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Special Topics Issue: Raptor Rehabilitation

Guidelines for Authors

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 purpose of returning them to the wild. All feature articles are subject to a review process, and our staff and reviewers work with authors to produce the most accurate information possible in our ever-changing field.

The *Journal of Wildlife Rehabilitation* has various feature categories. We solicit submissions in the form of feature articles, rehabilitation notes, book reviews, news articles related to major events in the field, and selected case studies. Articles on veterinary medicine as it relates to wildlife rehabilitation are also welcome. We would like to expand the *Journal of Wildlife Rehabilitation* to include wildlife rehabilitation articles from countries outside the United States and welcome inquiries regarding such articles. We also have two short features we use for basic information: "For The New Rehabber" and "Simple Things That Make a Difference." They are intended for basic information or new ideas that are easily demonstrated with a short text and photos. All authors receive a $25 honorarium for each article accepted for publication or provide a one year membership or renewal.

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1. An original and two copies of the manuscript should be submitted to the Jan White, DVM, Editor, IWRC, 944 Anderson Dr., Suisun, California 94585, USA. The manuscript should include references, photo legends, and footnotes, and should be typewritten, double-spaced, if possible. The author's name, address, and phone number should be placed on at least the first page of the article so it can be readily identified and the author contacted. Please note that each article should be accompanied by an abstract and a set of key words. If tradenames are used for products mentioned the author must indicate whether the product is ™ or ®. A product table at the end of the article should give readers the name and address of the companies involved and generic name or category of product.

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3. Manuscripts are received with the understanding that they have not been accepted for publication elsewhere. *It is the author's obligation to obtain permission or reprint rights if the submitted copy has been published elsewhere.* All accepted manuscripts are subject to editing. Unused manuscripts will be returned.

4. A separate page should be submitted with a short synopsis of information on the background of the author. Included should be the name, address, rehabilitation organization, phone number of the author and a biographical paragraph. Where appropriate, please list any academic degrees, special training, or experience that is relevant to the article (i.e., physical therapy—RPT, etc.) Articles pertaining to areas of veterinary medicine that involve tasks or decisions requiring a licensed veterinarian (e.g., soft tissue or orthopedic surgery) require a veterinarian as either the author or a co-author.

5. Black and white photographs are preferred for use in the *Journal of Wildlife Rehabilitation*. When no other option is available, color prints can be converted but this is costly and to be avoided when possible. Photo credits should be listed with the photographs.

6. Acknowledgments are limited to people who have contributed to the article in a major way.

7. References should be noted in the text with the last name of the author and the year published (i.e., Leighton, 1983) and listed at the end of the manuscript in alphabetical order in the following style:

LEIGHTON, FA; Peakall, DB; and Butler, RG. 1983. "Heinz-Body Hemolytic Anemia from the Ingestion of Crude Oil: A Primary Toxic Effect in Marine Birds." *Science* 20:871-873.

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Editorial . . .

Peer Review: What is it and what does it do for our profession?

Peer-review is a process of submitting manuscripts to "outside" peers for critical review and comments before publication. Those comments are then reviewed by both the editor and the author(s) and then important additions, deletions, and clarifications are made to the manuscript before the final version is published. What are outside peers? Journey-level professionals that are not connected with your work in any way (e.g., they are not your mentors, co-authors, work supervisors, board members). In fact, in general, they are chosen outside of your organization for several reasons. The first is validity. Readers rely on peer-review to provide them with articles that are state-of-the-art but it is the reviewers who make sure that no leaps of faith have been made. They are the gatekeepers for our professional literature. Through their efforts with the authors, progress is made in a linear fashion with articles that build upon previous work and make logical or proven extensions forward. Reviewers perform their services without fee and understand the pivotal role that they play in advancing wildlife rehabilitation as a profession. The second reason is perspective. It is important to get someone who is not working in the author's environment to read the manuscript "cold" and see if it is written in such a way that the reader can readily understand the article, or in the case of original research, can repeat the work after reading the article as written.

What is the difference between peer-reviewing and having an advisory board? Well, an advisory board provides input upon request. Peer-review is a consistent process. Reviewers are asked to comment in a very specific, consistent and thorough manner. Authors participating in a reviewed publication know that they will be working with reviewers to produce the best publication. They expect that they will be presented with comments and that changes will be required. With an advisory board, members are often sought from a variety of specialities and often, comments are sought from the one specialist in the area of the article. The process for publication is more varied and may not involve any review.

How much peer-review is enough? Well, the *Journal of Wildlife Rehabilitation* solicits three outside reviewers in addition to the editor. We are amazed at how much more we pick up on with this level of review. The quality of the publication results from the level of review (and from the reviewers). The IWRC conference proceedings has a small editing budget and consequently less editorial work is performed. Reviews are solicited from one reviewer. Comments are sent to the author. The author is encouraged to incorporate or ameliorate the concerns of the reviewer but is not required to do so. The editors of both the journal and the proceedings are responsible for all production (typesetting, layout, author proofs, etc.). The *IWRC newsletter* is not reviewed for content or accuracy. It is intended as a communication organ for the membership, not for the purveyance of technical information and at the bottom of each page we write "not suitable for citation" to underscore this fact.

What are examples of reviewed publications in wildlife rehabilitation? NWRA's conference proceedings must meet peer review (2 reviewers + the editor) and the quality of their publication reflects their efforts. Recently, they have begun a peer-review process (1 or 2 reviewers) for animal care articles in their newsletter, the *NWRA*

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Erwinia Slime Flux May Cause A Temporary Color Morph in Juvenile Western Screech Owls, *Otus kennicottii*

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Abstract

Western Screech Owls, *Otus kennicottii* are typically uniformly grey, both as juveniles and adults. In this paper we report a temporary color morph observed in the Rio Grande valley of New Mexico. Roughly 10% of juvenile Screech Owls brought to Wildlife Rescue of New Mexico are red in color, later changing to grey as adult plumage appears. The red coloration may be associated with nesting cavities located in trees infected by *Erwinia nimipressuralis*.

Key words: *Otus kennicottii*, color morphology, red phase, cavity nesting, *Erwinia nimipressuralis*

Introduction

The Western Screech Owl, *Otus kennicottii* is typically uniformly grey in color and easily distinguished from the "red phase" birds of its eastern relative, *Otus asio*. In this paper, after a brief review of the distribution and morphological variation of *Otus* species, we report several cases of red *O. kennicottii* juveniles. Slime flux, caused by the common bacteria *Erwinia nimipressuralis* found in tree cavities, is highly alkaline and may cause oxidation of the feathers as well as severe burns to the feet and other parts of the body of these cavity-nesting birds. Feather oxidation and consequently red coloration of the plumage is temporary. As blood feathers emerge and juvenile plumage is replaced by mature plumage, *O. kennicottii* returns to its natural grey coloration. Symptoms of chemical burns and identification of the etiology of this temporary plumage color associated with birds nesting in Erwinia-infected cavities are also given.

Discussion

In 1983 the American Ornithologists' Union (AOU) divided the Screech Owl genus *Otus* into three species: *Otus asio* (Eastern Screech Owl), *Otus kennicottii* (Western Screech Owl), and *Otus trichopsis* (Whiskered Screech Owl). Eastern and western birds are differentiated primarily by call, though an expert eye can sometimes distinguish adults of the species by the width and position of horizontal striations on the breast crossing the wide, dark vertical stripes. These horizontal striations may also be diagnostic by their presence or, in the case of *O. kennicottii*, absence on the backs of the adult (Kaufman and Bowers, 1989). Some *O. kennicottii* in the Pacific Northwest have been described as brownish or reddish (Bent, 1938; Johnsgard, 1988), but the eastern species *O. asio* has two distinct color forms, grey and red, with a low frequency (<10%) of intermediate color morphs (Owen, 1963). This red color ranges from deep auburn to a beautiful strawberry blond. Though the genes for red coloration are dominant (Eckert, 1974), less than 50% of the birds are red morphs in many populations. Owen (1963) mapped the relative frequency of red Screech Owls in North America east of 104 degrees W longitude. He reported clinal geographic variation from north to south in rufous colored birds, with higher percentages (>75%) of red morphs occurring in warmer latitudes. Other studies have also suggested that the frequency and color distribution of Otus spp. is highly dependent on environmental factors such as humidity and temperature (Hrubant, 1955; Mosher and Henny, 1976). Gehlbach (1995) describes influences on color discrimination such as staining during bathing and seasonal wear in his discussion of juvenal plumage. This staining can be caused by mineral deposits and result in rusty or brownish yellow feathers on the legs, feet and belly.

The range separation of the Eastern and Western Screech Owl is still being studied, but the red phase of *O. asio* has been reported in most places throughout its range (Hrubant, 1955; Owen,

1963). *O. asio*, however, has not been reported in New Mexico. No rufous owls are known from San Antonio, Texas, to the Rio Grande valley, Mexico (Gehlbach, 1995). A rare intermediate red morph in the subspecies *O. kennicottii macfarlanei* was reported in Idaho and Montana (Fitton, 1993).

Observations

An examination of six *Otus kennicottii* juveniles and 43 adults in the collection at the Museum of Southwestern Biology in Albuquerque, New Mexico show all specimens of *O. kennicottii*, with one exception, to be uniformly grey. One adult collected in Silver City, NM in 1928 and labelled a "Mexican owl" appears a little brownish. None of the juveniles in the collection are even remotely brown. In 1983, however, a nestling raptor was presented to Wildlife Rescue of New Mexico in horrendous condition: bald head, bare legs, yellow eyes, and rust colored plumage under the down. Severe burns on the feet up to the hock further compromised its condition. The bird was clearly an owl, but what species of owl was not clear. Though it had many characteristics of a Burrowing Owl, the legs were not very long and it tended to sit in a round ball suggestive of a Screech Owl. As time progressed and the burns were treated it became apparent that the bare legs were, in fact, feathered. These feathers which should have come in grey were rust colored. As the bird gained vitality, ears or horns could be seen on each side of the bald head. Body plumage, however, was quite red and the bird, now recognizably a Screech Owl, became known as the "red owl". Since the rehabilitator was from the east coast, the color of the bird did not at first seem remarkable. The juvenile Screech Owl was treated, raised and released.

Interest in the "red owl" was expressed by Dr. J. Hubbard, then the endangered species biologist with the New Mexico Department of Game and Fish, who informed us that red phase Screech Owls do not occur in New Mexico. Since the owl had been released prior to our discussion, we were unable to provide a specimen. The following year, however, three nestlings were brought from the same location to Wildlife Rescue of New Mexico. One nestling was dead upon arrival, one died in hand, and the third survived to release. All three birds were red. We immediately contacted Dr. Hubbard for positive identification. The meeting was memorable: "Tell me this bird is not red." His response, "My, that bird is oxidized."

In the Rio Grande valley, *O. kennicottii* nest in cavities of old trees, especially in the *Populus fremontii* or Valley Cottonwood (Martin, 1980), the tree species containing the nest cavity of these birds. Western Screech Owls typically inhabit seasonally wet, upland drainages (Gehlbach, 1995). The entrance to the cavity was located 25-30 feet above the ground. The interior walls of the cavity were slippery, wet and accompanied by a foul odor (Brown 1984, pers. comm., Assistant Director, New Mexico Department of Game and Fish). The slimy exudation in and around the cavity is symptomatic of a common pathogenic bacteria, *Erwinia nimipressuralis*, commonly known as Slime Flux (Tattar, 1978; Horst, 1979). *E. nimipressuralis* typically infects tree species of Elm, Poplar and Willow, all common nesting sites of *O. kennicottii* and other cavity-dwelling birds. The bacteria infect fresh wounds and thrive in the water-conducting xylem tissue of trees. Increases in the xylem due to the fermentation of plant tissues by *E. nimipressuralis* causes a condition called fluxing, a forcing of the sap out from the interior of the tree through cracks and wounds. The flux can wet large areas of bark, providing a rich medium for other bacteria and yeasts to grow. These large colonies of actively growing yeasts and bacteria are what cause the foul odor associated with slime flux. Collectively, the flux, sap, bacteria, yeasts and fermenting exudate produce a very alkaline solution.

To determine the pH environment of nesting cavities, we collected three different Slime Flux products from infected Cottonwood trees (Table 1): flowing flux (FF), decayed wood subjected to flux (DW), and pooled Slime Flux at base of tree (SF). Samples were taken from three typically Erwinia-infected Cottonwood trees and pH levels of each pure product were recorded using a standard electrode pH meter. In order to gain better accuracy of pH levels, each sample was diluted (1:10 and 1:100) for improved meter readings. To obtain the pH of decaying wood samples, decayed wood was eluted with distilled water in equal amounts by weight. For Tree 3, a sample of decayed wood could not be gathered because the cavity was not readily accessible without cutting into the tree. Table 1 shows a range of 7.74 to 10.14 of pH levels across trees and products. The most alkaline samples were those from the rotting wood itself, the very material on which young Screech Owls would be resting. A polycarbonate mouse cage placed under the drip of one flux tap has, over the years, shown signs of slow corrosion. Long tracks have been eaten in the sides of the box where the flux has been held by a fallen twig or leaf.

