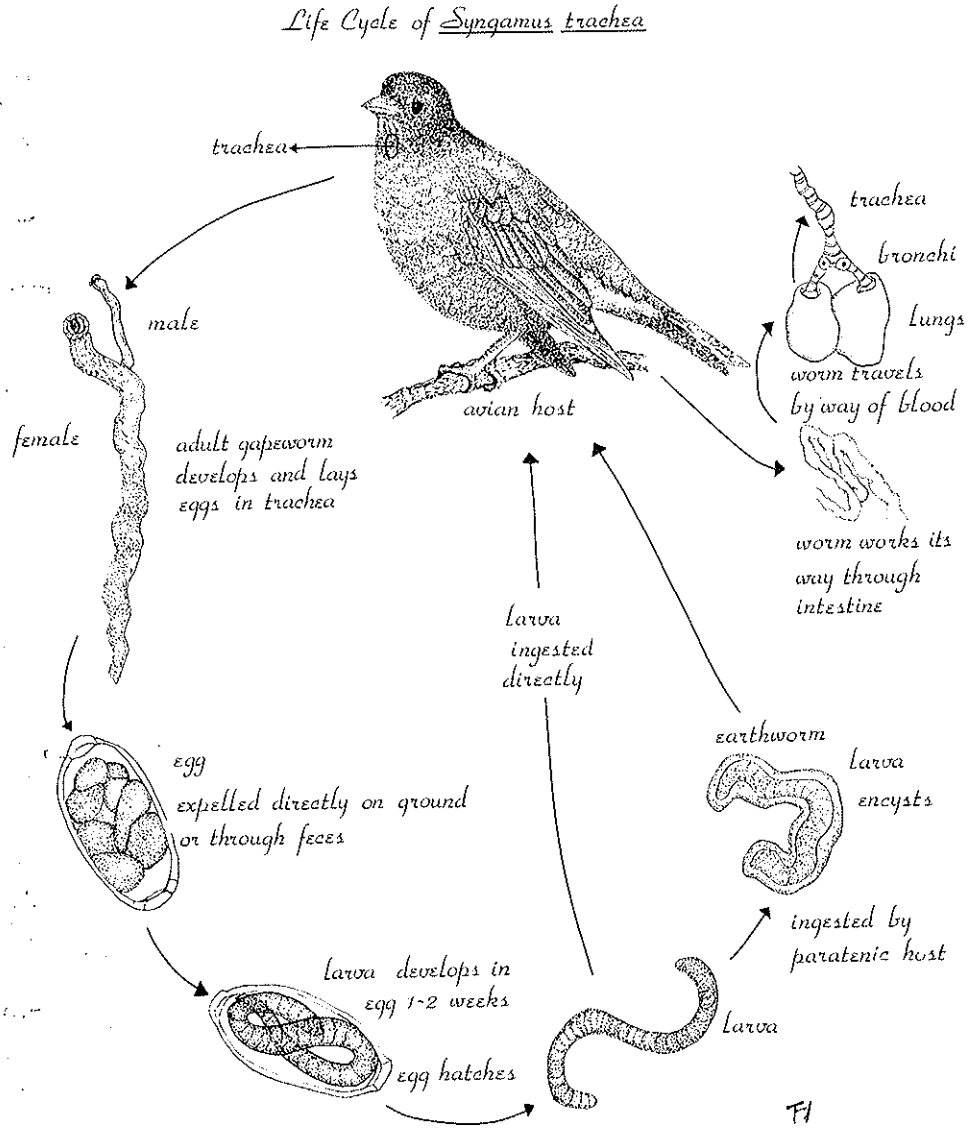
Spring 1988 Vol. 11, No. 1

WILDLIFE JOURNAL

Journal of the International Wildlife Rehabilitation Council



***Syngamus trachea* in Robins . . . Page 7**

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The following is a guideline for articles to be submitted to the Wildlife Journal. All manuscripts submitted to the Journal are subject to review and revision by our editors.

1. Submissions should be typed and double-spaced, with 1 inch margins.
2. Full length articles run between 1500 and 2500 words.
3. Fillers and short articles run between 500 and 1500 words.
4. The writer's name and address and organization should be typed on the first page, and name and page number should appear on all consecutive pages.
5. It is the writer's obligation to obtain permission or reprint rights if submitted copy has been published elsewhere.
6. Provide a stamped, self-addressed envelope if you wish manuscript and/or photos returned.
7. We accept black and white glossy photos only, preferably 8 × 10.
8. We pay $25 for any feature article accepted; $5 for any fillers and "Handy Hints" under 500 words.

Articles should be mailed to: Editor, IWRC Journal, 1171 Kellogg St, Suisun, CA 94585.

WILDLIFE JOURNAL

Spring 1988 Vol. 11, No. 1

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Cover drawing by Tina Hawkinson.

• The IWRC Journal is designed so you can use a 3-hole punch and store back issues in a binder for easy reference.

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Editorial . . .

Getting the Standards Program Underway

The joint Standards and Accreditation Program between NWRA (National Wildlife Rehabilitators Association) and IWRC has been in a process of evolution over the last four years. Recently, the proposed program was mailed to the NWRA membership for review and comment. It was well received and the comments made by rehabilitators will be forwarded to committee sub-chairs for review and integration. The program will be made available through both national organizations as a guide for rehabilitators.

There is as yet no funding mechanism to allow for this consulting program to be launched on a national basis and ideas for funding are being sought. We recently received word from the Resources Agency in Sacramento, California that they were willing to fund a pilot project in their state. This means that an organization or individual can apply to the program and have a team of rehabilitators to review their wildlife care program. Since the selection of the team is dependent upon the choice of the applicant, costs for the team (expenses, transportation, lodging) will vary. The bulk of associated program costs will be underwritten by this grant. We are very excited about this grant and have great hopes for this program.

Jay Holcomb, coordinator in California, (and IWRC President) will be putting together the application procedures and will be mailing them to rehabilitators during the fall. We hope to get the program underway by the end of the year. The pilot program will last 18 to 24 months. We hope to use the grant to establish a good track record and to attract other sources of funding. While rehabilitators have expressed a desire to have this program, most cannot afford to cover the costs of such consultation. What is needed is foundation support to help a new profession better its standards of care. We hope to assist in this process.

Special thanks need to be given to all of the members and invited reviewers who have helped create this program. Further, both the Board of Directors of IWRC and NWRA are to be commended for their courage and leadership because it was they who proposed such a program in the face of many nay-sayers who felt that the task was not accomplishable. This is a very difficult task, but we feel with tremendous effort and cooperation it can be done. The best thing is that the animals will benefit as well as the rehabilitators—and that is after all, why we entered this field.

—Jan White—

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For the New Rehabber:

Net-bottom Caging for Waterfowl

Jay Holcomb
International Bird Rescue and Research Center
Aquatic Park
Berkeley, California

Introduction

Housing waterfowl during rehabilitation has been a source of frustration and controversy for a long time. Water birds require perfectly waterproof feathers on release, therefore, their caging must enhance and preserve this vital condition. Caging with improper ventilitation, padding and drainage can result in feather damage, and keel or foot sores.

Birds such as loons and grebes are not capable of walking efficiently on land. They have long bodies and their legs are placed to the rear (caudal) and act like rudders. Their way of moving around is a series of jumps and slides. When they attempt this behavior in a cage, irreversible damage can occur.

