Printing and Reproduction	30,077.58
Professional Fees	1,750.00
Program Expense	9,189.38
Promotions	411.00
Publications	4,418.18
Rent & Utilities	9,872.00
Repairs	700.70
Service Contract	595.00
SS Program Expense	10,481.55
Tax Expense	1,771.56
Training Cost	290.00
Wages & Salaries	42,025.31
Total Expense	**188,774.25**
Net Ordinary Income	**2,462.08**
Other Income/Expense	
Other Income	
Interest Income	996.29
Other Income	1,283.11
Total Other Income	**2,279.40**
Other Expense	
Employee Investment Op.	1,464.27
Other Expenses	-1,485.40
Total Other Expense	**-21.13**
Net Other Income	**2,300.53**
Net Income	**4,762.61**

SUCCESS ON A VERY SMALL SCALE

The urge to release too quickly once an animal has been rehabilitated is always hard for me to suppress. After often spending many weeks on an animal, I have to remind myself that "where and when" becomes particularly important if it is a bird that migrates. So when, during the last week of September 1999, I received three tiny nestlings (close to fledging) from a nearby veterinary clinic, I worried about getting them released in time before winter. Their bellies had a yellow cast, and they were later identified as lesser goldfinches. One was a little puffy, hard to get to gape, and quite lethargic. They were so tiny and, my eyes not being what they once were, I bought a pair of magnifying glasses to see them better while feeding!

Unfortunately, a few days later the weakest succumbed, and several days after that a second one also died. Their deaths were puzzling, as both took food up until a few hours before they died, and both were of good weight. Their bodies were taken to an ornithologist at the University of New Mexico for the bird collection. The third, however, was very robust although it seemed to take forever before he was self-feeding. Seeds of all sorts were offered in the next couple of weeks, as well as grasses, wild sunflower seed tops, and various other vegetation gathered from my property in the mountains. Thistle seed seemed to be the favorite.

What luck to have quite a warm October here in central New Mexico. Just the same, deciding that the little bird would have to be over-wintered, I had prepared a large cage for him in a heated building. Lesser goldfinches who had been at my outdoor waterfall and pond had left some weeks earlier, and I was certain they had left the area for the winter. My decision to over-winter caused me increasing anxiety as I considered all the risks long-term captivity present to a wild bird. I don't know much about the migration habits of lesser goldfinches. Maybe some only go as far as southern New Mexico. Or maybe they are at many lower elevations year round. I just know they aren't at my place in the winter!

On one of the last days in October, I released a blue-winged teal near a small lake within a refuge near Edgewood, New Mexico. While talking with the director, I was thrilled to see a male lesser goldfinch alight on a bush not five feet in front of us. I had been so certain they had migrated already! A few days later I returned and released the goldfinch in the vicinity of two adult male lesser goldfinches. By this time he was fully self-feeding and wary of people.

This little bird had taken so much of my time as I fed him and taught him to self-feed—and worried about him. However, I had as much pleasure in his release as any other larger bird I have rehabilitated. He taught me the value of getting out into the wilderness and becoming aware of the surrounding bird life before making release decisions. In the future I will take the time to call local agencies and bird people to ask "when and where" that species may still be if it's close to migration time.

Catherine Foster
Rehabilitator, Wildlife Rescue of New Mexico and Talking Talons

Call for Papers
International Wildlife Rehabilitation Council
23rd Annual Conference

25–29 October 2000
Hotel Wyndham Montreal
Montreal, Quebec, Canada

"WILDLIFE WITHOUT BORDERS"

Each year at our annual conference, the International Wildlife Rehabilitation Council (IWRC) invites rehabilitators and other associated professionals to make presentations that will assist those working in the field of wildlife rehabilitation. We encourage papers that describe new and current methods, as well as innovative concepts and ideas. Your audience will range from volunteers to veterinarians. The focus of your presentation should be chosen based upon personal and field experience.

At this year's conference we will be focusing on the following areas:

- Migration
- Land Management/Corridors
- Urban Wildlife
- International Wildlife Treaties
- Illegal Pet Trade
- Wildlife Diseases/Zoonosis
- Translocation Issues
- Euthanasia Criteria
- Crossing Borders/Customs Agents
- Rescue vs. Kidnap
- Herpetology
- Public Education
- Zoos/Sanctuaries
- C.I.T.E.S.
- Animal Husbandry

If you feel that you can contribute to this important meeting, please submit your proposal on the application on the next page. All submissions are subject to review by the conference committee. Presenters will be notified in writing regarding the acceptability of their proposal(s).

CATEGORIES OF PRESENTATIONS

PAPER: A factual presentation of methods, report of research findings, and new or improved techniques.
Note: A manuscript *must* be submitted for publication in the conference proceedings.

WORKSHOP: A demonstration of technique or skills using a hands-on approach. The workshop should conclude with a finished product, a completed project, or a learned skill (e.g., "Turtle Shell Repair," "Cage Building," or "Publishing a Newsletter"). Attendance may be limited.

TUTORIAL SESSION: An opportunity to share successful skills, techniques, knowledge, or information, usually with a smaller group. A written outline or handout will be submitted for publication in the conference proceedings.

DISCUSSION GROUP: Discussion between peers on a specified topic or area of concern moderated by a group leader. A summary of the discussion and final conclusions will be submitted by the group leader for publication in the conference proceedings.

CASE STUDY: A short report (usually 10–15 minutes) on one case describing treatment and outcome. A written report will be submitted for publication in the conference proceedings.

POSTER SESSION: A presentation on paper for display during the conference. Specific time will be scheduled for presenters to be available at their poster for questions and comments from attendees.

DEADLINE: Proposals for the 2000 Conference should reach the IWRC office postmarked no later than 1 June 2000.