TABLE 1: pH levels of *Erwinia*-infected nesting cavities

Tree	Material	SAMPLE pH		
		1:0	1:10	1:100
1	FF	7.74	8.98	8.88
2	FF	8.26	8.61	8.54
3	FF	8.49	8.73	8.67
1	DW	8.20	10.14	9.90
2	DW	7.93	9.61	9.16
3	DW	n/a	n/a	n/a
1	SF	8.29	9.49	8.52
2	SF	9.44	9.42	9.25
3	SF	8.95	8.75	8.31

***FF** = sap flowing directly from tree; **DW** = flux-soaked rotten wood from inside cavity; **SF** = fluid from the puddle of exudate at base of infected tree.

Over time, especially as the season progresses and xylem pressure increases naturally and by infection, the exudate collects in these cavities, creating a dangerously alkaline environment for nestlings of cavity nesting birds. Eggs may be laid in the cavities before active fluxing of the diseased tree begins. By the time the eggs hatch, however, varying amounts of this flux can collect in the nest. The alkaline solution pools in the bottom of the cavity and begins to oxidize and bleach anything in contact with the flux, including the bark of the tree (E.P. Elliston, pers. observ., June, 1996). Feathers develop a reddish color and skin is burned. As the birds develop, the high pH of the cavity and resulting chemical burns may force the nestlings out of the nest prematurely.

We soon realized that the condition of this cavity may not be unique and began to search for evidence of bleaching and burns in other juvenile Screech Owls brought to Wildlife Rescue. Over the past 10 years, Wildlife Rescue of NM has received an average of 6.5 juvenile Screech Owls each year for problems ranging from displaced nestlings and premature fledgings to various injuries such as accidents with cars and cats. Since 1983, we have treated seven birds for burns on the feet, all of which accompanied the characteristic red colored plumage. Roughly 10% of all immature *O. kennicottii* brought to Wildlife Rescue of New Mexico had fledged prematurely from cavities containing slime flux. As blood feathers grow in and mature feathers replace juvenal plumage, the grey color replaces the temporary red plumage. The red feather color seems to be a result of bleaching by the very alkaline slime flux caused by *E. nimipressuralis* of the infected tree.

In addition to bleached feathers, burns on the feet are common and sometimes respiratory problems develop. Since Screech Owls have this condition relatively often, we began to look for similar conditions in other cavity nesting bird species. For example, *Falco sparverius* (American Kestrel) frequently nests in cavities of *Populus* spp. Although we receive many more nestling/fledgling *F. sparverius* than *O. kennicottii*, very few *F. sparverius* have actually been presented with respiratory problems and/or chemical burns. Only one documented case of a nest containing five F. *sparverius* siblings, four of which expired from respiratory complications, has been found in our records. Perhaps kestrels are more meticulous about choosing a nesting cavity than Screech Owls. It is possible that they are more resistant to the effects of the alkaline environment, but judging from the high mortality in the one observed nest, it is likely that this is not the case.

Conclusions

Adult and juvenile *Otus kennicottii* are typically uniformly grey. In 1983, Wildlife Rescue of New Mexico was presented with an injured juvenile Screech Owl from the Rio Grande valley area of New Mexico. This bird showed symptoms of chemical burns resulting in a temporary red color phase. Since that time, roughly 10% of juvenile Screech Owls brought to Wildlife Rescue of New Mexico have been red in color, later changing to grey as adult plumage emerged. The red coloration is associated with nesting cavities located in trees infected by *Erwinia nimipressuralis*. To avoid misidentification of red phase Screech Owls in the southwest, observers of this color should assess the likelihood of the bird being a juvenile expressing the phenomenon described above. Rehabilitators, when faced with a red fledgling, should check for chemical burns and respiratory difficulties in the bird. It would be interesting to know how many other such cases of temporary color morphs exist in riparian forests of the Southwest.

Acknowledgements

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Errata:

The email address listed for Corey Bradshaw in Vol. 19 # 2, "Scaring Macaws to Survive", was incorrect (i.e., was missing the "e" in Corey). The correct address is: corey.bradshaw@stonebow.otago.ac.nz. We regret any confusion that the omission may have caused.

Author Profiles

Elizabeth P. Elliston was born in London, England and grew up in Massachusetts. She earned a Master of Science degree from Johns Hopkins University School of Hygiene and Public Health (JHSHPH). She has worked in the field, in Maryland, Tchad and India, with teams from JHSHPH and began rehabilitating wildlife in the 1980s with Wildlife Rescue of New Mexico (NM). She also serves on the Citizen's Review Committee for the NM Habitat Program, Central Region, and as the IWRC State Representative for NM.

Lelia C. Orrell is a Ph.D. candidate at the University of Massachusetts / Boston, Department of Environmental Biology, where she is conducting research on ecosystem management and conservation biology of rare and endangered temperate, coastal plain plants. She holds degrees of Bachelor of Science from the University of Massachusetts / Boston and an Associates degree in Horticulture from Massachusetts Bay Community College. Her work has included research on Botrytis infection and bud dormancy factors in Paeonia spp., and conservation and population genetics of tropical tree species. Lelia is on the board of the Weston Forest and Trail Association, a local land conservation group. She also owns and operates a small flower farm, producing cut flowers for the wholesale cut flower market.

IWRC Jobline

Wildlife Rescue, Inc.
4000 Middlefield Rd. Building V
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Full-time Animal Care Coordinator

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The What, Why, When & How of Avian Blood Transfusions: A Discussion of Protocols and Literature Review

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Abstract

Raptors (and other animals) present in a variety of conditions with a variety of underlying causes. If a bird has experienced an extended period of starvation, is critically anemic, has exhausted its immune defenses, or has suffered an extremely high volume of blood loss, a blood transfusion may provide some extra resources for the dangerously ill patient. Various avian protocols and procedures for cross-matching samples are described.

Key words: blood transfusion, autologous, homologous, heterologous, plasma, whole blood, packed cell volume (PCV), serum protein, total solids (TS), intravenous, intraosseus, regenerative anemia

Introduction

Raptors, or any wildlife, do not automatically walk up to a caring human and ask to be taken to a rehabilitation center. Unless found immediately upon injury, a raptor must often be injured, ill, or starving for an extended period of time before it is weak enough to be observed and subsequently caught. Sometimes the actual injury is the least of the patient's problems: by the time the bird is received at the rehabilitation center, it is often dehydrated, emaciated, anemic, overrun with parasites and has a systemic infection—as well as, say, a fractured metatarsus. When reviewing the scientific and medical literature related to giving avian blood transfusions, it is clear that while some facts are known, there are still large gaps in the available information. As a result, veterinarians working with Cascades Raptor Center, other rehabilition centers, and oil spill responses have developed protocols for providing blood transfusions, which are the basis for this discussion. As more information becomes available through experience, standardized avian protocols will undoubtedly be developed.

WHAT is a blood transfusion?

A blood transfusion is the addition of blood, from a donor animal, to a recipient. It can be of whole blood, directly as drawn from the donor, or as plasma (or even as other components, which we will not be discussing here). *Plasma* is the clear fluid portion of the blood in which particulate

components are suspended; *plasma* is distinguished from *serum*, which is the cell-free portion of the blood from which the fibrinogen has been separated by clotting. (Blood & Studdert 1988; Dorland's 1974). When you spin a blood sample in a non-heparinized container, the clear fluid that results is serum; when you use an anticoagulant, the clear portion is plasma. An *autologous* transfusion is the transfusion of the animal's own blood, taken, for example, some days prior to a surgery where a lot of bleeding is anticipated (this is popular with humans in order to avoid the risks of transfusions of possibly contaminated blood from someone else). *Homologous* transfusions are those taking blood from one member of a species to donate to another of the same species; *heterologous* is, obviously, cross-species transfusions. In humans, critical attention is paid to blood type and, therefore, compatibility. In animals, much recent work has led to the identification of eleven blood group systems and fifteen or more blood group factors in the dog alone, with the expectation that the number of both will increase with on-going research. All transfusions carry some degree of risk due to incompatibility. Animals have different blood types and complete compatibility is nearly impossible (Kasper 1996). Although it is felt that the blood grouping system in birds is probably more complex than than found in most mammals, among birds, only chickens and turkeys have well-deciphered blood groups, with 11 separately inherited blood-group systems in chickens and 7 in turkeys (Bell & Freeman 1971, Fowler 1986, Jain 1986).

In birds, cross-matching and compatibility work-ups can be done but often are not, with the feeling based on research to date that, as a *one-time therapy*, the benefits outweigh the possible antibody reactions. Antibodies lead both to destruction of the transfused cells and possibly to subsequent transfusion reactions. Those reactions, it has been felt, could cause a severe impact and possible death with any subsequent transfusions (Harrison 1986, Jain 1986, Altman 1982), although the period before which such a reaction might be expected, or after which a subsequent transfusion might be safe, is controversial.

Studies performed with birds indicate that antibodies develop within the first several days following a transfusion (Lawler and Redig 1982) and one study indicates that in heterologous transfusions, red blood cells survive less than 24 hours (Sandmeier *et al.* 1994). In autologous and homologous transfusions, red blood cells survive longer (see Table 1). This information is used by veterinarians to estimate how long the effects will last and when a second transfusion puts the animal at risk for an antibody response. Lawler and Redig (1982) studied the hemagglutination response in three raptors transfused with sheep blood and found that the antibody response, as measured by microhemagglutination assay titers, peaked four days after sensitization (transfusion) and remained increased for seven days, and returned to normal three weeks after sensitization. Thus, if a second transfusion is going to be administered, earlier is probably better. The Raptor Center at the University of Minnesota regularly and successfully utilizes second transfusions, either on Day 2 (if the packed cell volume continues to fall more than can be accounted for by rehydration) or Day 3.

Table 1. Red Blood Cell Survival Time in days.

Study	Number of Animals	Transfusion Type	Estimated RBC Survival Time (days)*	Species
Hoopes et al.	n=4	Autologous	~ 19	Ring-necked Pheasant
	n=6	Homologous	~2-6	Ring-necked Pheasant
Sandmeier et al.	n=2	Autologous	35.1; 26.8	Red-tailed Hawk; pigeon
	n=1	Homologous	7.1	Pigeon
	n=1	Heterologous	0.51	Pigeon to raptor

* Defined as the point at which it is estimated that nearly all RBC's have been removed from circulation as determined using ^{51}Cr labelled RBC's. (Sandmeier's techniques for cell labeling were from: International Committee for Standardization in Haematology, "Recommended method for radioisotope red-cell survival studies," *Br J Haematol* 1980; 45:659-666; and Moroff, G, RP Sohmer, LN Button, et al, "Proposed standardization of methods for determining the 24-hour survival of stored cells," *Transfusion* 1984; 24: 109-114.)

WHY a blood transfusion?

There are times when the immunological system of an animal is exhausted or the animal is so debilitated that its own ability to produce blood cells is compromised. Transfusions are probably most often used with severely anemic animals, whose volume of circulating red blood cells is so low that the ability to carry oxygen to the cells is at risk. Milder degrees of anemia are usually treated more conservatively, with parenteral fluids and iron dextran therapy (Bos *et al.* 1990). When the buffy coat exceeds 1% and iron dextran is not recommended (due to the suspected presence of an infection), the application of 20 mg/kg IM of B-complex once a week has a positive effect on speeding up red blood cell formation (Kasper 1996). Transfusions have also been used during or after surgery with excessive hemorrhage (e.g., removal of a large tumor) to replace lost blood volume without putting pressure on the animal's own bone marrow to produce new cells (Ritchie *et al.* 1993). According to a study with pigeons, severe hemorrhage (loss of 70% of total blood volume) in an otherwise healthy animal may not alone merit a transfusion (Bos 1990) and, in fact, there is some indication, based on studies with chickens, Japanese quail, and ducks, that a healthy bird can lose as much as 30% of blood volume with minimal clinical problems, recovering its normal volume of red blood cells within 72 hours (Ploucha *et al.* 1981; Djojosugita *et al.* 1968).

Although acute hemorrhage does not theoretically change PCV, it is rare that such hypovolemia is the only problem; most practitioners use transfusions in response to anemia rather than simply hypovolemia. With the severely anemic bird, some practitioners feel that a homologous (or even a heterologous) blood transfusion could buy time for other supportive therapy to work, e.g., B-complex, iron dextran, warmth, food.

WHEN is a blood transfusion indicated?

Different rehabilitation centers and veterinarians have different mileposts for when a transfusion might be indicated, taking into consideration a range of criteria. At *The Raptor Center at the University of Minnesota (TRC)*, transfusions are considered essential when PCV on intake is less than 20% (bearing in mind that PCV will typically fall with rehydration) and Total Solids are less than 1 g/dL (Arent 1996). *The Wildlife Clinic at Tufts School of Veterinary Medicine* uses transfusions when PCV is low and decreasing. When PCV is low but stable, rather than using a transfusion, they try to supply optimal nutrition and deal with the bird's other medical problems. When Total Solids are in the range of 1-1.5 g/dL, Tufts considers doing just a plasma transfusion or they supplement with iron dextran or hetastarch (Pokras 1996). Hetastarch is a synthetic colloid used to restore normal colloid pressure and plasma volume; bolus applications of 10-15ml/kg TID for up to 4 treatments are tolerated with no ill effects; however, consecutive infusions should be used with caution in the presence of congestive heart failure or renal failure (Stone & Redig 1994)

Working primarily with oiled seabirds, Jan White, DVM, recommends a blood transfusion when the PCV is less than 13% or a plasma transfusion when the Total Solids are less than 1 g/dL. (White 1992). At *Cascades Raptor Center (CRC)*, transfusions are performed when the PCV is less than 15% and Total Solids are less than 1 g/dL. Other health criteria reviewed by the authors include general attitude, blood pressure and circulating volume (a transfusion is difficult when veins are too collapsed to enter easily). Although anesthesia can usually be avoided with current techniques, somewhat prolonged restraint is necessary and the bird must be able to withstand the stress.