Why Netting Pens Were Developed

The use of cotton fish netting as the bottom surface of a cage prevents most of these problems. Feces and spilled foods, especially fish, have chronically soiled feathers and prevented many a healthy bird from release. With the use of netting, these potential problems are virtually gone. Any liquid matter falls through the net keeping the bird on a dry surface. The softness of the net tautly stretched across the bottom provides both a supportive surface as well as a cushion for the birds as they move around, thus reducing pressure on their keels. The occurrence of foot sores and bumblefoot have rarely been seen on birds kept on netting. However, loons and grebes do attempt to swim on netting and can scrape their feet if kept on netting for longs periods of time. The delicate webbing on loons, ducks, and many other birds should be examined on a regular basis to see that they are not dried out or worn between the toes. An application of water soluble KY jelly ® helps keep feet soft and moist. Emaciated birds have the additional likelihood of developing keel lesions due to the lack of pectoral muscles across their keel. The netting may not prevent this problem if the birds stay caged for long periods of time (greater than two weeks) in a continuing state of emaciation. Birds should be placed on feeding protocols that will result in weight gain and should be allowed to swim regularly so they can build their pectoral muscles. While emaciated, they should be allowed access to water as much as possible. Close monitoring of these problem areas (feet and keel) are advised.

Keeping waterbirds clean (and therefore waterproofed) has been a challenging task with traditional caging. Three to four cage cleanings per day were required. With the netting, waste and spilled food fall away from the bird and through the netting thus eliminating the need for the multiple cage cleanings. Newspaper can be placed under the pens and changed once daily. The beauty of this system is that the bird does not have to be handled during the cage cleaning. The netting cage also provides air flow which is an improvement over traditional caging. Since aspergillosis and other respiratory diseases constitute a major threat to waterfowl, especially those housed on spongy wet bedding, the netting allows fordrier conditions and better ventilation helping to reduce these risks. Birds dry more quickly and do not get as cold when they are on dry bedding. Most dirty corners that can act as culture media for bacteria and *Aspergillis* are gone.

Three Netting Methods

1] **Net-bottom Rehabilitation Pens:** These pens were originally designed to house many birds at an oil spill and can be built to conform to your space limitations. They are basicially wooden boxes (sealed to repel water and to be cleanable) with tightly stretched netting on the bottoms. Legs with

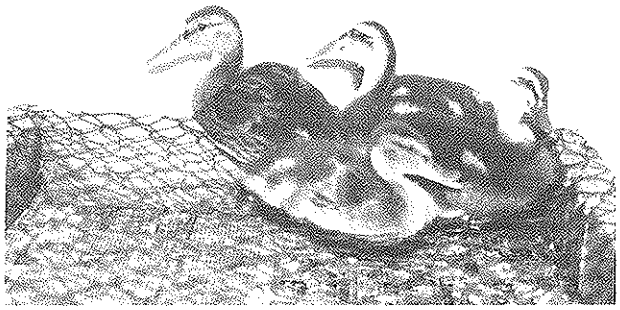
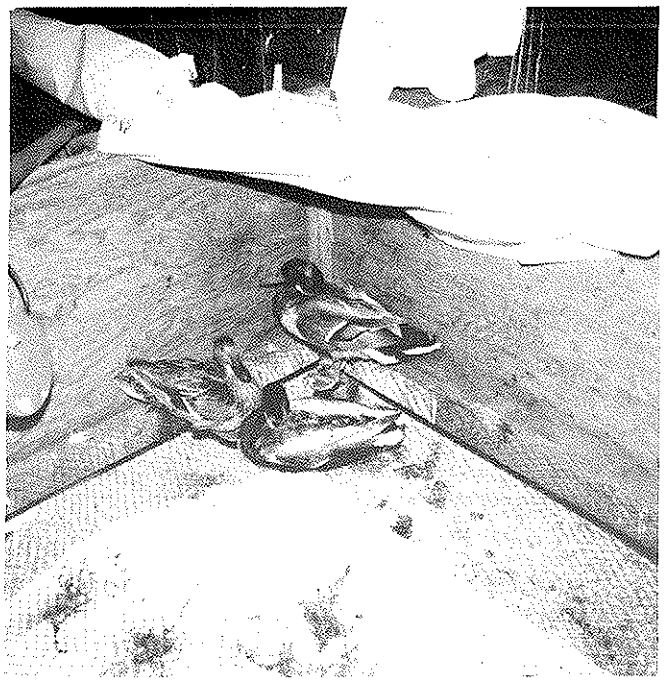


Photo depicts veterinary caging with net bottom frame.

wheels are mounted on all four corners to keep the cage movable and off the ground. Support beams under the netting can be made out of PVC pipe or 2" by 2" pieces of wood. Since the netting will bounce when birds are moving around the cage, a clip from a dog leash or key chain fastened to the side of the pen will hold the food pans in place.

2] Netting Frames for Standard Veterinary Cages: A frame made of 1" by 6" pine with tightly stretched netting works well in these cages. Newspaper placed beneath the frame is easily removed for cleaning. These same frames have also been used in pools as islands on which birdsmay to take refuge while waterproofing themselves. They allow a clean surface and plenty of drainage for drip drying birds. Netting may be removed from caging to be throughly disinfected, hosed, and sun dried. Netting can be easily replaced when necessary.

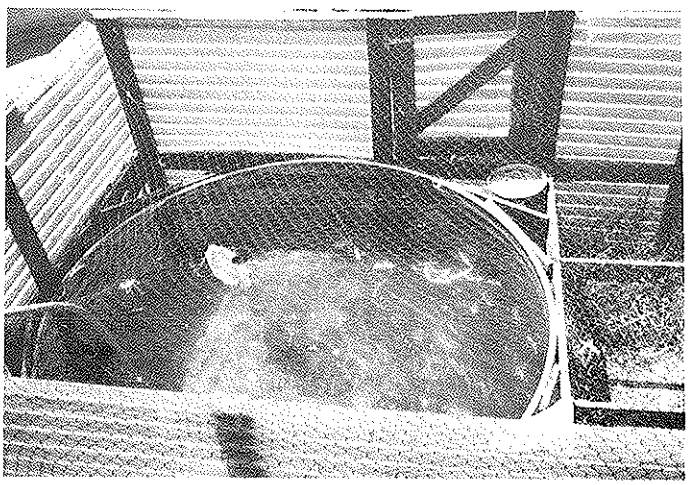
3] Waterproofing Pools: Any strong pool on a flat surface can be used. Galvanized round livestock troughs with bottom drains work very well. A square frame is built around the pool with a side two to four feet high. The flat corners of the frame where a platform would normally be (like the deck of a hot tub), should have stretched netting so that any bird getting out of the pool will always be on netting. Attaching netting to the sides of the pool takes a little imagination. A rope wrapped tightly around the pool can be used as siding around the exterior of the frame. One or two doors cut into the sides of the frame help for easy access. It is important that the water in these pools always be kept at the overflow point. A hose running at a trickle will maintain the fresh surface water that is a must for waterproofing birds.



The netting pen is used to house small groups of waterfowl.

Conclusion

The netting cage is a significant improvement in the methodology for rehabilitating waterfowl. It can be adapted in various ways and is an affordable, cleanable, flexible system. Best of all, it is the first system designed for extended waterfowl rehabilitation that actually allows the birds to maintain feather condition and given the opportunity, regain their waterproofing.