Table 2. Multipliers for Calculation of Amount of Blood Needed For Transfusion Per Pound of Recipient Cat, Based on Post-Transfusion PCV OF 18%

PCV of Recipient	PCV of Donated Blood (including anticoagulant)										
	30%	32%	34%	36%	38%	40%	42%	44%	46%	48%	50%
4%	16.3	15.3	14.4	13.6	12.9	12.3	11.7	11.1	10.6	10.2	9.8
6%	14.0	13.1	12.4	11.7	11.1	10.5	10.0	9.5	9.1	8.8	8.4
8%	11.7	10.9	10.3	9.7	9.2	8.8	8.3	8.0	7.6	7.3	7.0
10%	9.3	8.8	8.2	7.8	7.4	7.0	6.7	6.4	6.1	5.8	5.6
12%	7.0	6.6	6.2	5.8	5.5	5.3	5.0	4.8	4.6	4.4	4.2
14%	4.7	4.3	4.1	3.9	3.7	3.5	3.3	3.2	3.0	2.9	2.8

Table is taken from Kirk & Bistner *Handbook of Veterinary Procedures & Emergency Treatment*, 4th Ed., p. 624

As used at CRC, transfusion is essentially an attempt to ward off otherwise certain death - sometimes it works, often it does not. Birds in the category requiring transfusion (generally extremely emaciated) often have a multiplicity of problems (blood parasites, aspergillosis, pericarditis, gape worm, internal injuries, suspected Warfarin poisoning), many of which may not be apparent until necropsy. At oil spills, Dr. White has occasionally used a transfusion to 'perk up' seabirds with hemolytic anemias, with the goal of inducing self-feeding, a necessary adjunct to recovery and release and one which significantly reduces stress by eliminating force-feeding or tube-feeding.

HOW MUCH blood do you use?

The goal of a whole blood transfusion is primarily to raise PCV and/or Total Solids values. There are several different ways to calculate the amount to be transfused. A chart (based on felines) provides an idea of the volume necessary to raise PCV from intake levels to a desired target level of 18%, based on the PCVs of the donor and recipient (see Table 2).

Example: 1200 g. Great Horned Owl

PCV of recipient	10%
PCV of donated blood	42%

1. Convert recipient bird's weight to lbs: 2.6 lbs
2. Multiply 6.7 (from Table above where donor PCV column and recipient PCV row intersect) times 2.6 lbs = 17.42 ml needed to raise the recipient PCV to 18%.

The above formula is taken from Kirk & Bistner *Handbook of Veterinary Procedures & Emergency Treatment,* 4th Ed., p. 624)

Similar results can be reached by using another formula for calculating the volume of blood to transfuse:

[Recipient Weight (lbs) x 40 ml/lb x (PCV desired - PCV of recipient)] ÷ PCV of donor

Using the great horned owl example from the table 2, this would result in

[2.6 lbs x 40 ml/lb x (.18-.10)] ÷ .42 = 19.8 ml
(Formula provided by Animal Blood Bank, Dixon, CA.)

The above formulae assume there is a free availability of donor blood, which is not always the case, especially when attempting a homologous transfusion. Besides the ideal amount needed to raise the recipient's PCV to a target level, the ability of the donor bird to supply that amount must be considered. In practice, at least at CRC, the amount of blood given is based on the size of the donor, rather than the true amount needed by the recipient. That is, if the great horned owl of the example above needs a transfusion, but only one donor great horned owl, which weighs 1700 gms, is available, CRC would only take some 15 ml. The typical assumption of circulating blood volume is 10%, in ml, of the body weight in grams: so it is assumed that a 1700 gram great horned owl has some 170 ml of circulating blood, and that it is safe to take some 10% of that, or 17 ml. CRC usually backs off slightly from that norm and would take 15 ml. According to one study (Hoopes 1978) the blood volume of pheasants averaged closer to 5%, in ml, of body weight in grams. However, given the other studies referenced here (Bos 1990, Ploucha et al 1981, Djojosugita *et al* 1968), it is probably not harmful to the donor to take more than the estimated 10%.

HOW do you do a blood transfusion?

Blood transfusions can be done either IV, in a variety of veins, or IO (intraosseous), in a variety of bones, either hollow or marrow (Ritchie *et al.* 1990). CRC's preference is IV, in order to eliminate the necessity for anesthesia, and the authors usually use either the jugular or cutaneous ulnar vein. Dr. White, working primarily with seabirds, uses the metatarsal vein. It is possible to do a cross-matching procedure; though neither CRC nor TRC routinely do them, Dr. White does (see below for both 'washed' and 'unwashed' procedures). CRC has generally found that drawing blood using syringes larger than 3 ml results in so much pressure that the donor bird's veins collapse with each draw back on the syringe, causing the process to take longer than necessary, as the veins must refill before the next draw-back is possible. Heparin is typically the anticoagulant of choice, rather than EDTA, in order to avoid excessive binding of calcium (Harrison 1986). Tufts uses a citrate buffer and an in-line filter to avoid microclots in circulation. The Animal Blood Bank recommends a Hemo-Nate® blood filter when transfusing 50 cc or less; the Hemo-Nate® is a bi-directional filter that fits on any syringe or venoset and comes in several sizes. The 18 micron size is used in many small birds (Kasper 1996).

Following are two possible protocols:

Cascades Raptor Center: The protocol developed by CRC involves three people and eliminates the need for anesthetics.
Donors are typically permanently disabled education birds, conspecifics of the recipient (healthy pre-release rehab conspecifics have also been used) with PCV's of at least 40%.
Equipment and anti-coagulant: Both drawing blood from the donor bird and transfusing into the recipient, a 23 gauge or larger butterfly catheter is used; because the entire length of the catheter, not just the syringe, is heparinized, CRC uses a 1% heparinized saline solution rather than heparin alone; the saline solution is drawn into the catheters and expelled (leaving it in the hub), as well as into the requisite number of 3 ml sterile syringes.
Cross-matching: no
Technique: One person (A) immobilizes the bird, whose head is hooded or covered, on its back, holding the feet with one hand; one person (B) uses both hands to place and hold the catheter; A uses his/her other hand to draw back on the syringe, slowly filling it with donor blood; person C stands ready to replace each syringe as it fills with an empty one, and rocks the filled syringes to mix with heparin and help keep them from clotting. Once the syringes are full, person A holds off the vein, while B removes the needle, and C keeps rocking the filled syringes. Once the donor bird is checked for clotting and returned to a carrier, the process is essentially replicated on the recipient bird: with A immobilizing the bird and slowly infusing the blood, once B has placed and is holding a new sterile, heparinized butterfly catheter in the recipient's vein, and C is rocking syringes and replacing empty syringes with full ones on the hub of the butterfly catheter.
Medications: Immediately before or after transfusion, the recipient bird receives IM injections of Benadryl® (an antihistamine) at 2 mg/kg body weight and dexamethasone (a corticosteroid) at 1 mg/kg body weight to limit allergic response.
After-care: Iron dextran is given IM q 7 days at 10mg/kg, unless an infection is suspected (due to the fact that iron is a growth factor for bacteria) or, CRC's preference is to give oral Pet-Tinic® in tube-feeding formula or injected into food; injectible B-complex is given on admission; antibiotics are almost always administered; powdering for external parasites is done at intake to limit possibility of spreading blood parasites or disease to other birds in care; treatment for internal parasites is done after the bird is stable; typically no treatment is done for blood parasites. At TRC in Minnesota, birds are placed on antibiotics and antifungals (typically itraconazole: Sporanox® by Janssen Pharmaceuticals) after transfusion and are given iron dextran and vitamin B complex on admission.

Dr.White's protocol, used primarily at oil spills:
Donor: PCV greater than 40%; Total Solids greater than 3 g/dL; Blood cross-matches with receiver; donor birds are pre-release or permanent care birds, preferably of the same species. Bald Eagles served as successful donors for anemic seabirds in the *Exxon Valdez* oil spill.
Equipment and anticoagulant: 22 gauge needle to draw, heparin flush; 24 gauge butterfly to infuse.
Cross-matching: Yes, unwashed method.
Amount: Amounts vary based on the need of the recipient and the size of the donor. Two goals are possible: giving enough blood to "perk up" the bird and induce self-feeding OR giving enough to raise the PCV to 18%. In the case of a 1000 g Common Murre, the first goal (of "perking up" to induce self-feeding) would be met with 4 ml's of blood and the second would require 10-11 ml's, depending on PCV's of donor and recipient. A small auklet would be given 2-3 ml's to "perk up." Self-feeding birds have a much better chance of recovery.

If the recipient is not severely anemic but has little or no total solids, a plasma transfusion may be helpful. The guidelines for amounts are the same as for whole blood, though it is obviously going to be more difficult to get that much plasma. Be sure to cross-match the plasma to be given with a whole blood sample from the recipient.
Technique: Draw up the appropriate amount from donor; immediately remove the 22g needle and place a 24 gauge butterfly onto the syringe; infuse the donor blood into the recipient slowly, and observe for any sign of a transfusion reaction (e.g., collapse, increase in rate of breathing). Dexamethasone Sodium Phosphate (1-4 mg/Kg IV) may or may not be used in conjunction with a transfusion to reduce the possibility of a reaction. The use of a citrate buffer and in-line filter are encouraged.

WHAT is compatibility & how do you test it?

There are a variety of opinions about the need for cross-matching. The general assumption is that a cross-match for a first-time transfusion is probably not necessary in birds: that it is safe to do one transfusion without concern of an antibody reaction and, in fact, TRC routinely does two transfusions, a day or two apart, without cross-matching and without using an antihistamine or corticosteroids to reduce reactions. Some rehabilitators do cross match each time, whether

doing a homologous or a heterologous transfusion. Besides compatibility information, outside the parameters of a controlled experiment, one must consider practicality. Rehabilitators are working with extremely debilitated, stressed birds as recipients: The cost to the bird of the extra capture, handling, and restraint may outweigh the benefits of cross-matching, especially given the possible lack of accuracy of the unwashed method and the necessary time and expense of the washed/incubation method, which requires some equipment that may not be easily available to a wildlife rehabilitor (see below).

Methodology - Although a study cited by Harrison (1986) indicates that cross-matching with unwashed red cells and sera is not an accurate means of determining compatibility (the study was limited to heterologous transfusions - see Altman, RB, "Heterologous blood transfusions in avian species." Proceedings of the Annual Meeting of the AVMA, San Diego CA 1983, pp 28-32), unwashed cross-matching is by far the simpler means. In one of the major studies cited in all other work, (Bos 1990) performed both major and minor crossmatch tests utilizing a washed cell-incubation method - but transfusions were performed, whether heterologous or homologous, regardless of compatibility results, with no apparent transfusion reactions or impact on study results.

UNWASHED CROSS-MATCHING PROCEDURE

1. **From receiver:** draw up 2 plain (non-heparinized) capillary tubes of blood.
2. Spin both tubes in a microhematocrit centrifuge for 2-4 minutes; break tubes and place serum onto one clean microscope slide.
3. **From donor:** draw up 2 plain (non-heparinized) capillary tubes and spin as above (2.) Major cross match: take a drop of fresh blood (do not place in a heparinized hematocrit tube) and immediately place onto the recipient's serum on slide. Minor cross match: take a drop of donor plasma and mix it with a drop of the recipient's blood.
4. Stir gently and watch the reaction under a microscope (low power) if available. Observe grossly or with a microscope for red blood cell clumping that does not undo when stirred. *If the recipient's red blood cells agglutinate (clump and won't separate) when mixed with the serum of the recipient, the donor's blood is NOT compatible with the recipient's (e.g., it is a major cross match failure). If only the minor cross match agglutinates indicating a minor incompatibility, a veterinary judgement must be made depending on the availability of any other options.*

WASHED CROSS-MATCHING PROCEDURE (Jain 1986)

Materials Needed:

1. 2 ml EDTA anticoagulated blood from recipient and 1 ml of donor blood
2. Test tubes (75 mm x 10 mm) and rack
3. 0.9% saline solution
4. Serofuge (or serum centrifuge) that spins at a fixed rate of 1,000 G.
5. 1 ml. pipettes and pasteur pipettes and bulbs

Procedures:

1. Spin recipient and donor bloods in a serofuge or other appropriate centrifuge for 1 minute (use 75 mm x 10 mm test tubes).
2. Remove plasma to prelabeled test tubes.
3. Make a saline 2% red cell suspension for recipient and donor by adding 0.02 ml packed red blood cells to 0.98 ml saline or by drawing up the packed red blood cells with a pasteur pipette to the point where the pipette starts to widen. Add this to a full tube of saline. Label tubes:
   a. Recipient plasma (recipient 2% cells)
   b. Donor plasma (donor 2% cells)
4. Wash cells three times with saline and each time resuspend washed cells in an equal volume of saline solution.
5. Cross match tubes
   a. Major cross match: 2 drops recipient plasma plus 2 drops donor 2% red cell suspension. Label tube: Major and donor ID.
   b. Minor cross match: 2 drops donor plasma plus 2 drops recipient 2% red cell cell suspension. Label tube: Minor and donor ID.
   c. Control tube: to check for auto agglutination: 2 drops recipient plasma and 2 drops recipient 2% red cell suspension. Label tube: Auto.
   d. Shake rack of tubes and incubate 30 minutes at room temperature.
   e. Spin serofuge one minute.
6. Results: Visually check "button" of the cells in the test tubes for signs of agglutination.
   Gently tap the tubes and examine the supernatant (liquid fraction) for signs of hemolysis (broken red blood cells will make the solution have a reddish hue). If no agglutination is observed, transfer sev-

eral drops to a microscope slide and examine under low power of the microscope. Significant hemoloysis and/or agglutination in one or both of the cross-matched tubes but not in the controls indicates an incompatibility and the need to choose a new donor.

Summary

Blood transfusions have been used in a number of cases, from 'perking up' oiled seabirds with hemolytic anemias to help induce self-feeding, to giving an emaciated bird enough of a head-start to help its own defenses start regenerating. Obviously, a facility must have access to donor birds in order to do transfusions. This presupposes either a large operation, with a fairly dependable supply of healthy, pre-release rehabilitation birds as donors, or a substantial education program, where donors can be found among the healthy, permanently non-releasable birds. Although this may not apply to all readers, it is the authors' hope that this will provide a workable idea to be used within the rehabilitator's arsenal when the right situation arises.

In the course of providing care to oiled birds showing signs of regenerative anemias during the *Exxon Valdez* oil spill response, six seabirds with intake PCV's between 8-16% and total solids of 1 g/dL or less were given whole blood transfusions and three recovered (White, 1992). (The bird with an initial PCV of 16% dropped below 13% upon rehydration.) The recovery time was as little as 11 days. Without this treatment, it is doubtful that these birds would have recovered.

CRC has provided blood transfusions to a variety of birds, from a brancher screech owl to half-grown barn owl and red-tail nestlings, to adult red-tails, and great-horned owls. Of 10 cases reviewed, two survived to release, 1 was euthanized, and 7 died, including one during the transfusion; typical presenting complications were massive internal parasites (coccidia, ascarids, capillaria, gape worm), as well as blood parasites (leucocytozoan, hemoproteus, plasmodium, microfilaria); necropsies revealed aspergillosis, pericarditis and necrosis of lung tissue, gape worms, kidney problems. The two birds (great horned owl adult and screech owl brancher) that survived to release had intake PCV's/Total Solids of 12%/1.0 g/dL and 10%/0.5 g/dL, respectively, and would not, it is felt, have survived without the transfusion. The red-tail hawk nestling that was euthanized showed improvement through Day 7, with decreased blood parasites and steadily improving blood values (from PCV/Total Solids of 20% and 0.9 g/dL to 34% and 4.8 g/dL); but was euthanized on Day 16, due to progressive weakness and radiographs showing an enlarged liver and opaque airsacs; necropsy revealed aspergillosis and severe pericarditis.

It was not that long ago that recommendations could be found in the veterinary literature for euthanasia of these types of cases, in fact where Total Solids were below 2.3 g/dL, because the prognosis was considered grave (Harrison 1977); wildlife work with emaciation protocols and transfusions has had a hand in changing that.

Products Mentioned in the Text

Benadryl®: Park-Davis, a division of Warner-Lambert, Morris Plains, NJ 07950.

Citrate Buffer: Citrate Phosphate Adenine Solution, USP (CPDA-1), Fenwal, Division of Travenol Laboratories, Inc., Deerfield, IL (mix 1 part anticoagulant to 5 parts blood)

In-line Filter: Blood Component Infusion Set, Fenwal, Division of Travenol Laboratories Inc., Deerfield, IL)

Hemo-Nate Blood Filter®: Manufactured by Gesco, San Antonio, TX and distributed by Animal Blood Bank, P.O. Box 1118, Dixon, CA 95620-1118 (916) 678-7350 at a cost of $3.65 in 3 sizes: 18, 40 and 170 microns.

Pet-Tinic® Oral Iron Supplement: Pfizer Animal Health, 812 Springdale Dr, Exton, PA 19341 (800) 366-3424.

Hetastarch Product: Hespan®, Dupont Pharmaceuticals, Wilmington, DE.

Iron dextran injectible: 100 mg/ml; Phoenix Pharmaceuticals, 4621 Easton Rd., St. Joseph, MO 64503.

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Author Profiles

Louise Shimmel has been a state- and federally licensed wildlife rehabilitator for 10 years, currently specializing in raptors. She founded and has been director of the Cascades Raptor Center for 5 years and has worked with over 1000 birds of prey. Louise has served as assistant editor of the Journal of Wildlife Rehabilitation for several years, and has been on the IWRC Board of Directors, including serving as president, vice-president and treasurer, for 7 years.

Jan White, DVM has been active in the field of wildlife rehabilitation since 1975 when she founded the Suisun Marsh Natural History Association which built and operates a 2,000 sq. foot wildlife care facility. Following her graduation from veterinary school, she served as operations manager and staff veterinarian for the International Bird Rescue Research Center, Berkeley, CA. She returned to the University of California in 1992 where presently she is an assistant researcher at the Institute of Toxicology and Environment Health studying the effects of oil on birds. Jan served as the Executive Director of IWRC for 8 years and currently is the editor for the Journal of Wildlife Rehabilitation.

Kathy Snell, DVM is a graduate of the University of Minnesota and worked extensively at the Raptor Center located there. During her undergraduate years, she volunteered at the UC Davis Raptor Center and the Northwoods Wildlife Center in Wisconsin. She continues to see a lot of wildlife, especially raptors, in her private practice.

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Aspergillosis——The Silent Killer

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Abstract

Fungal infections caused by *Aspergillus spp.* are difficult to detect and treat. The type of infections seen, susceptible species often affected and the diagnostic tests required to detect this disease will be addressed. Treatment programs for ill birds and preventative programs for those birds in high risk situations will be outlined.

Key words: Aspergillosis, susceptible species, high risk situations, diagnostics, treatment, prevention

Introduction

One of the most severe fungal infections seen in birds today is caused by *Aspergillus spp. A. fumigatus, A. flavus, A. glaucus, A. niger and A. nidulans* have all been isolated from birds with A. *fumigatus* being the most common. Aspergillus, along with other fungi, grows readily in decaying organic matter (old feed, feces, vegetation, etc.) and is enhanced by damp conditions with poor ventilation. A ubiquitous organism, aspergillus fungal spores formed during development are very resistant and easily spread throughout the environment. These spores may remain dormant for an extended period of time and may become a problem under certain conditions. While this fungus can cause disease in any bird species, rehabilitators working with raptors, waterfowl, penguins and hummingbirds may see this problem more often.

Modes of Infection

Birds in good health and clean environments with excellent ventilation are not candidates for a fungal infection. Predisposing factors that may enhance an aspergillosis infection include malnutrition, pre-existing disease (bacterial, viral, parasitic), unsanitary environmental conditions, stress and immunosuppression. There is a relationship between environmental humidity, spore concentration and the onset of pulmonary disease. Dry, dusty conditions interfere with the normal mucociliary action of the respiratory epithelium and predispose the bird to respiratory aspergillosis. In contrast, nesting material with organic debris that becomes moist, stimulates mold growth and can contaminate incubating eggs or nestlings. Aspergillosis most commonly affects the respiratory system of birds (air sacs, trachea, syrinx, bronchi, lungs) and rarely the gastrointestinal system (Redig 1981, Stroud 1982, Tsai 1992). Three stages of the disease have been documented (Redig 1981, Redig 1987).

Acute aspergillosis is fatal within one to seven days. In this situation, the bird has inhaled an overwhelming number of fungal spores. Fungal lesions may appear throughout the lungs and air sacs giving the clinical signs of pneumonia. Other organ systems may also be involved. The clinical signs come on suddenly with the bird showing "sick bird syndrome", i.e., fluffed and depressed.



Figure 1. A granulomatous aspergillosis lesion of the primary bronchi.

Subacute aspergillosis is slower to develop, often taking one to six weeks before clinical signs appear. The respiratory tract is again the target organ with granulomatous type lesions developing. It is not uncommon to find involvement of the trachea, syrinx, bronchi or air sacs with this stage of infection (Figure 1). Clinical signs may include a voice change, respiratory wheeze or increased respiratory effort. Some birds may present with vague signs of inappetence, lethargy and increased thirst. Most birds are in good flesh until the last stages of an aspergillosis infection.

The chronic form of aspergillosis may take weeks or months to develop and usually affects the lower respiratory tract (Figure 2). It is important to point out that aspergillosis infections, although primarily affecting the respiratory system of birds, may spread to pneumatized bone or the peritoneal cavity. Isolated cases affecting the mouth, central nervous system, eye, kidney, adrenal glands and aorta have been reported. Clinical signs of aspergillosis may include lethargy, depression, anorexia, voice change, wheezing, dyspnea or prolonged recovery of normal breathing following exertion. Emaciation is evident with the chronic form of aspergillosis. With invasion of the fungus to the kidneys or spinal cord, ataxia or paralysis may become evident.

Susceptible species

Certain species of raptors are considered high risk species. These include Gyrfalcons (*Falco rusticolus*), Golden Eagles (*Aquila chrysaetos*), Osprey (*Pandion haliaetus*), Goshawks (*Accipiter gentilis*), Roughlegged Hawks (*Buteo lagopus*) and Red Tailed Hawks (*Buteo jamaicensis*). Many of the waterfowl such as swans, geese and pelagic species are also susceptible to an aspergillosis infection. Certain situations may increase the chance of any bird contracting aspergillosis. Following are examples of such situations (Joseph 1994).

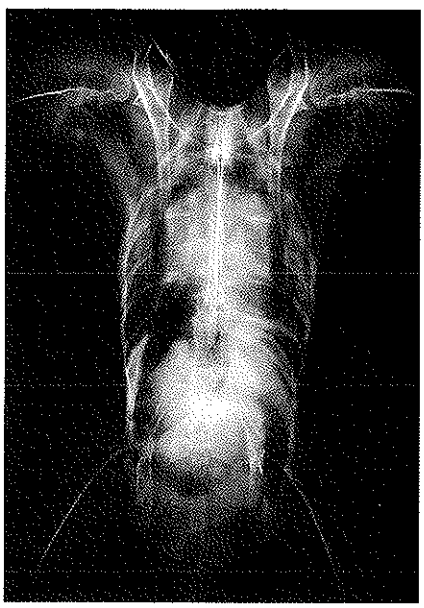


Figure 2. Aspergillosis infection involving the air sacs of a falcon.

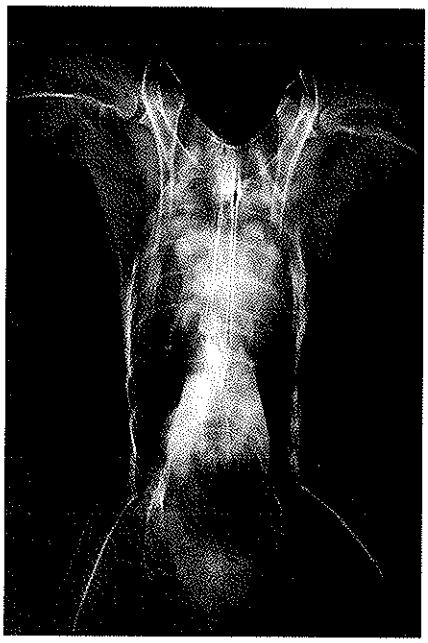


Figure 3. The same bird as in figure 2, three months following itraconazole and clotrimazole nebulization.

Passage birds or first year birds presenting to the rehabilitator are often highly stressed and diseased. In addition to their presenting complaint, many are heavily parasitized both externally and internally and may already harbor aspergillosis.

Nestlings or fledglings, whether captive bred or taken from the wild are a challenge to nurture through their first year. With an underdeveloped immune system these birds may be at risk.

Malnutrition resulting in a thiamine or protein deficiency may predispose a bird to aspergillosis.

Chambers or flight cages littered with food remains and feces increase the risk of infection. Soil floors and poor ventilation also add to the risk.

Bacterial, viral or parasitic infections that a bird is fighting off may cause the bird to be more susceptible to a secondary fungal disease.

Stress plays a major role in aspergillosis infections. Stress factors may include environmental or diet changes, capture, transportation, disease or injury. Any prolonged stress period will certainly adversely affect a bird and make them more prone to a secondary fungal infection. This scenario is often seen with oil spill birds where the loss from aspergillosis can be devastating.

Diagnostic Evaluation

Aspergillosis infections are difficult to diagnose. A series of diagnostic tests may be required to identify those birds sick with aspergillosis.