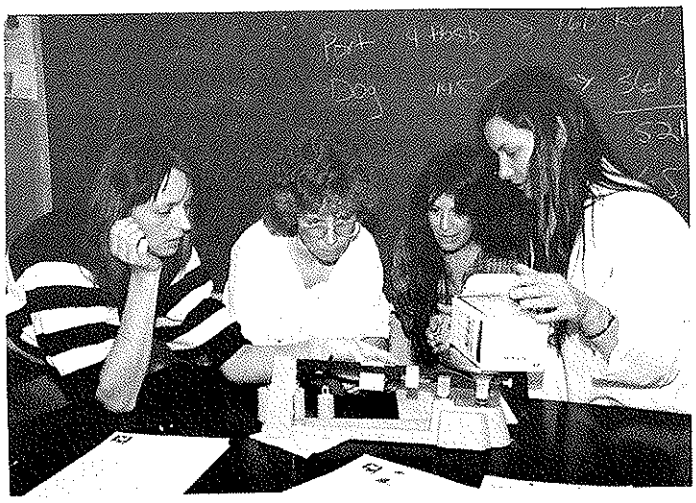
This photo illustrates an outdoor enclosure with netting frames around the pool to prevent feather-fecal contamination.

Skills Seminars:

A Program of Continuing Education for the Wildlife Rehabilitator

Jan White
IWRC Skills Seminar Coordinator
Suisun, California

The skills seminar program originated in 1982 to standardize training for the wildlife rehabilitator. While many organizations offer their staff and volunteers classes on basic care, there was nothing tailored to the needs of the wildlife rehabilitator once the basics were learned. IWRC Board members, Sandra Emanuelson D.V.M. and the author felt that more training was needed and would be welcomed by people in our field. Together, we developed curriculum for a set of advanced classes in California. Writing to some of the best people in our field, we asked for assistance in developing the first skills seminar series together. To our delight, the response from potential instructors was very positive. Our first instructors included Murray Fowler D.V.M. (Wild Animal Nutrition), and Terry Schultz (Raptor Rehabilitation) from the veterinary school in Davis, CA, Ms. Nancy Horowitz, R.P.T. a registered physical therapist who applies her trade to birds, Sandra Emanuelson, D.V.M. (Physical Examination and Initial Care) and her animal health technician, Tami Anderson, A.H.T. (Clinical Pathology), and Pat Gullett, D.V.M. (Gross Pathology and Chemical Restraint) and Gary Bogue (Restraint).



Instructor Dara Nance illustrates proper weighing techniques in a Basic Wildlife Rehabilitaion Seminar.

The program was so well received that restrictions had to be placed on the number of participants each organization could send. Using feedback from the participants and instructors, the series evolved over the years to include classes by Mike Murray, D.V.M. on Fluid Therapy and Wound Management, and by Jay Holcomb and Alice Berkner on the cleaning of oiled birds. Additionally, the California Department of Fish & Game (Dave Hunter, D.V.M. & Bill Clark) has offered annually a short course on Chemical Restraint and Immobilization. This year, we have again added a class on Volunteer Administration by Sally Lewis.

We discovered that while nearly all of the participants had some background classwork, the content of that classwork varied, which presented the advanced skills seminar instructors with some interesting challenges. It became apparant that a standardized basic training course was needed in wildlife rehabilitation. Sandra Emanuelson, Doris Van Winkle, and myself then developed a basic course that would bring everyone to the level where they could successfully complete the advanced series. We began with a 40 page manual and a two day class that included a hands-on skills laboratory period and homework. The class concluded with a one-hour examination. Participants who achieved a score of 70% or greater, completed both the homework and laboratory skills tasks are awarded an IWRC Basic Wildlife Rehabilitation 1AB certificate. The class was offered in northern California and was well received. Then, we began to receive requests from rehabilitators in other areas and decided to take the class on the road.

We first took the class up the pacific coast to Portland, Oregon, and Seattle, Washington. The response from the students was very revealing. We

had little idea that the class would be so welcomed by rehabilitators. Over the next few years, we traveled across the country from Long Island, NY to Bowie, MD to Orlando, FL to Wichita, KS, to Boulder, CO and even to Sitka and Anchorage, AK. In our travels we learned much about wildlife rehabilitation today.

The basic course contains much material and it is with great pride that participants earn their completion certificates. The 40 page manual is now some 120 pages long and increased with each revision. The homework and exams are revised annually. Our small team of instructors has grown to include some of the finest rehabilitator/teachers in our field. We have Curt Clumpner from Progressive Animal Welfare (PAWS) in Seattle, WA., Linda Wolf D.V.M. from the University of Minnesota Wildlife Center in St. Paul, Debbie Daniel, M.T. formerly of Tri-State Bird Rescue in Delaware, and from California: Margie Comstock, A.H.T., Kathy Hobson, Kris Thorne, and the author.

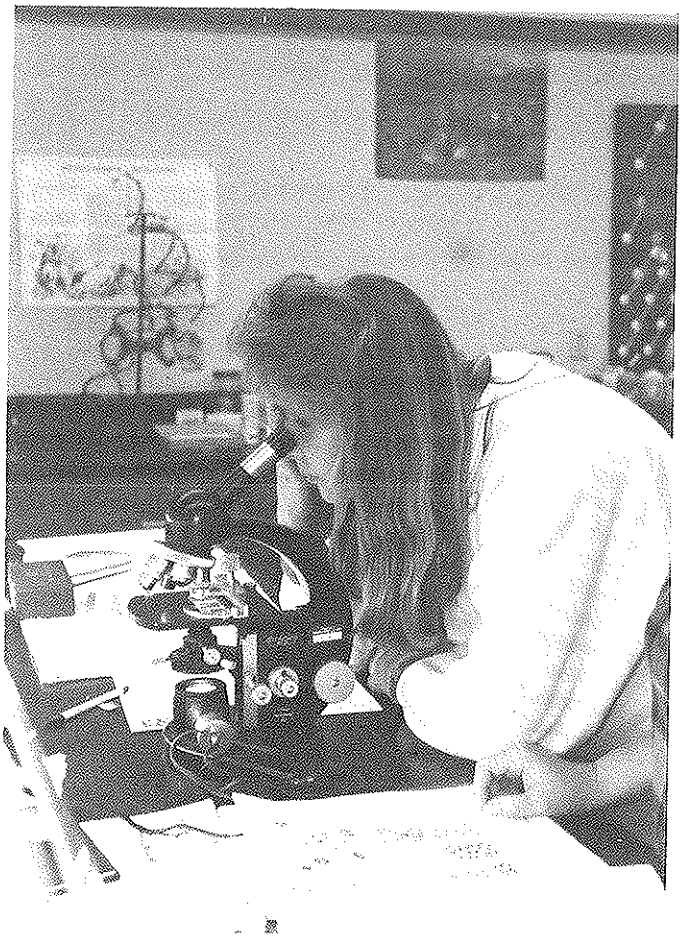
The advanced classes have been offered in sets of three: Clinical Pathology for the Wildlife Technician, Wound Management and Fluid Therapy. We have taken the class to areas where we have had two of basic classes (since participants must be basic graduates) and where the necessary equipment is available (we need oil immersion microscopes, centrifuges, etc.). These classes have been held at the IWRC conference, at a Solano Community College, Suisun, CA, in Portland, OR, and in Bowie, MD. This year we hope to offer these classes in the northeast, possibly New Jersey.

Besides the efforts of the Skills Seminar committee members who schedule and coordinate classes, travel arrangements, etc. (the author for national classes and Susan Heckly of the Alexander Lindsay Museum in Walnut Creek, CA for the California classes), the hosts of these classes put a great deal of energy into making them a success. They often coordinate registration for the participants as well as housing and lunch arrangements. Hosts assist us in notifying potential rehabilitators by gathering lists of permittees and sometimes make mailings to them. Additionally, hosts make site arrangments, gather the live animals needed for the class, recruit the assistance of a local veterinarian and a rehabilitator for the skills lab, make housing arrangments for the instructors and even meet the instructors at the airport! All and all, the success of these classes very much depends on the efforts of these coordinators. Their professional efforts have been vital to the program and we are very much indebted to them.