Complete blood count (CBC)—an elevated WBC is often present with an aspergillosis infection. Depending on the bird species and the stage of infection, a mild increase (resembling a stress leukogram) or marked increase (WBC> 30,000) may exist. The WBC numbers may be difficult to interpret if other disease processes are ongoing. Anemia (responsive or nonresponsive) is often present.

Chemistry panel—will evaluate the metabolic and organ functions. An elevated total protein with increased globulins is common with the subacute or chronic forms. Invasion of the kidneys, liver, spinal cord, etc. may be reflected in the enzyme changes.

Aspergillosis ELISA——is an antibody titer test run on a blood sample. An elevated titer often reflects an active infection. A negative titer may need to be repeated a week or two later if aspergillosis is still a consideration. With time the aspergillosis titer will rise. Titer levels are also used to evaluate the efficacy of treatment. This test is a tool to be used in conjunction with other laboratory tests.

Radiographs—are a necessity with any aspergillosis infection. Air sacs, lungs, trachea and internal organs can be evaluated radiographically.

Cultures of the suspicious areas (trachea, air sacs, sinuses, gastrointestinal, etc.) will aid in the confirmation of an aspergillosis infection.

Endoscopy—direct visualization of the affected areas will enhance the diagnostic and treatment capabilities.

Treatment regimes

Fungal infections are difficult to treat, expensive to treat and carry a very guarded prognosis for a complete recovery. Treatment programs initiated for the bird should be decided by the veterinarian and carried out in a hospital setting. Once the bird is stabilized and maintained with oral medications and nebulization, transfer to the rehabilitation center for long term care is possible.

The main drug used to fight an active infection of aspergillosis is Amphotericin B (Fungizone® -Squibb). This drug is fungicidal and will act the quickest to halt the spread of aspergillosis. With severe cases of aspergillosis the Amphotericin B treatment may need to be repeated. Nebulization may be carried out for several weeks or months (Redig 1987). Amphotericin B can be toxic to the kidneys thereby limiting long term use. Fluids must be given daily while the birds are receiving injections of this drug.

5-Fluorocytosine (Ancobon®-Hoffman LaRoche) has been used as an oral antifungal in conjunction with Amphotericin B. Ancobon is a fungistatic drug and may be used for extended periods of time as long as liver toxicity does not arise. Some birds will regurgitate this drug adding to the difficulty of treatment. More recently, Ancobon has been replaced by itraconazole.

The Azoles are a group of antifungal agents that have shown alot of promise towards treatment of aspergillosis (Prus1993, Flammer1994, Jones1995). Itraconazole (Sporonox®-Janssen) has shown the greatest effect as an oral medication to treat aspergillosis of the respiratory tract, while Fluconazole (Diflucan®-Roerig) is the drug of choice to treat infections of the eye or central nervous system (Como 1994, Hines 1990). Both are fungistatic so months of therapy may be required. Liver enzymes should be watched with the administration of these drugs. Itraconazole has the advantage of once daily administration. Clotrimazole (Island Pharmacy) is an antifungal agent dissolved in polyethylene glycol to a concentration of 10 mg/ml and is used in nebulization to treat respiratory aspergillosis. This drug acts by direct contact and is highly effective against aspergillosis (Joseph 1994).

To help alleviate some of the confusion on how to treat aspergillosis, here is a guideline on a treatment regime.

1. Any bird moderately ill with aspergillosis will be started on the Amphotericin B treatment regime.

> Intravenously—1.5 mg/Kg TID for 3 days
> Intratracheal—1 mg/Kg SID-TID for 3 days (dilute with saline before administration)
> Nebulization—1 mg Amphotericin B with 1 ml of saline for 15 minutes BID for 1 week.

Oral itraconazole at 10 mg/Kg SID is also started.

2. Once the bird is stable (approximately one week of treatment), clotrimazole nebulization is started. Nebulization is performed once daily for 30-45 minutes for a 3 day on, 2 day off regime.

3. Itraconazole and clotrimazole nebulization may be continued for several months, depending on the severity of the infection (Figure 3).

Legs—During flight, the legs should be in one of two positions. They should either both be tucked up so they are barely seen, or both hang straight down (this often is seen with creance flying or relatively short flights in a pen). Birds with wing disabilities will often exhibit a leg shift to the left or right (usually compensating for a weak wing on the opposite side) and birds recovering from a broken leg may fly with one leg tucked up and one dangling. The leg shift usually decreases as a bird's wing regains normal strength and function, but the dangling leg condition often remains even after a bird is in good physical shape.

Tail—The tail position during flight is another component of a bird's flight mechanics. When flying in a straight line with little wind, a raptor's tail should be positioned almost completely horizontal. Often, following recovery from a broken wing, a tail tip down on the opposite side results (along with the leg shift mentioned above). As a bird's wing strengthens, the tail position is corrected.

Feathers—It goes without saying that a raptor must be in good feather condition to fly and be evaluated appropriately. A bird with numerous broken feathers must either be imped or allowed to go through a molt before being exercised and released. Broken feathers will reduce a bird's flight ability and thus its ability to hunt successfully.

In addition, the position of feathers during flight can be an indicator of a problem in mechanics. For example, wing fractures that heal without good alignment may cause odd spacing between feathers near the fracture site, as can soft tissue and/or feather follicle damage. A more common feather problem seen is the displacement of the alula feathers following metacarpal fractures. Alula feathers are important for maneuverability. If they are displaced even slightly, their function can be reduced and they can prevent a bird from being releasable, especially species that require a high degree of maneuverability such as accipiters and falcons.

Goal 2: Appropriate strength and endurance

Raptors that are "grounded" for more than a week or two lose muscle condition fast. Because they depend on their ability to fly strongly to catch food and some must migrate long distances, it is critical that their muscles, respiratory system and circulatory systems are strong before release. To provide raptors with the exercise they need to regain lost conditioning and perform the tasks they must, they should be put on a scheduled fitness program. A bird's strength and endurance can be evaluated by the total distance it is able to fly, the height it can achieve (if exercising in a flight pen), its respiratory rate throughout an exercise session, and the power of its flight.

Flight Distance

The distance a bird is able to fly during an exercise session is a measure of its endurance. The maximum distance a bird should be encouraged to fly during a session varies between species because their body size, flight styles and hunting styles differ as does their requirement and utilization of energy. Chaplin, *et. al*, in 1988 and 1989 performed studies looking at the amount of blood lactate produced during the standardized creance flight of a variety of raptor species (Chaplin *et al.*, Chaplin 1989). The build up of lactic acid in the blood following exercise is an indicator that the working muscles are not receiving adequate oxygen. In addition, Chaplin created power output-blood lactate profiles for different species to help determine the maximum distance a species should be exercised before its muscles rely on energy produced from lactate (anaerobic metabolism) instead of available oxygen (aerobic metabolism) (Chaplin 1989). A raptor's muscle strength and endurance improve when oxygen, not lactate, is being utilized for the exercise. From these studies, it was determined that the maximum distance a small raptor should fly during any one session is about 2000 feet and the distance for medium-sized birds is about 1500 feet. Large raptors (eagles, vultures) and peregrine falcons were not tested in these studies. Thus, based on the size of the flight pen or length of the creance, you can determine the optimum number of flights. Keep in mind that these distances are maximum distances; most birds beginning an exercise program will not be strong enough to fly the entire distance and will need to work up to it with repeated exercise.

Flight Height

The height of a raptor's flight can also reflect its strength. In a flight pen, a conditioned raptor should be able to easily fly from the ground to the highest perch and maintain its height when flying from one perch to another. In limited spaces, ascending flights are very energy demanding and repetitive ascents can build muscle. One of the techniques currently used by many falconers to begin reconditioning their birds after a molt and/or keep their birds in shape between hunting trips is called "high-jumps", which basically involves training a bird to jump repetitively from the ground almost completely in a vertical to an elevated fist for small pieces of food (Fox 1995, Layman 1994).

One hundred to one hundred and fifty repetitions in a well conditioned falcon is not uncommon. The height gained during a creance flight is not as good an indicator of condition because numerous variables can affect a bird's performance. Weather conditions, species, sex of the bird, and weight of the creance can all affect a bird's ability to gain altitude with this method of exercise.

Respiratory Rate

A raptor's respiratory rate throughout a flight session can also be an indicator of its fitness. If a bird can fly the recommended distance with few or no rests and its breathing is not labored, it is in pretty good shape. However, a bird will often need periodic rests during an exercise session, especially if it is in the beginning of its program. A bird should not be pushed too hard too fast (you wouldn't run a mile with a recently healed broken leg, would you?). Also, keep in mind that panting (hawks) and gular fluttering (owls) often accompany stressful situations, such as handling and excessive heat production, and can make a quantitative assessment of a raptor's respiratory rate difficult (Chaplin *et al.*). A subjective analysis of general respiratory rate will tell you what you need to know.

Flight Power

The power of a raptor's flight can also indicate its strength and endurance. While evaluating a flight, consider the following: Is the bird capable of repeatedly taking off from the ground? Do its wing tips hit the ground during flight? Does the bird use extremely labored strokes to maintain lift? How far can the bird fly while maintaining good flight mechanics? If a raptor cannot take off from the ground, maintain height at least so its wing tips don't touch the ground, fly gracefully without labored strokes, and fly the recommended distance while maintaining good mechanics, it needs more conditioning before it can be released.

Goal 3: Recovering appropriate flight style

Different species of raptors possess different flying and hunting styles (Fox 1995, Kerlinger 1989). Therefore, it is important that a bird have adequate strength and maneuverability to fly in its characteristic style. For example, accipiters are sprinters which often maneuver through trees to capture prey, and also fly with a flap-flap-glide style in more open spaces. They depend on this maneuverability to survive. Peregrine falcons are built for speed and power. They must be able to sustain powered flapping flight for extended periods, maneuver quickly, and dive. Owls tend to have a perch, glide, pounce strategy and therefore do not require the maneuverability of accipiters or the power of peregrine falcons. If injuries prevent a raptor from flying in its characteristic style, it is not releasable. If you are not sure how a certain species should fly, review natural history information or ask other rehabilitators or local falconers for assistance.

Exercise Techniques

There are three major exercise techniques currently used to accomplish the goals of reconditioning mentioned above. Flight pen exercise, creance flying, and free flying using falconry techniques are all used successfully and vary in their effectiveness with different species.

Flight Pen Exercise

Exercising a raptor in a flight pen can be done by one person, does not involve direct handling of a bird, and can be a quicker procedure than other methods (Engelmann and Marcum 1993). However, the flight pen must be sufficiently long and high to allow a bird to adequately condition its muscles. The Raptor Center recommends minimum lengths of 30 feet for small birds (American kestrels, Eastern screech owls), 80 feet for medium-sized birds (great Horned Owls, red-tailed hawks), and 150 feet for large birds (eagles, osprey, vultures), and minimum pen heights of 10-12' for all species. In addition, exercising in a flight pen can limit the number of birds that can be housed together. Disturbing patients not yet ready for intense active exercise, and creating chaos in a group of birds housed together can result in injury to the birds, especially with a flight pen limited in size. If you decide to exercise your birds in a flight pen, design the pen for that purpose. Contact other rehabilitators for suggestions on cage size, shape, and other design considerations.

Creance Flying

Creance flying involves removing a bird from its housing structure, attaching equipment to keep it under control (leather straps on its legs and a 150-400' line), and encouraging it to fly in a large open area (fig. 1). This process requires two people, takes a little more time than exercising in a flight pen and can be limited by weather conditions. However, creance flying does provide a good workout and allows you to clearly evaluate a bird's flight without a roof, walls, or other birds interfering. It is important to evaluate a bird's ability to fly in normal wind currents. For example, a raptor

with a healed metacarpal fracture may seem to fly fine in a pen but when out in the wind be unable to turn and maneuver appropriately.

Creance flying allows numerous birds to be housed together in a pen and only briefly disturbed as the bird to be exercised is removed. Also, flight pens large enough for adequate exercise can be costly and not an option for many rehabilitators.

Falconry Techniques

Falconry, the sport of hunting with trained hawks, has contributed greatly to the field of raptor rehabilitation. The equipment put on raptors (jesses, leashes, hoods), the method of creance flying, the process of hacking, and reentering birds on game were all developed by falconers and modified for rehabilitation. In addition, many falconers themselves have a wealth of knowledge on bird behavior, hunting and flying styles, and reconditioning techniques that they are willing to share.

Falconry involves developing a relationship with a bird, one that is based on food. Birds trained for the sport are often exposed to as many situations and activities as possible so they become calm and unafraid of normal occurrences. However, the techniques used by falconers for conditioning and providing live game for a raptor do not require that a bird be "tamed." The extent of training only needs to be to the point where a falconer can get a bird back once it flies free. This is the type of training that is extremely useful for birds in rehabilitation.

Not every raptor needs what falconry training offers. However, young birds recovering from starvation and peregrine falcons are two groups of birds that benefit greatly from extensive conditioning and exposure to prey in natural settings. From personal experience, these birds develop a strength and survival attitude that cannot be matched from other conditioning techniques.

Several techniques are used by falconers to recondition birds: lure flying (training a bird to fly to a leather object containing a piece of meat) (Beebe and Webster 1989, Fox 1995), high-jumps (repeatedly encouraging a bird to jump from the ground up to a glove garnished with a small piece of food) (Fox 1995, Layman 1994), flying to the glove (encouraging a bird to fly increasing distances to the fist for a piece of food), and flying to a kite (training a bird to fly increased heights to "catch" a piece of food attached to the kite line) (Scarbrough 1995). An average time of 3-4 weeks of training, followed by about 4 weeks of regularly scheduled exercise is usually required. Thus in two months a bird can be physically and mentally conditioned using falconry techniques, about the same amount of time often needed for other conditioning methods. Only experienced falconers should undertake the reconditioning of these birds.

All three methods of reconditioning can be effective, and the exercise program designed should be tailored to the needs of each individual bird. Therefore, rehabilitators should feel free to combine techniques. Some people exercise birds in a flight pen and then conduct a pre-release session of creance flying to assess the effectiveness of their flight pen exercise program. Since flight pens are built with different dimensions (often based on available resources), this is an excellent idea to ensure that the birds are getting adequate conditioning.

Mental Reconditioning

When a raptor is in captivity for several months, it should be reintroduced to live prey before being released. This is especially true of young birds that were starvation cases or injured before they gained enough hunting experience to survive. Young hawks tend to be easily trainable, and falconers can assist with their reconditioning by exposing them to natural prey in natural settings and providing them with opportunities to refine their hunting skills (through free flying and hacking). Orphaned hawks should be put through a hack and once released exposed to live prey. Young owls, on the other hand, tend to be less trainable and develop their hunting skills more slowly. Therefore, a rather lengthy period of live training in a flight pen before release is recommended (McKeever 1987). For older birds that hunt rodents, the process of live training involves providing them with mice or small rats in a "mouse proof" flight pen. Many birds need several days of fasting before they will pay attention to the live



Figure 1. An adult Red-tailed Hawk is exercised using the creance flying technique.

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The ABC's of Housing Raptors

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Abstract

Housing for raptors can be a complicated matter once all factors are considered. This paper offers suggestions for construction, building materials and design ideas. The designs put emphasis on function and safety while keeping individual species requirements in mind. Simple tips are provided that may improve release statistics and shorten time in captivity for raptors in rehabilitation. Space limitations, climate, and predator control are discussed. Enclosures for permanent birds, and habitat displays for public viewing are also addressed.

Key Words: Mews, wildlife rehabilitation, wildlife housing, raptors, hawks, falconry

Introduction

Mew *n* [ME mewe, fr. MF mue] The building in which hawks are kept at night and in inclement weather. Traditionally the building in which they were molted.

Weathering *vb* ['weth-(e-)rin] Ground — Area exposed to the open air. Traditionally the area where hawks can be exposed to the elements (Beebe and Webster, 1989).

Modern enclosures for raptors are generally a combination of the two traditional forms of separate mews and weathering ground. Structures must provide safety, health and comfort for the raptors in your care. When designing enclosures, it is important to pay close attention to the natural history of the species you are housing (Parry-Jones 1993). Eagles and the soaring buteos, for example, require long flight areas. They tend to do more gliding than flapping if they are housed in short mews. Harriers and falcons require height to perfect their hovering hunting technique. Height is also important for the short runway, stop and go flyers, like woodland buteos. Social species, such as falcons and some buteos, benefit by having windows to observe the outside world, thereby stimulating their interest and keeping their senses active. Some of the more sensitive species, such as accipiters, require enclosed mews to shut out the world, focusing their attention on recovery instead of the frustration of being in captivity.

Other factors will affect your caging. Space limitations, water access, drainage, noisy traffic, or uncontrollable human exposure may be a consideration. Extreme weather conditions or other unusual situations peculiar to your area may influence the style of enclosure you choose. Whatever your situation, evaluate all possibilities carefully before construction begins. Building is expensive, but making changes and corrections is even more so.

Appropriate sizes for flight areas are the subject of the IWRC/NWRA wildlife rehabilitation standards program, and are too lengthy to reproduce here. They are easily obtainable through IWRC and NWRA. I encourage everyone to obtain a copy of the standards prior to planning new structures.

Building Materials

Because of the powerful distance vision of raptors (which have been housed in captivity for thousands of years), these birds do best in cages with vertical barring. The rehabilitator must avoid wire or lattice that they can climb like a ladder, with resulting feet and feather damage.

Vertical barring is difficult for them to grip, discouraging them from hanging on it. When spaced correctly, vertical bars prevent the cere and even eye damage that can result from birds trying to go through wire.

The building material of choice for raptor housing in most cases is wood (Beebe and Webster 1989). However, other materials can be used to good advantage under special circumstances. Corrugated plastic panels can be used for a semi-solid wall option (McKeever 1987). An advantage of corrugated plastic is light can filter through and it is easy to clean; however, it allows no ventilation, and retains heat. I have had mixed success with the black plastic bird wire available in home improvement stores. It is appropriate to use for roofs, and housing education birds that are comfortable with confinement. It should not however, be used for rehabilitation flight mews. While the plastic wire is easy to install, it is expensive and needs to be replaced every few years, as it gets brittle, breaking with little or no warning. The plastic wire also causes feather damage in active birds. Heavy duty shade cloth is a good choice for roofing material in hot and sunny climates. It works best if framed in section's first, so it appears firm. This will discourage raptors, particularly owls, from hanging from it. Corrugated plastic panels are also a good roofing option (Parry-Jones 1993). It should be noted however, that plastic panels should be checked often particularly in extreme temperatures to check for cracking or weakness.

Vertical bars can be made of several different materials: If using lumber, 2"x 3" boards can be used for eagles and 1"x 2"s for most other species. Wooden dowels or PVC pipe also work well. PVC has the added advantage of being inexpensive, easy to clean and doesn't warp. (Holderman 1992, CA Hawking Club 1990). However, it may bend and allow accidental escape. Metal conduit may be a good option if you don't live in a cold climate. In freezing conditions a hawk may be injured by freezing its eye or wet feet to the metal (Kimsey/Hodge 1992). Chicken wire should never be used in any aspect of raptor housing (Arent and Martell 1996, Durham and Walters 1994). Chicken wire damages eyes, ceres, feathers, feet and talons when raptors fly against or hang from it. The wire can break or crimp in the talons of the powerful birds and cause deep cuts to the feet or other body parts that happen to protrude from the wire. This damage can occur quickly. I have seen a great-horned owl lose two talon sheaths and break several tail feathers within a half hour of being put into a chicken wire enclosure. *Chicken wire should never be used to house raptors.*

Perches should be covered with long-artificial turf, plush pile carpet or sisal rope. The artificial turf is the type usually purchased as door mats at home-improvement stores. Inexpensive continuous loop carpet should be avoided as it can catch around a birds' talons, or be torn and ingested. If you are housing heavy bodied raptors, such as eagles or large buteos, you may want to pad the perch first with egg crate type mattress pad foam, or double layers of pipe insulator. We at Raptor Education Group discovered this application while researching methods for keeping our birds more comfortable in extreme cold temperatures. We found it provides not only additional padding to the feet while perching, but acts as a shock absorber during landing, and creates a warmer perch for cold weather climates. The perches with egg crate foam as an under pad will also dry out quickly when wet. Take care to cover all the foam with perch covering discussed above, making it non-visible to the birds or they may tear at it and/or ingest it. Duct tape is a good method by which to attach the turf or carpet to the perch. Duct taping eliminates the need for nails or staples that can work their way out to become a hazard to the bird's feet, or become accidentally ingested.

Your flights should have a variety of perch sizes, styles and coverings. Individual birds will choose the perch they prefer by position, style, covering texture, and availability. Offering several choices in each enclosure will decrease the frequency of foot problems not only for physical reasons, but psychological as well. Once again the natural habitat of the birds in your flights will dictate the types of perches offered. For instance, fence post style perches should be installed in enclosures that house grassland birds, such as Short-eared Owls, Harriers, Swainson's Hawks, or Burrowing Owls. These birds utilize low perches in nature. Falcons prefer shelf perches, which mimic the ledges they normally nest upon. Their wide feet and long toes can rest comfortably with equal weight distribution, important to relieve pressure points and prevent foot problems. Wide-winged buteos and kites that select open branches on tree tops in the wild, should have the same opportunity in your facility.

Construction

Mews are "inside out" buildings, compared to normal frame construction. The frame should be outside with the slats and flat boards on the inside so there are no ledges on the inside of your housing. Horizontal two by fours will be attractive perching places to raptors with possible resulting feather or foot problems. If you already have housing that has inside ledges, it is best to

treat them like a perch for foot protection, covering them with appropriate material discussed above.

Rectangular flights work best if you have that option (Figures A,B,C and D). One end of the flight should be solid, with a few solid panels down either side to give the birds security. (Arent and Martell 1996) Four to six feet of solid panel should run along the bottom of the wall before vertical strips begin (figures E and F). This solid section serves multiple functions. It creates a protective visual barrier from small children and animals, both wild and domestic. The solid barrier will allow the flights to be used for live prey release and hunt training. Spacing between the parallel bars will vary from 1 1/2 inch for eagles to 1/2 inch for kestrels and pygmy or saw-whet owls. Cavity nesting birds are masters at diving into and getting out of small spaces. Be sure to keep this in mind when planning your bar spacing. Entry doors should be on the side of the flight away from the solid security wall. This allows the birds to see you and anticipate the disturbance before you enter.

Longer irregular flights can be created by using a block complex design. This combines common walls with detachable or movable partitions that can be removed or lowered when long flights are required. This design maximizes facility space while developing the bird's flight muscles and strength for turns and controlled stops. Examples are given (figures C and D); however, you may use your own configuration, form fit to your specific lot size, shape and species rehabilitated. Irregular flights are not suitable for eagles.

In assembling the housing, consider using screws instead of nails. Screws hold pieces together securely, and aren't as likely as nails to protrude through wood panels or lumber. Check for any sharp points or edges visible to the inside and remove or file them flush to the surface before raptors are housed in the facility. Sharp points are a hazard to raptors.

Double door or safety entries are important, particularly in flight mews that house many raptors at once (see figures). This safety area will not only prevent birds from escaping prematurely, but can serve as storage for essential equipment, freezer space, or water access.

Flights of 40 feet or more will require shock absorbers to be put on one side of the perches. This is an important feature and will help prevent injury to the sensitive foot pad (Gibson unpub.). Repetitive foot injury leads to bumblefoot (Redig 1984). A good choice for a shock absorber is a section of a tire, or for smaller perches, rubber tie-down straps. The rubber should be attached to the wall and the perch itself, creating a bridge between the two. Your goal is to have some "give" to the perch when the bird lands. The perch should not however, be unstable or shaky when the birds are standing on it. Altering one side of the perch is generally enough to create this effect; wide flights, however, may require double-sided attachment. Swinging perches have also been used with success in mews with a small number of birds, but injuries have been reported when several birds take flight at once and find the swing in motion.

Your housing should include an area that will allow the birds privacy. Falconers refer to this area as a "jump box" or "shelter box" (Beebe and Webster 1989). With several raptors in a flight, the shelter area may need to be as large as a closet or small room when housing large birds. McKeever suggests that owl species require as many individual shelter boxes as there are residents to prevent aggression (McKeever 1987). Raptors are predators and thus need to be able to observe, without being observed, if they choose. This sense of privacy comforts, and will go far in reducing captive stress.

Roof

Roof designs and materials will vary greatly with climate. Keep in mind that part of the roof should be covered to give adequate shade and shelter. A section of the roof should also remain open to the elements, allowing the birds to sun themselves, take advantage of rain for bathing, and in some cases experience snow if it is natural for them. Check the condition of your roofs frequently. A small tear, or damage from, perhaps, a fallen limb, can be a source of "premature release" to your birds (Durman-Walters 1994).

Flooring

The best covering for floors of mews or flights is small round pea gravel. Sharp gravel can cause injury to raptors' feet. Natural turf, or grass is also a good choice. Hay, sawdust or wood chips are not recommended, as they are difficult to clean, contaminate food items, and when wet can be a source of aspergillosis (Joseph 1994). Sand floors can cause digestive problems if food items are dropped into it before eating (Ford 1992). The pea gravel should be at least 3-4 inches deep (for drainage) and up to 12" is preferred to help prevent impact injuries on the feet. This depth will allow the pea gravel to act as a cushion when the birds land, moving to surround the foot rather than simply braking it. Pea gravel is easily cleaned and, unlike dirt, will not turn to mud or sully

drinking/bathing water after the bird walks on it. The pea gravel should be changed annually, or more often if necessary.

Ventilation

Ventilation is an issue that is frequently overlooked. Birds require proper ventilation to remain healthy (Jannika and Redig 1989). If you have a multiple flight complex, with solid common walls and whole roofing, you will need additional air exchange. Some air movement can be created by using screen-covered openings at floor level. If this option does not produce enough exchange, you may have to invest in roof or side wall ventilators available from farm equipment suppliers. Ventilators come in a wide variety of styles and sizes. Some are electric, others wind operated. Choose the quietest unit possible so as not to stress your raptors. Owls will be particularly sensitive and distracted by sound.