In 1987, we were able to teach nine basic wildlife care classes to 360 particpants from 17 states. Our advanced classes were attended by about 200 participants from five states. In 1988, the program is continuing to grow with seven basic classes and eight advanced classes being offered in the first six months alone.

We are very proud of the efforts that we have made to serve our members and have learned a lot in providing that service. We look forward to the development of more such programs as the field of wildlife rehabilitation evolves.

If you are interested in hosting a class and would like more information on our current scheduling efforts, contact our office at 1171 Kellogg St. in Suisun, California 94585. Calls can be made to (707) 428-IWRC.



IWRC Advanced Skills Seminar Participant is shown learning hematology cell identification skills.

The Treatment of the Gapeworm Parasite, *Syngamus trachea*, in American Robins, *Turdus migratorius*

Katherine Moran
849 E 13th Ave.
Vancouver, British Columbia
V5T2L4

Introduction

In June 1987, eighty-nine of the four hundred twenty seven cases admitted to the Wildlife Rescue Association of British Columbia were American robins, *Turdus migratorius* (Table 1). Many of the robins arrived at the facility with gapeworm *Syngamus trachea* and many of them developed evidence of adult gapeworm after arrival.

Gapeworm is transmissible from one bird to another (Fowler, 1986), therefore it was never established if the birds picked up the parasite inside our crowded facility, or whether they arrived with the parasite already ingested. Gapeworm can be fatal, especially in young robins because of the small diameter of their trachea. Since we were not aware of any effective treatment at that time, humane euthanasia was chosen. When over 20% (Table 1) of our caseload were robins and many of them had the parasite, euthanasia was a depressing choice. After reviewing the literature and consulting with our veterinarians we decided to try Ivermectin* in the treatment of gapeworm in robins. It is important to understand the robin, the drug and the lifecycle of the parasite to evaluate the effectiveness of Ivermectin.

Life Cycle of Syngamus Trachea

The adult gapeworm, *Syngamus trachea,* is blood red and visible in the glottis of an infected bird. The female worm can be up to 40 mm long and the male up to 6 mm (Petrak, 1982). The worms in the glottis and trachea may appear to have a y-shape because the tiny male is cemented at an angle in copulation with the larger female (Fowler, 1986). (See Figure 1.)

Adult female parasites deposit fertilized eggs in the trachea of the host bird. The eggs are coughed up by the host and expelled directly on the ground, or swallowed and later passed in the feces (Petrak, 1982). These eggs are easily recognizable under a microscope owing to their large size (80 to 100 µm by 40 to 60 µm) and their distinct shape. They are ellipsoidal and have a thick shell with plugs at each end. Inside a morula can be seen. These eggs are quite hardy and can withstand freezing (Fowler, 1986).

After an incubation period of one to two weeks under optimum conditions, third stage larva develop within the egg. The infective larva hatches and is eaten directly by the host, or it can enter the avian host through a paratenic host, such as an earthworm, snail or slug. The gapeworm larva is ingested by the paratenic host where it becomes encysted within the muscle. The larva is still infective to a robin after one year (Petrak, 1982).

Once ingested by the host bird, the larva penetrates the intestinal wall and travels by way of the blood to the lungs. This can happen in as little as six hours. A second molt occurs in the lungs and the young worm travels up the bronchi to the trachea.

Signs

One of the most obvious signs that robins have gapeworm is the presence of red worms in the glottis. They may also show signs of weakness and emaciation. In more severe cases, they may spend much of their time with their eyes closed and head drawn against their body (Hofstad, 1978). They may also suffer voice change or loss (Harrison, 1986). As more worms crowd the trachea, infected birds may gasp, cough and have dried bloody mucus at the corners of the mouth. Respiratory problems caused by tracheal blockage greatly debilitate the bird. Sudden death by suffocation may occur as a result of exertion (Harrison, 1986).

*IVOMEC,® Division of Merck Frosst Canada, Inc., Kirkland, Quebec, Canada.

Visual inspection is one method used to determine if robins have gapeworm. This is done by extending the necks, transilluminating the trachea from below with a small flashlight or penlight, and looking into the glottis where the adult worms are apparent. Another method of determining *Syngamus trachea* infection is by fecal flotation. Robins are very susceptible to numerous endoparasites, such as coccidia, *Capillaria* and hookworms in addition to gapeworm. Eggs of the gapeworm are easily discernable under low power on a microscope, because of their large size and unique shape. Due to limited time and facilities, the method we used for diagnosis was visual inspection of the glottis.

Post Mortem

When a bird dies from gapeworm infestation, sometimes the trachea is almost completely blocked by a mass of worms. There is excessive mucus in the trachea. Granulomas and perforations are left in the trachea where the female adults were attached. The lungs may be edematous (Petrak, 1982; Harrison, 1986).

Treatment

The treatment we used for gapeworm was Ivermectin. Ivermectin comes from the family of drugs known as Avermectins. Ivermectin was approved for used in horses in the United States in 1980 (Clubb, 1983). Dr. Clubb used the dosage recommended for small mammals to treat a variety of domestic birds. She found it to be useful in the treatment of several ectoparasites and endoparasites. Avermectins are unique because an extremely small quantity is needed for antiparasitic activity. Action of this drug against parasites relates to the inhibition of the worm's motility. Ivermectin causes an increase in the release of a neurotransmitter, GABA, which inhibits nerve transmission (Booth, 1982).

Treatment of gapeworm has been recommended when the bird is weak or thin. A healthy adult bird showing some gapeworm ova in feces is unlikely to develop a life threatening problem. Johnson (1986) suggested treating gapeworm with the equine form of Ivermectin (1.87%) diluted at a ratio of 0.25 ml in 10 ml of saline solution. The diluted medicine is then administered by intramuscular injection (IM) at a dosage of 0.25 ml/Kg (i.e., 0.11 mg/Kg).

Harrison recommended the use of Ivermectin in its injectable form (10 mg/ml) given IM or orally at a dose of 0.2 mg/Kg (200 µ/kg). When repeated in 10 to 14 days, this is an effective treatment of intestinal nematodes, coccidia and gapeworms in domestic birds.

Our treatment consisted of diluting 0.1 ml of bovine Ivermectin in 0.9 ml of sterile lactated ringers resulting in a strength of 1 mg/ml. Bovine Ivermectin diluted with propylene glycol, given orally can also be used. A standard dosage of 0.02ml (of the diluted solution) was given IM. The standard dosage was based on the average weight range of robins, 63 to 103 g (Johnson, 1986) and a dosage of 0.2 mg/kg., as follows:

$$63\text{g to } 103\text{g} \times \frac{1\text{Kg.}}{1000\text{g}} \times \frac{0.2\text{mg}}{1\text{ Kg}} \times \frac{1\text{ ml}}{1\text{mg}} = .013 \text{ to } .021\text{ml}$$

Since our rehabilitation centre was very busy, a standard dosage seemed the most efficient way of treating the parasite. Ivermectin has a wide margin of safety; toxicity and death resulted in cattle only when the dosage was 40X recommended level. Thirty times the normal level did not produce signs of toxicity (Alexander, 1985). Additionally, for reasons of the birds' safety, no young robins were injected with Ivermectin until they were fully feathered, approximately 2 weeks of age.