Climate Needs

Climate affects housing and building requirements. Additional cover must be used in extreme climates, either hot or cold. Heavy snow load may require roof strengthening. High wind or tornado belt wildlife centers may want to sink part of the mews below ground, particularly tall mews, so as not to be easily toppled in strong winds. If possible, investigate raptor centers in your climate zone and make inquiries of building contractors to get a sense of problems that may affect structure durability. The enclosures you provide will be home to the raptors in your care while they recover from injuries. They need to be safe from predators, sheltered from inclement weather, and as comfortable and stress free as captivity allows them.

Education Birds or Housing Permanent Captives

When planning housing for permanent birds, consider that raptors are long lived and will be part of your organization for many years. Educational birds work hard for our wildlife centers. They are our partners in educational programs, many times act as foster parents to orphans of their species, and even serve as blood donors for birds in need. They deserve a safe and comfortable place. As always, your focus with housing will be safety, health and comfort, both physical and psychological.

Your structure and building materials will be the same as those mews that house rehabilitation birds. The differences will be the size, perch location, and those touches that create a more natural habitat for the birds. If this housing is used as a display for the public, you may want to include extras such as native shrubs, ponds or even a large tree trunk with a visible Plexiglas™ cut-away cavity for an enclosure that houses a cavity nester (Figure G). These special touches can serve as natural history/ habitat education for the public. A pond could be used for drinking and bathing, or even stocked with trout for fish-eating raptors (Gibson 1994). These accouterments can only make your bird more comfortable. Your creativity, available space and funding will be your only limits.

Many decisions for educational birds will depend on their disability. For example, let us consider a bird with a fully amputated wing. Ponds pose a danger to an amputee. If it falls in the water and cannot right itself, it will drown. High perches also create dangerous situations for amputees. While it is true high perches can be made accessible with ladder type inclines, the bird must get down from that height, too. Many birds learn to "ladder" down from the perch, but if frightened they may jump, sustaining additional injury upon landing. Birds naturally choose the highest perch they can attain within their confined area. The enclosure height for amputees is a judgment call for the caretaker. If the bird can jump down 4 ft. without injury, make the enclosure ceiling no higher than 7-8 ft. A 20 ft ceiling for a bird that is restricted to a 4 ft. height will produce a stressful situation for the bird as it continually looks skyward to reach greater heights. Birds with total amputations of one wing will also need a heat source in cold weather, as they have lost much of their natural insulating qualities. I might add that the example of a total amputee is done merely to show one of the most difficult physical problems to be encountered for management, not to suggest their use in education (Arent and Martell 1996).

Educational housing, after a few basics, is a matter of common sense and judgment. Nothing can replace observation of the birds in your care. Anticipate possible problems and correct them. If the bird develops a problem with any aspect of the housing, change it (Parry-Jones 1993). Offer alternatives until you have housing that fits the individual. Providing safety, promoting good health and making your bird comfortable both physically and psychologically is your goal. For permanent birds, this is their final stop in life. It is your responsibility to make it a quality one. There are currently no national standards for permanent housing in rehabilitation; choices, as much space as possible within budgets and viewing should be your criteria.

Predator Control

Predator control must be incorporated in your building from the blue-print stage. Predators are a normal and natural part of our world. Attempting to enter enclosures with the scent of fresh meat within is as natural for them as it is for a fish to live in water. It's amazing that people seem surprised when it occurs. Without an underground barrier or cement floor, you run the risk of endangering the raptors in your care. A more recent problem is hantavirus, which is transmitted through rodent urine and droppings. While not yet felt throughout all of the United States, it alerts us to the danger of wild rodents entering enclosures (U.S. Dept. Of Human Services 1993) (Beishe 1991).

Most predators will be stopped by a 2-3 foot deep perimeter foundation under the walls of your mews. Several materials create adequate guards. Half inch hardware mesh, buried around the building perimeter, works well and is the least expensive. It can also be installed around an existing building without compromising the integrity of the structure. Most burrowing predators start tunneling close to the building and will get discouraged if they find wire blocking their entry. A more secure guard would be cement blocks acting as foundation, or a poured cement perimeter foundation. Again both should be 2-3 feet deep. A solid cement floor or galvanized wire under the entire cage will predator proof as well, but both need to be covered with several inches of substrate, so as not to come into contact with the raptors' feet. Add drains in solid cement floors or slope to facilitate cleaning.

In some areas of our county, experts are suggesting the use of chain link fencing or electric fencing around wildlife center complexes. This is to discourage larger predators, such as bear, mountain lions and wolves, or human intrusion (Oregon Dept of Wildlife 1996). Check with your regional USF&WS office and state natural resources agency for regulations that may affect your center.

Human Intrusion

People are attracted to wildlife for many reasons. Some folks are just curious or excited, forgetting momentarily that these creatures are wild, not the pets they are accustomed to. Some unsavory individuals may desire entry with less honorable intentions. Prevention is the key to most human intrusion problems. A solid perimeter fence provides a visual barrier and will eliminate the curious. A small fence or barrier gives a gentle reminder to most people that they need to stay behind it. Signs which emphasize your function as a wild bird rehabilitation center are helpful to remind the enthusiastic that the birds are not pets. It is mandatory to have a clear policy with your volunteers as to restricting visitors (even their very best friends). It is equally important to explain the reasoning behind such a policy. Locks on your cage doors are a must.

Conclusion

Raptor housing is more than just a temporary place for confinement of birds. It is a valuable tool in rehabilitation efforts. Incorporating natural history information can make the difference between releasing a sane, healthy, well flighted bird or losing it altogether. Well-designed enclosures prevent accidents and additional injury, and lessen stress. Sometimes simple inexpensive additions suffice. Other measures may be more expensive, but necessary. The ultimate goal never changes: to get your recovering patients on the wing and on their own at the earliest possible date and in the best possible condition.

Acknowledgements

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Items Mentioned in the Text: The Standards Booklet is available for $6.50 at IWRC, 4437 Central Place, Suisun, CA. 94533.

Reconditioning Raptors Continued from Page 22

prey and catch it. Others don't seem to get the concept at all and sometimes need to be housed with a bird that will take live prey to show them what to do. Live training is a bit more complicated with bird-eating birds such as the larger falcons and accipiters. They need to catch birds in the air. This can be done in a large enough pen for accipiters but does not work as well for large falcons. Once again, falconers can be of great assistance by working with the larger falcons until they are strong and agile enough to catch food in the air. The kite method mentioned previously works extremely well (and quickly) for peregrine falcons (Scarbrough 1995).

Conclusion

Raptor Rehabilitation is a very complex process that extends far beyond treating the medical problem with which birds are presented. Physical and "mental" reconditioning are necessary prior to release, and the methods used must be tailored to the individual needs of the patient. Several options are available and each may be called upon at different times. The challenge is to determine which one or combination will be the most effective for the individual and provide it with the strength and mind-set to be a survivor.

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Author Profile

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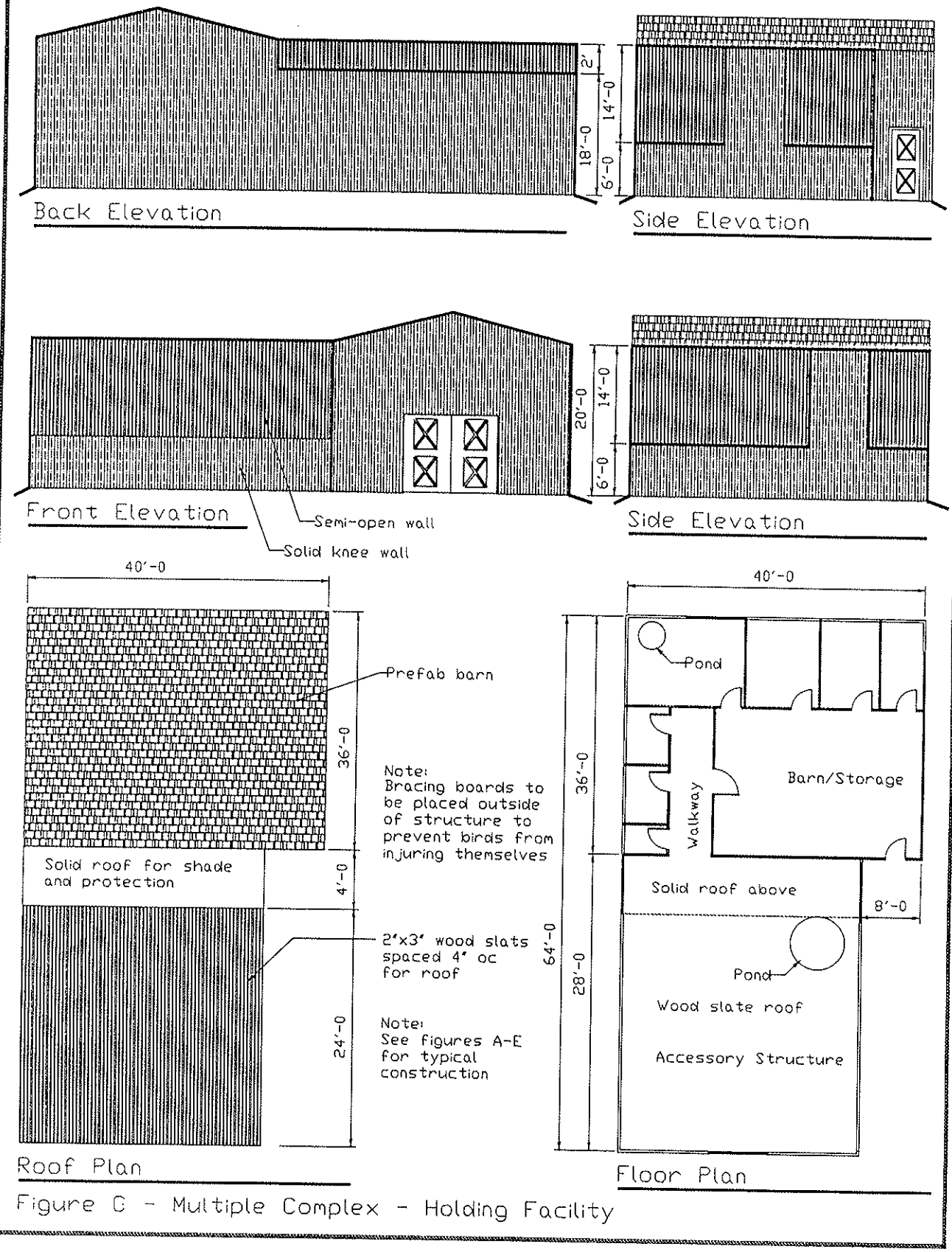


Figure C - Multiple Complex - Holding Facility

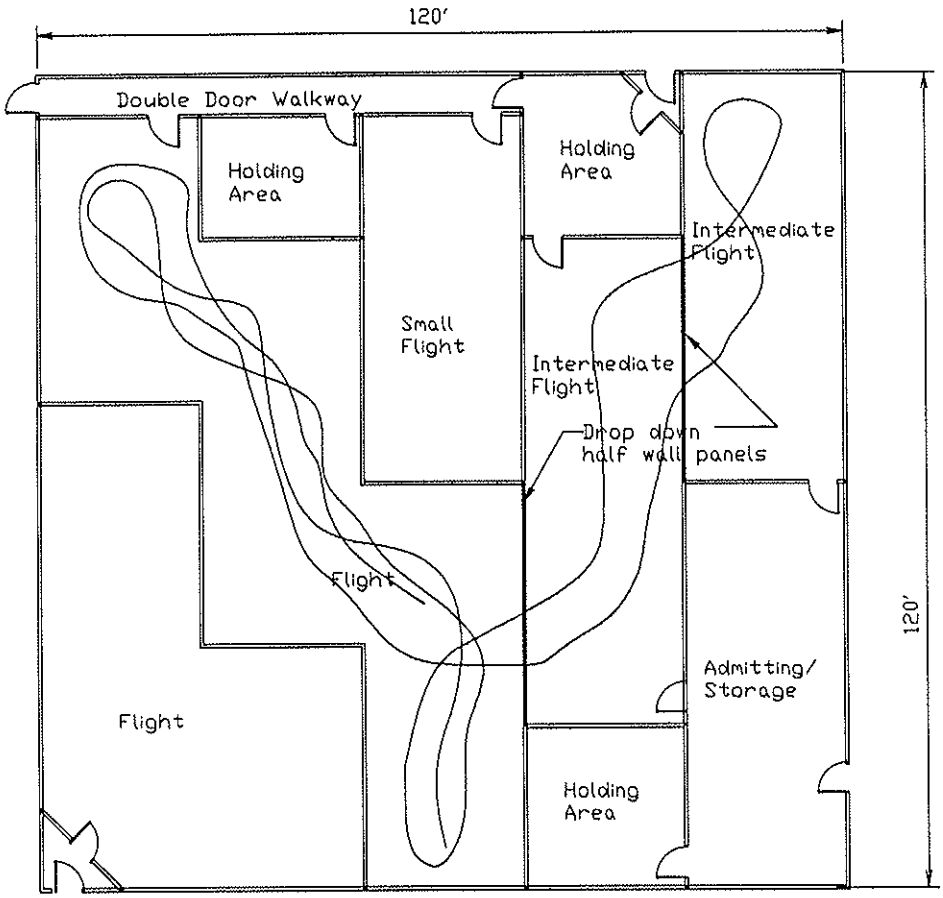
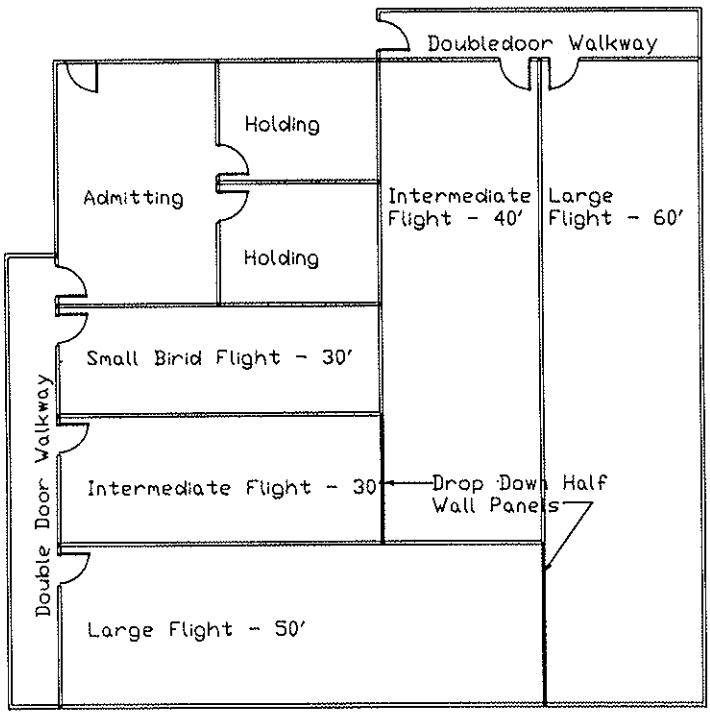


Figure - C



The drop down half wall panels create larger flight areas when needed.

Flight Complexes

Figure - D

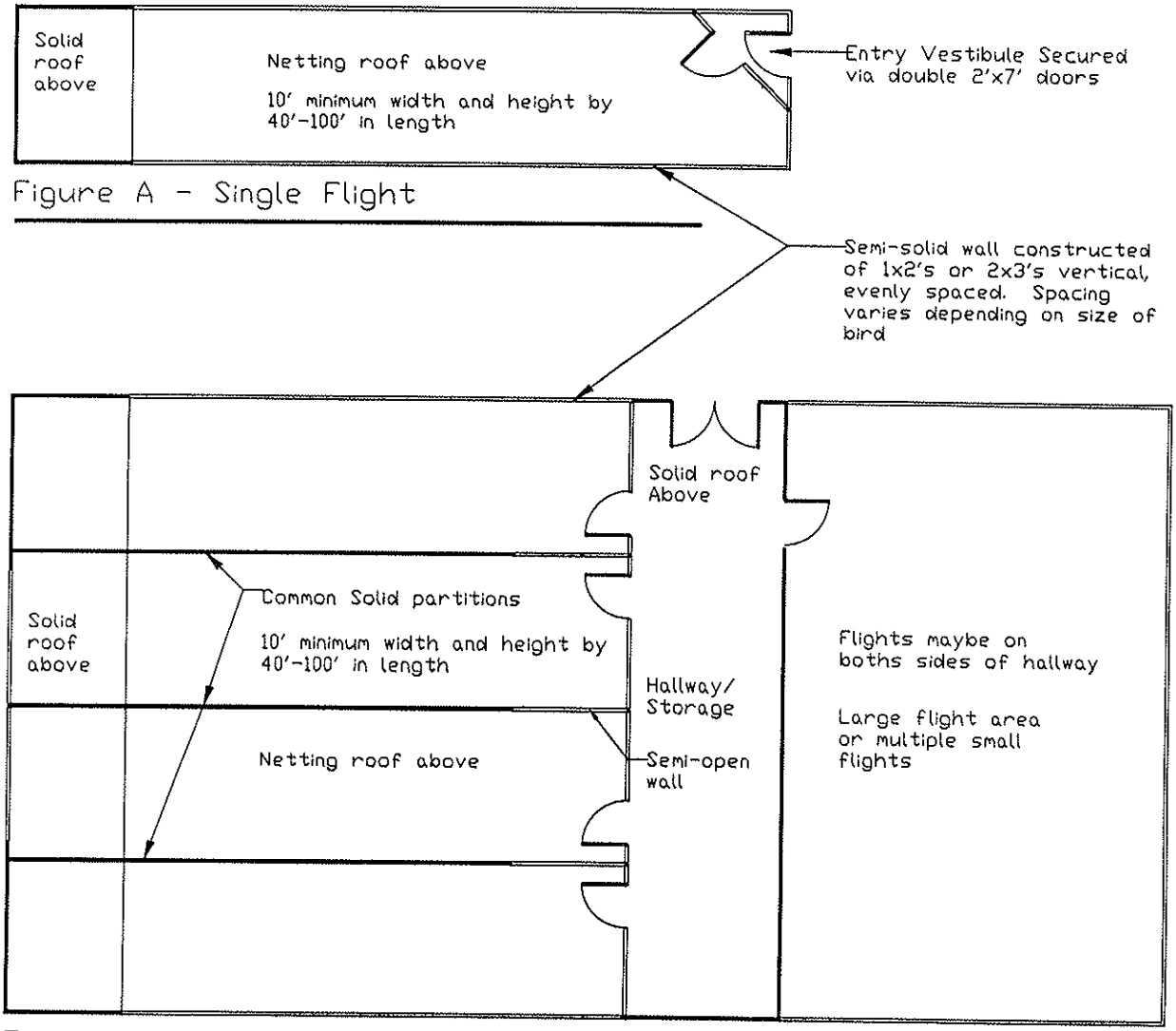


Figure A - Single Flight

Figure B - Multi-Flight Complex

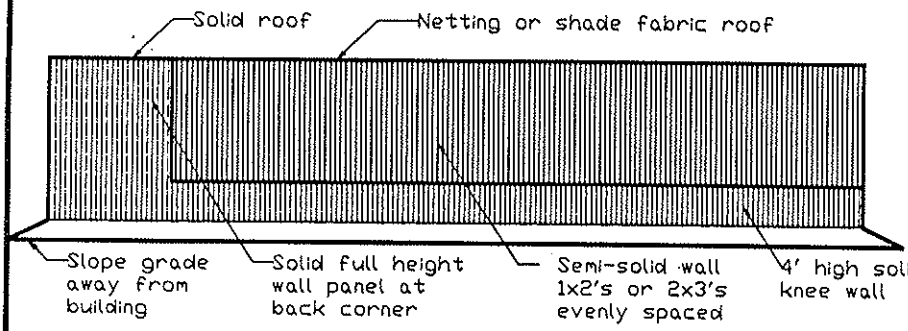


Figure F - Typical Side Elevation

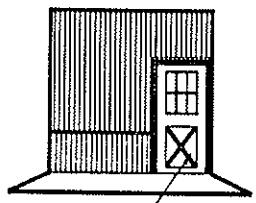


Figure E - Front

Wildlife Web Page Input is Requested:

If you have been watching TV, you probably have seen the world wide web (www) addresses being advertised in many of the tv commercials (such as "http://www.misccompany.com"). The www allows people from all over the world to have access to information on every computer connected to it through web pages and sites.

Recently, a site has been started that is devoted to information on wildlife rehabilitation. This site is called The Wildlife Rehabilitation Information Directory. The address for this site is: "http://www.cc.ndsu.nodak.edu/~devold/twrid/html/hp.htm".

We need your help to contribute to this site. There is a section on it, whereby people who are looking for someone to call regarding an injured/orphaned wild animal can look up listings of contact people by state and country. These contact people then can refer the caller to an appropriate wildlife rehabilitator in their area.

By having an extensive listing of rehabilitation contacts, people throughout the country and even the world will have quick help for theinjured animals that they find. We have already had a few wonderful"cross-country" and even "multi-national" success stories of peoplegetting connected to nearby wildlife rehabilitators.

If you are willing to be listed in this directory, please send to the address below the following information:

Your state, city, and country (if non-US), your name, phone number, and the organization you are associated with (or just licensed rehabilitator if none).

Send the information to Ronda DeVold by either:
email: devold@badlands.nodak.edu
or by regular mail:

Ronda DeVold
North Dakota State University
Van Es Laboratories, room 159
Fargo, ND 58105

If you have any questions, please call Ronda at: 701-231-7519.

Thank you so much for your assistance in this effort!

Continued from page 2

Quarterly. What are examples of publications that are not peer-reviewed? Last year many of you received a copy of *Beaks, Brains & Bones: Experiences in Wild Bird Trauma* (by Kit Chubb, Ontario, Canada) that was not peer-reviewed. Most rehabilitation organization newsletters are not peer-reviewed. Additionally, the internet offers anyone a chance to say whatever they think—but it should be regarded as just that, what someone thinks.

Should we not give credence to these publications because they have not been peer-reviewed? No, but we must weigh our confidence more heavily with those publications that have really looked deeply at the information presented. What you read in a newsletter may develop into a peer-reviewed article a year or two later as more data is accumulated and what is being presented can be scientifically validated or may evolve into something quite different. Remember, just because something is typeset, printed and distributed does not mean that anyone has seriously looked at whether the information is valid. With desktop publishing, anyone with the right equipment can produce a nice looking booklet, rehabilitation manual, or occasional paper. What you need to look for is the process by which it came to be published. Read and think critically. The lives you save may be your patients.

-jan white-

Attention Bat Rehabilitators:

As more pesticide issues come up for this species, we are interesting in setting up a dialogue with rehabilitators who may be seeing problems. If you rehabilitate wild bats, please drop the editor a line at 944 Anderson Dr. Suisun, CA and I will put you on my correspondence file for this species.

Call for Article Submissions for Special Topics Issue (Vol. 20, #3, Fall of 1997) on Waterfowl and Pelagic Birds

The fall issue next year will focus on rehabilitation topics related to waterfowl and pelagic species of birds. Our special topics issues have articles relating to a wide variety of rehabilitation topics (e.g., natural history, conservation, housing, veterinary care, education, rehabilitation methods, post-release papers etc.). If you have an interest in writing a paper for this issue, please drop the editor a note at 944 Anderson Dr. Suisun, CA. Papers are due by May 15th, 1997. I can be reached by phone/fax at (707) 422-1035.

Join IWRC where it we began for our 20th Annual Wildlife Rehabilitation Conference

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Mark your calendars now! *IWRC, will be celebrating 25 years of service to our membership at our 20th annual conference hosted by The Lindsay Museum of Walnut Creek, California. We are coming home to the place where we first grew to serve wildlife rehabilitators outside of the San Francisco Bay area and will be working with a conference committee from the organization which helped found IWRC in 1972.*

As is our custom, we will schedule our Basic Wildlife Rehabilitation 1AB Skills Seminar℠ and three advanced skills seminars just prior to the opening of the conference. This allows members from remote areas that do not have a large enough pool of potential class registrants to come to several courses and then attend the conference thus saving on travel costs.

The conference will offer a variety of venues for continuing education ranging from general session papers of a wide variety of interesting topics to workshops.

We plan to mail a "Call for Papers" this fall and hope to put the program together by next May. Registration materials, with program specifics will be mailed to all members in July of 1997.

By the way, if you would like to host an upcoming conference in your areas, contact the IWRC office for details (707) 864-1761.

Late Entry for IWRC Jobline:

The Central Wesconsin Wildlife Center, Inc. (CWWC) is searching for an ***Executive Director*** *to be the full-time manager of it's new hospital and education resource center. The center serves an eight county area and has 3 paid positions. There are more than 50 volunteers and several university interns to assist the staff handle a caseload of 700-900 animals per year. CWWC is starting to build a $350,000 state-of-the-art facility to be completed next year. CWWC is governed by a 15 member Board of Directors and several working committees. The Executive Director will work with the Operations Committee.*

Requirements: *A suscessful candidate for this position will need to have a B.S. degree in Biology, Wildlife Management Animal Science and have experience in wildlife rehabilitation. Management experience is desired. Ability to plan, schedule, delegate and supervise a staff, to communicate clearly and freely, to inspire, to manage resources, and to keep abreast of current information in the wildlife rehabilitation field are also prerequisites for this position.*

Salary: *Starting salary range is $22,000 to $27,000 depending on credentials and opportunity for advancement is possible. Deadline for applications is November 15 or until the position is filled. Applications should contact Al Koerten, Chair, CCWC Operations Committee at 2501 Church Street, Stevens Point, WI for specific application information.*

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Wildlife rehabilitation can be challenging or frustrating depending upon how quickly you find answers to those first-time problems. When you are faced with a new situation or looking for a new solution to a recurring problem, avoid that feeling of hopelessness or isolation and give us a call. The Wildlife Hotline is designed to help *wildlife rehabilitators* by connecting you to an expert who can give you the answers you need. IWRC office personnel staff the hotline and make referrals to any number of specialists in our diverse field. So if you are stumped or just want a second opinion from someone who's been there—give us a call and we will try to help. Calls should be placed during business hours (PST) for the fastest response, however, the answering machine will take messages at any hour of the day.

To use the hotline: call (707) 864-1762; leave a message detailing your question, problem or what specific information you need. The Wildlife Hotline will return your call COLLECT with the help you need.

Photo by Tom Millham