Results

The following information is not intended to be conclusive evidence that Ivermectin will rid robins of *Syngamus trachea,* but rather the results of a practical application of a drug previously used in domestic birds. This information may be used to initiate further study. The injection of Ivermectin had no apparent ill effects on the health of the robins. Eight robins were released 12 days after a single injection of Ivermectin, 3 after 24 days, 1 after 28, 1 after 32, and 2 after 38 days. The three birds released after 32 days were in captivity long enough for reinfection and for the larva to mature to the adult stage. All birds were inspected visually for gapeworm periodically and again on the day of release. No birds had evidence of adult parasites and none of the birds died after being injected. Upon visual inspection, the glottis of robins treated with Ivermectin were free of gapeworm within 24 hours.

Due to a shortage of aviary space, robins treated for gapeworm were caged uninfected robins. Yet, all birds were presumably free from the parasite. This suggests that the previously infected birds did not pass the worms to the other birds.

Most of the robins which were admitted to the facility were injured by cats. Our standard protocol for this type of case is to put the bird on a regimen of oral antibiotics (amoxicillin) for five to six days to prevent possible septicemia. In two cases robins

were diagnosed as having gapeworm as well as cat injuries. When Ivermectin was given in conjunction with amoxicillin, both birds were dead by the next morning. This may have been a drug interaction, added stress to the bird, or that the robins were going to die anyway. We found a more suitable treatment was to run the course of the antibiotic, wait one day and then give the Ivermectin injection. Three robins were released after this method of treatment.

Discussion

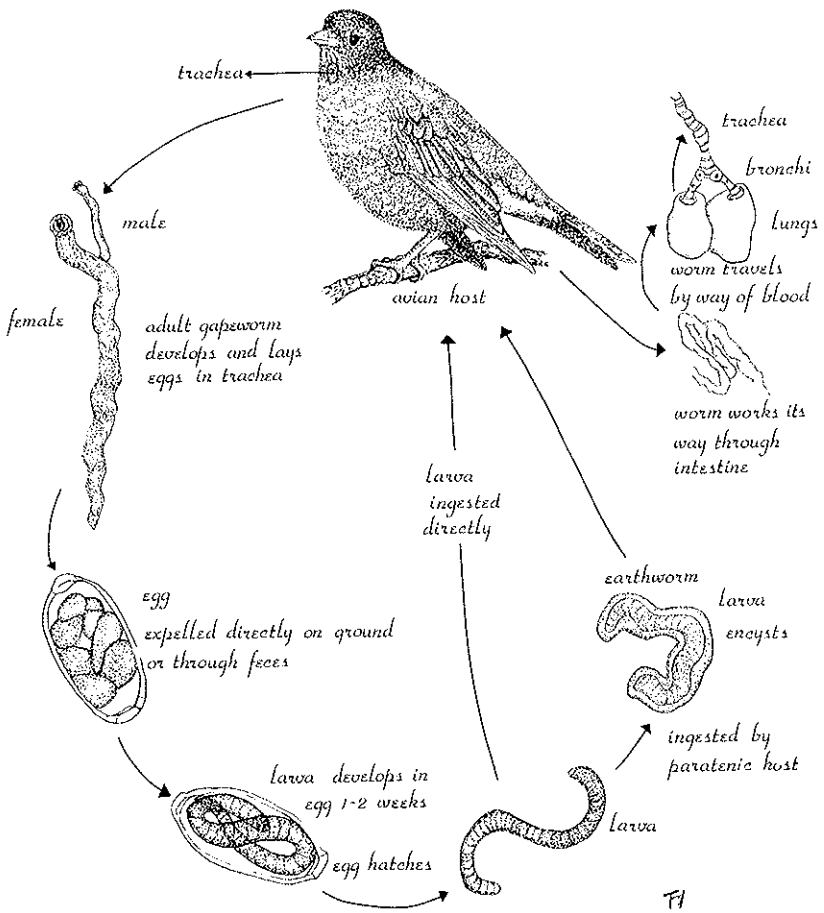
Syngamus trachea is not immediately fatal to robins. Healthy adult birds can survive a minimal infection. As the number of worms increase, the more difficult it is for the bird to breathe. This means flying and feeding become more difficult and the bird will become weaker and more susceptible to predation. In a bird with a small trachea such as a young robin, a mass of worms could completely obstruct the passage of air resulting in death by suffocation.

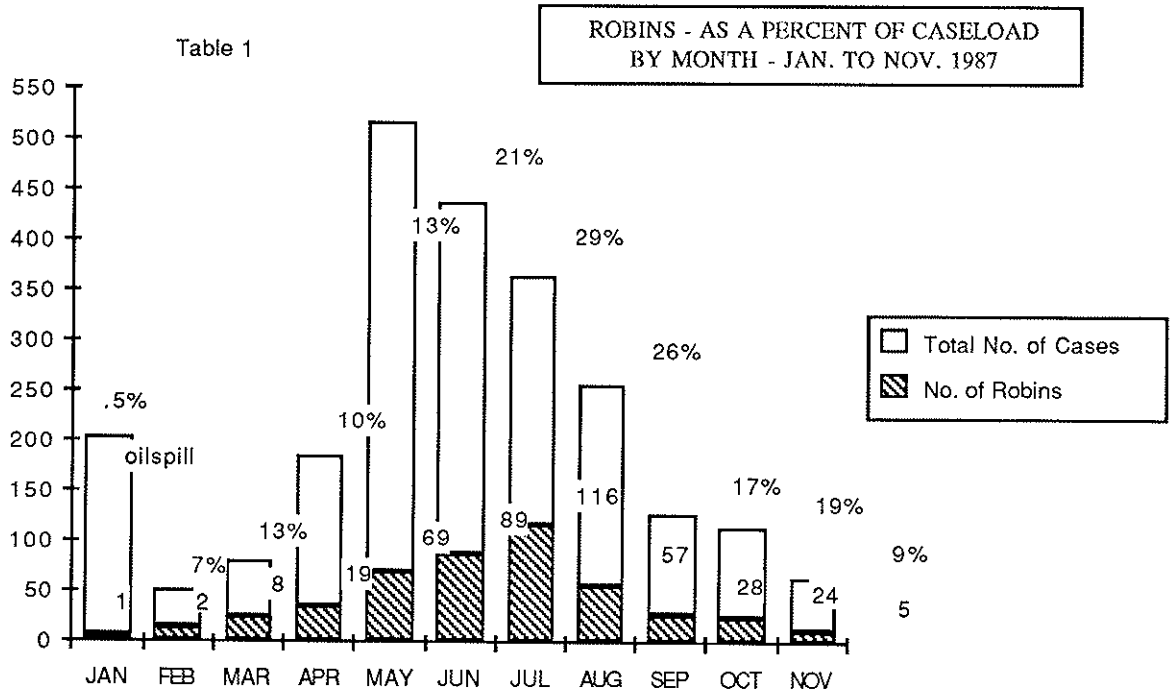
The gapeworm is very hardy, its larvae exist in paratenic hosts for over a year and its eggs are able to resist freezing. It is also transmissible from parents to young, and to different avian species.

These facts have several implications. First, immediate treatment of gapeworm is not necessary because birds can survive with the parasite in the glottis. Therefore, more serious disabilities, such as wounds from a cat can be treated first. Secondly,orphans arriving at a facility can remain untreated until mature enough to handle better the dose of the drug. (We waited until the bird was fully feathered, approximately 2 weeks of age.) Thirdly, gapeworm should be treated in all birds to prevent spread in the facility to non-infected birds. Also, effective treatment of *Syngamus trachea* may prevent further spread in the wild.

The best treatment is prevention. Several things can be done within a facility to slow or stop the spread of the parasite. Each bird should be checked for gapeworm upon arrival at the facility. Birds

Figure 1
Life Cycle of Syngamus trachea





with the parasite should be isolated from non-infected birds. Each bird should be housed individually in a disposable cage, such as a cardboard box to prevent accidental contamination.

It is not always immediately obvious that a bird has gapeworm when it arrives at the facility. The time it takes for the larvae to develop into adults which are visible in the glottis of the host is less than two weeks. During this time the birds should be housed separately. After this period if the birds show no sign of the parasite they can be housed together.Handlers should be aware that they can pass the eggs of the parasite from cage to cage with their hands during cleaning and feeding.

If a robin has gapeworm, a standard dose of 0.02ml of 0.1ml bovine Ivermectin (IVOMEC) diluted in 0.09ml sterile lactated Ringers, given IM is a good treatment. Since bovine Ivermectin is in a propylene glycol base and will not have uniform distribution in lactated Ringers, it is recommended that the Ivermectin solution be shaken just prior to use. There is also a new oral equine solution which is diluted with water. The results of the this paper are by no means conclusive, further study is required. This paper is a summary of data gathered in a practical application of the drug along with relevant information found in the literature.

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The Rehabilitation of Owls on an Outshelter Basis

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Introduction

For the beginning wildlife rehabilitator, the first encounter with a juvenile or injured adult owl can be a traumatic one. The following is offered as a guideline to those novice rehabilitation groups that do not have a center. It is not intended to replace professional care.

Identification

It is important to know which species of owl you have in order to design an appropriate care program. Juvenile owls, like all baby birds, should be alert, fully fleshed, and appear "fluffy". It is vital to be able to differentiate the normal bird from the abnormal. When in doubt about the condition of the bird, consult your veterinarian, or if he/she is unavailable have an experienced rehabilitator look at the bird to determine if veterinary care is necessary, or if the condition warrants special care.

Handler Safety

Owl talons are DANGEROUS. Leather welding gloves should be worn when examining all but the very young and featherless juveniles. Even a young screech owl can puncture the skin of the handler. Any puncture wounds received to the handler by the owl should be treated immediately with a Betadine solution and covered with a bandaid. Proper eye protection such as glasses, goggles, or a full clear shield face protector should be worn when handling owls. When frightened they usually throw themselves on their back and try to protect themselves with their beaks and talons. They are extremely fast, so it is best to be fully prepared and protected. Keep human tetanus shots current.

Intake Procedures

These are the standard IWRC recommended intake procedures as outlined in the class manual *Wildlife Rehabilitation 1AB.*

Information to be gathered on the phone:

1) Where did the bird come from?
2) How was it captured?
3) Get the name, address, and phone number of the finder.

Information to be gathered upon arrival:

Quickly evaluate the bird in the container:
1) Is the bird placing its weight evenly on both legs?
2) Is the bird holding its wings normally (i.e., is one drooping)? Can the bird fully extend its wings? Can it fly?
3) Is there a continuing blood loss? Is there blood in the container?
4) Is the bird alert and reactive to environmental stimuli? Does the bird follow your movement with its eyes?
5) Are the mandibles aligned?
6) Are the droppings of normal quantity, appearance, and texture? Is there any fresh blood (red) in the droppings?
7) Touch the keel bone. Is it sharp or well- fleshed.

Assessment of stability is of the highest priority. If the bird is weak or in shock, put a heating pad under half of the container, and place the container in a dark and quiet place. Fluids and steroids can be life-saving. If medical treatment is not available on an outshelter basis, unstable birds should be presented for further veterinary assessment and care.

Ask the finder more questions at this time (if possible)

1) How long has the bird been in captivity?
2) Has any food been offered? If so, what types? Did it eat? When?
3) Has a veterinarian seen the bird? Who? When? Medications?
4) Further questions concerning the injuries should be asked to try to determine the etiology of the injury (i.e., hit by car, predator trauma, electrocution, trap injuries etc.) These things give useful information in the medical management of the cases.

If the bird's condition is not life-threatening, and if the bird is not in stress, start the physical examination. Monitor throughout the exam for stress. If stress is evident, stop the procedure and replace the bird back into a warmed container in a dark, quiet place. The examination should be done in the same order each time so that no step of the physical examination is neglected. The author once completely overlooked a dislocated leg on a bird because of concern about a ruptured air sac. Blood flow, stress, and airway obstruction take precedence during the examination. USE YOUR COMMON SENSE.

Questioning The Finder

Be sure to ask every question on intake procedures. This is extremely important. Also, be sure not to let obvious physical problems distract you from doing a complete examination.

Equally important is to ascertain if anything has been fed to the owl. If this questioning is done in a friendly, non-accusing manner, the finder will be more willing to reveal just what they have done. Sometimes the diet given has been incorrect. If the owl does not look alert, full-fleshed, and healthy, with bright eyes and functioning limbs, present it to your veterinarian for further assessment and care.

The Examination

As the examination proceeds, write up the findings as each area is covered.

1) Temperature (shivering, fluffed, or panting) (Fowler, 1986)

SPECIES	Normal Temperatures C°	F°
Elf Owl	37.2	99.0
Pygmy Owl	39.1	102.4
Saw Whet Owl	39.0	102.4
Whiskered Screech Owl	37.9	100.2
Burrowing Owl	38.4	101.2
Eastern Screech Owl	37.5	99.5
Great Horned Owl	39.8	103.6
Snowy Owl	40.9	105.6

2) Check feathers for condition, breaks, or parasites.
3) Check eyes (direct and consensual response, equal dilation, foreign bodies, discharge, trauma, swelling). Remember that owls have voluntary control over the size of their pupils and can constrict and dilate at will. They can, when excited, give the appearance of constricting one while dilating the other. We are looking for the abnormal which would be when one pupil stays constricted in the face of low light over a period of time or stays dilated in the face of bright light over time.
4) Check the ears for parasites, discharge, or foreign bodies.
5) Check the mouth for oral lesions, evidence of trauma, discharge, or foreign bodies.
6) Check cere/nares for blockages, trauma, bruises, foreign bodies, or discharge.
7) Examine the crop. Is it full, empty, or partially filled? Is it intact?
8) Check skin turgor for dehydration. Separate the feathers and pinch the skin on the back, breast or leg. If the skin returns to normal in less than one second, it can be assumed that a state of dehydration does not exist. If the skin takes one to two seconds to return to normal, a state of 5 to 7% dehydration exists. Two to five seconds to return to normal indicates a 7-10% dehydration. If the skin does not return to normal it indicates a 10-12% dehydration. Dehydration requires veterinary attention.
9) Listen to heart and lungs. Can rales or gurgling be heard?
10) Look at musculature and bones. Is the bird emaciated, thin, or well fleshed? Palpate each joint and long bone for fractures. A drooping wing or lameness may indicate a fracture or bruising.
11) Gastrointestinal disorders can be difficult to detect. It is important to observe droppings as it gives an indication of food intake as well as whether food is being digested. Vomiting is an indicator of malfunction. Birds that are consistantly vomiting should not be fed orally. They are generally kept hydrated with fluids containing small amounts of dextrose (i.e., 2 1/2 %) given subcutaneously or intravenously. Do not confuse vomiting with the normal casting or pellet regurgitated by an owl several hours after a meal of its normal diet of rodents. The casting consists of undigestible fur, bones, etc. Normal owl droppings are dark brownish black (feces), with some white urates. Droppings should not be dry, which would indicate dehydration, nor green, which could indicate that the owl had not eaten for more than a day. Fresh blood in the droppings can indicate internal injuries. Any droppings that deviate from the normal should be noted and called to the attention of the veterinarian.
12) Check uropygial gland (oil gland) for any swelling, inflammation, or abnormal discharge.

Juvenile Diet

Determine if the owl is nocturnal or diurnal and plan to feed the owl according to its normal feeding schedule. This does not mean all night feeding is in order, however, it is appropriate to feed after dark with nocturnal species. An excellent captive diet for owls is adult mice, or for the smallest species of owls (i.e., Elf, Pygmy, or Screech Owls) large insects such as crickets can be offered. For young owls the mice should be skinned and the intestines, feet and head removed. The remaining mouse parts should be cut into 1/4"-1/2" size pieces and fed to the owl with a toothpick. If mice as a food source are not available chicks or beef heart cut into small pieces can be substituted. Calcium must be added at a ratio of 100mg per 28.4 g. (1oz.) of beef heart. Oyster shell calcium tablets (500 mg.) can be obtained at the drug store. Adding the calcium supplement to the beef heart diet equals the correct calcium/phosphorus ratio of 2:1. Supplementing is essential to the proper formation of strong bones in the owl. A sprinkle of Superpreen adds the necessary vitamin D3 as well as other vitamins. The beef heart/calcium/Superpreen diet is not ideal but it is adequate when properly balanced and used for a rehabilitation diet (not for permanent long term care). From a realistic standpoint, however, when large quantities of chicks, rats, or mice are not available, this diet is economical, readily available, and easily stored.

For juvenile owls presented having been fed improper diets (hamburger, liver, steak, etc.), calcium is added at a rate of 200 mg. to 300 mg. per 1 oz. of beef heart for the same number of days as the owl was fed the improper diet. I have raised and released healthy owls that were fed the incorrect diet up to four days, with no deleterious effects.

At first, juveniles must be hand fed. Toothpicks or wooden shish kebob skewers work well, depending on the size of the owl. Tap the side of the beak with the meat on the end of the toothpick to encourage the owl to open its beak. Gently place the meat toward the back of the mouth of the owl, taking care to avoid the toothpick or skewer coming in contact with delicate oral tissues. Have the food close to the owl so it can visualize the food. When the owl is in the "brancher" stage of development, a shallow dish of water may be added to its cage. Change the water as needed. Encourage the bird to self feed by delaying hand-feeding (when the bird is about 2-3 weeks or age of slightly older) while making food available to encourage self-feeding. Once the bird is self-feeding, hand feeding should be discontinued. This of course may not hold true if the bird has health problems or otherwise does not eat adequately.

Young owls are very appealing, and are easily tamed, habituated, or imprinted (depending on the age of acqusition) to humans. Do not speak to them while hand feeding. The author plays the *National Geographic* owl call record as I hand feed a young bird. This is an attempt to associate food with owl sounds, rather than human voices. Once the bird is self -feeding, place it with other birds of its own species. This action induces communication, learning and socializing with its own kind. Never remove the owl from the cage to amuse yourself. It will not enhance the survivability of the owl.

When the owls are self-feeding and are fully feathered, they can be introduced to whole, killed mice. (The mice, obtainable at pet shops as "feeder mice", can be humanely euthanized with carbon dioxide. Infant mice ("pinkie mice") are not recommended because they do not have calcified bones, and so are lacking in that essential mineral, calcium. If pinkies are the only mice available, insert 10 mg. calcium tablet in every third pinkie fed. When the owl is flying and eating whole mice or rats for at least two weeks, live food training should begin.

The owl, at this point must be transferred to a large aviary. If more than one owl is in the aviary being trained to catch live food, be sure to weigh each owl every day, as one owl may become the "Alpha" bird, and dominate the food.

The aviary should have solid walls at least four feet high so live food will not escape. Galvanized fine mesh wire should be attached on the bottom four inches of the interior baseline of the aviary, and extend to a depth of four inches into the soil so the mice cannot escape. Exposed wire on the aviary interior should then be covered with wood. Do not feed the owl the night before beginning live food training. The bird must be hungry to have the desire to pursue moving food. Some rehabilitators stun a few mice to get the owl started. When hunting and self-feeding for two weeks, it has completed this part of its rehabilitation. Release is discussed further in a later section.

Juvenile Housing

For the very young, featherless owl, I use a plastic notion basket obtained from any large market or drug store. The same basic baby bird suggestions apply for warmth and cleanliness.

The container should be lined with paper towels, which are changed each time they are soiled. An aquarium thermometer should be used, and the ambient air temperature should not fall below 85° F; this varies by age and size of owl. A heating pad should be placed under half the basket and closely monitored. In warm weather monitor the bird closely for signs of overheating (panting, beak open, prostrate). Once the bird grows down feathers, remove the heating pad. The bird should be cloistered from human or other species companionship so it will not imprint. Try to simulate the natural environment of the bird. For example,

cavity nesters such as Saw-whet owls prefer an inverted Kleenex box with an opening cut into the side of it, and until they are "branchers" will spend much time in the box. (See photograph below.)

When fully feathered, the bird should be moved to larger quarters. It can be placed with other owls of its own species and general age group, or in close proximity to adult owls of its own species. A large cage, big enough for the owl to fully extend its wings and turn about, should be used. If weather permits, this container can be placed under a covered patio, and the entire set-up may be placed in the morning or evening sun for an hour or so every day so the bird can manufacture vitamin D3. Keep any children or pets (especially dogs) away from the cage. When the owl's mature plumage begins to appear and it is self-feeding, transfer it to an aviary where it can begin flying. The owl should be weighed every second or third day, and the weight recorded on a chart, with the date. Before release the owls should weigh at least what is listed for that species in *Body Weights of 686 Species of North American Birds* by J.B. Dunning. Northern owls tend to be heavier than southern owls of the same species. Owls that were acquired in an emaciated state should be weighed on a daily basis. This provides valuable feedback to the rehabilitator on the progress of the bird, and should be provided to the veterinarian treating the case.

Adults

The owl acquired as an adult can be a handful. The first cardinal rule is your safety. ALWAYS wear arc welding gloves and eye protection. This is a full grown owl capable of causing great damage to human skin, muscle and tendons. The bird is in shock and has no idea that you are trying to help. Owls are strong. You have to be stronger, quicker and anticipate what will happen next. Always have a helper (not the finder) properly restrain the owl while performing the examination.

Prior to the examination, get a history from the finder. Obtain the exact directions to the place where the owl was found, because you will want to release the owl in its own territory whenever possible. Owls are territorial and if not out of circulation too long, will still have retained their territorial rights.

Any life-threatening conditions should be alleviated immediately. Above all, "do no harm". First aid measures that may be necessary include simple wing wraps, staunching hemorrhage with direct pressure, administering topical medications, and opening the airway. Generally, the owl can be kept in a clean box with air holes, and papers on the bottom. Place this container in a darkened, quiet, warm room. Transport to the veterinarian as soon as possible when veterinary care is required.

IMPORTANT: Always scrub your hands thoroughly with a 50% Betadine scrub both before and after handling wildlife to reduce possible zoonotic diseases, or transmission between animals.

First Aid: Check the mouth of the bird for obstructions and if any are present remove them. Clean open wounds with a diluted betadine solution (25% Betadine and 75% water). Serious wounds and compound fractures (bones exposed) should be covered with Nitrofurazone, Nolvasan or Betadine ointment to prevent the bone from drying. Cover the wound with sterile gauze and tape in place with masking tape. (It doesn't damage feathers as badly as adhesive tape.) Reapply ointment every 8-16 hours.

If the wing has a simple fracture of the humerus, radius or ulna, the tips of the flight feathers can be taped together behind the bird in the natural position to prevent further injury. This is not a substitution for the basic wing wrap, but useful only if the bird can be immediately transported to the veterinarian. Do not use this type of taping for any fracture below the wrist joint.

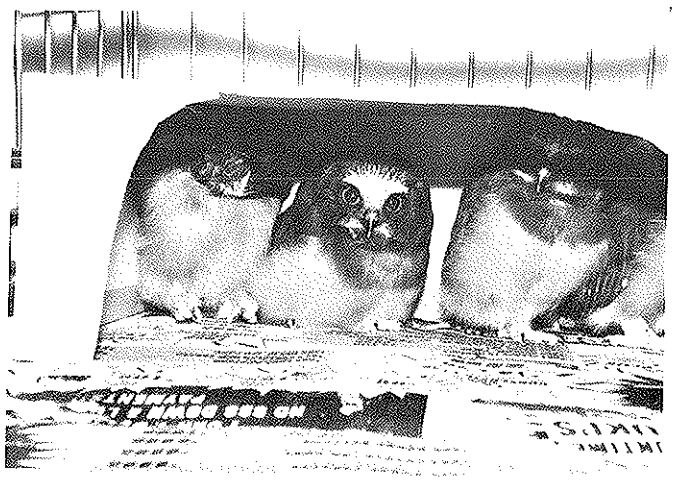


Photo illustrates box housing used with young raptors.

Housing:

A seriously injured shocky owl requires a warm, quiet, dark place in which to recuperate. A box lined with newpapers is acceptable and provided with adequate ventilitation. The box should be replaced with a new, clean box every second or third day for proper cleanliness. If the owl has had surgery or open wounds it must be confined indoors in a clean environment to prevent bacterial contamination, and to keep insects out of the wound (maggots). If the enclosure is kept properly clean, odor will not be a problem.

Keep track of the bird's progress with a chart.

making notes on anything to discuss with the veterinarian. Be sure to take your record when visiting the veterinarian. When the owl is ready to go to an outdoor cage or aviary, put it in the recommended enclosure. The adult owl should not be exposed to chicken wire or other types shown to cause feather damage. Feather damage may result in having to hold a bird until it moults. Large cages can be covered with soft shade cloth on the interior.

Food:

The adult injured owl can be offered killed mice or rats as a food source. If the bird is not eating on its own, the diet can be force fed. An owl that refuses to eat and is below normal weight should be force fed. Owls with fractures may be given 300-400 mg. additional calcium per day to promote callous formation if the veterinarian so desires.

An adult owl that has been immobilized for a few days should be given ample time to build up flight endurance. Prior to release a full grown owl requires live food for three to five days to ascertain its skills in obtaining food. This is especially true when limbs have been injured. Observe the owl when hunting.

Aviaries

We suggest that rehabilitators who are interested in working with owls and who do not have large exercise aviaries for the completion of the rehabilitation process confine themselves to either the initial care, or initial care and recovery period of owl rehabilitation. Then the rehabilitator can give the owl to someone who has an exercise facility, and who is willing to feed live food prior to release. The following aviary size and building material guidelines are from the IWRC/NWRA National Standards and Accreditation Program.

Building Materials

For mews/indoor facilities, solid walls may be constructed of wood, fiberglass, or the equivalent. Vertical wood lathes are an acceptable wall covering. If wire is used, it is to be covered with fiberglass screen netting, shade cloth, vertical wood lathes, or bamboo shades that are attached vertically. Another acceptable material is a plasticized fencing material manufactured by ADPI Enterprises, Inc. 3621 B. Street, Philadelphia, PA. If the aviary is an outdoor facility, three sides should be enclosed with wood or fiberglass as a protection from wind and rain. A double door is highly recommended to prevent accidental escapes.

Perches: In order to prevent "bumblefoot", wood doweling covered with matting hemp rope or indoor/outdoor carpeting should be used. This can be hot-glued into place, or nailed with roofing nails. Do not attach carpeting with a staple gun, as the staples work loose and can cause injury to the owl(s). Carpeting attached with a hot-glue method can be removed with a putty knife for replacement later. Platforms must be covered in the same way. Inspect and replace as necessary, as owls are known to chew on their perches. Natural branches are acceptable, but should be cleaned between uses by different birds.

Flooring: Newspapers or wood shavings are acceptable materials for covering the bottoms of boxes and cages. Change once or twice daily. For aviaries, pea gravel over sand drains well and should be changed biannually or as necessary. Natural flooring (dirt) is acceptable when turned over and disinfected periodically. A non-toxic enzyme culture product such as Stable-Rite, available from Omaha Vaccines, P.O.Box 7228, Omaha, NE 68107-0228, accelerates the decomposition of the droppings. Woodchips are inexpensive, but must be replaced when soiled. Concrete with good drainage is easy to keep clean, but is hard on birds' feet and stays damp when hosed; it is best used as a base covered with several inches of gravel or wood chips.

Minimum Caging Sizes

Species	Initial Care	Recovering	Primary Exercise Aviary
Common Barn Owl	18"x14"x18"	24"x18"x24"	15'x8'x8'
Great Horned Owl	22"x16"x21"	24"x24"x24"	20'x10'x15'
Long-Eared Owl	18"x14"x18"	24"x18"x24"	15'x8'x8'
Short-Eared Owl	18"x14"x18"	24"x18"x24"	15'x8'x8'
Spotted Owl	22"x16"x21"	24"x24"x24"	15'x8'x8'
Great Gray Owl	36"x36"x36"	72"x72"x48"	20'x10'x15'
Burrowing Owl	17"x17"x10"	17"x17"x17"	8'x8'x8'
N. Saw-whet Owl	17"x17"x10"	17"x17"x17"	8'x8'x8'
Flammulated Owl	17"x17"x10"	17"x17"x17"	8'x8'x8'
Pygmy Owl	17"x17"x10"	17"x17"x17"	8'x8'x8'
Elf Owl	17"x17"x10"	17"x17"x17"	8'x8'x8'
Western Screech Owl	17"x17"x10"	17"x17"x17"	8'x8'x8'

Cleanliness: Along with the standard cleanliness procedures of hosing off platforms on a daily basis, the methods and materials for disinfection of aviaries and cages are set forth in the Standards & Accreditation packet.

Hacking Back and Release Sites

"Hacking back" or "hacking" is the term used to provide a released raptor with food daily and in the same location until the bird becomes proficient at catching prey. This is done on a platform or roof with a light-colored dish. Killed mice or rats are provided, a few fewer every day for approximately ten days. Try to release the bird when there is a reasonable assurance of three days of clear weather. Plan to offer food for a longer period of time at the hack site if it rains.

Release sites should be carefully chosen. It is absolutely necessary to obtain the permission of the property owner if you plan to release the owl on private property. It is an excellent idea to have a release form signed by the property owner absolving you and your group of any responsibility, should anything occur. If the rehabilitator has done a good job, the owl will probably not be looking for any human companionship. The author always makes sure the property owner has an appreciation for owls and for their inherent abilities as rodent exterminators. Do not overload a given territory. This will vary by species, (e.g., a great horned owl has a territorial range of about five miles), depending on availabilty of food.

Monitor the project nightly. Some owls, when released, fly off without a backward glance and never return for food. Placing owls at a young age into the hacking station will increase the likelihood that they remain the area and feed post-release. Those that stay may feed for ten or more days.

Conclusion

Owls can be successfully rehabilitated on an outshelter basis if the above guidelines are followed, and if the appropriate sizes of aviaries are available for the final stages of rehabilitation. Our organization rehabilitates as a team effort, with some members handling injured adults and juveniles up to the point of live food training and transferring the owl to another member who cares for the owl through live food training and hacking back.

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