Summary of Veterinary Care Guidelines for

Otters in Zoos, Aquariums, Rehabilitation, and Wildlife Centers

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Photograph: N. A. river otter hind foot, J. Reed-Smith, John Ball Zoological Garden

VETERINARY CARE

Veterinary services are a vital component of excellent animal care practices. A full-time staff veterinarian is recommended for all types of wildlife facilities; however, in cases where this is not practical, a consulting/part-time veterinarian should be under contract. In these cases the contracted veterinarian should make at least twice monthly inspections of the animal collection and be available for any emergencies. Veterinary coverage also should be available so that any indications of disease, injury, or stress may be responded to in a timely manner.

Protocols for the use and security of drugs used for veterinary purposes should be formally written and available to animal care staff. Procedures should include, but are not limited to: a list of persons authorized to administer animal drugs, situations in which they are to be utilized, location of animal drugs and those persons with access to them, and emergency procedures in the event of accidental human exposure to these drugs.

Animal record keeping is an important element of animal care and ensures that information about individual otters and their treatment is always available. A designated staff member should be responsible for maintaining an animal record keeping system and for conveying relevant laws and regulations to the animal care staff. Record keeping should be accurate and documented on a daily basis. Records should be kept for each individual animal and include all information regarding their daily care, diet, behavior, enrichment program, enclosure they are kept in, other animals in their enclosure, staff responsible for their care, reproductive history, general health, chronic health conditions, and veterinary procedures performed. An end point for all listed medical problems should be entered and dated in the medical record.

INDIVIDUAL IDENTIFICATION

All otters should be identified as soon as possible after birth or arrival at an institution in some fashion. If possible transponder chips placed subcutaneously over the bridge of the nose/forehead area, subcutaneously in the intrascapular area at the base of the ears, or subcutaneously between the scapulas (recommended placement for giant otter) is the preferred method of identification. Placement in all of these areas has been met with success and failure (migration, loss, unable to read them as planned). Placement location of the transponder chip, chip number, and type of chip should be recorded in the animal's medical record. If the use of transponder chips is not possible, other methods of identifying individuals should be considered. These may include tattoos placed on the inner thigh of a hind leg or photographs of face, nose, and/or individually, identifying patterns.

QUARANTINE

Wildlife institutions should have holding facilities or procedures for the quarantine of newly arrived animals and isolation facilities or procedures for the treatment of sick/injured animals. All quarantine procedures should be supervised by a veterinarian if possible, or an individual with veterinary training, Quarantine procedures should be formally written and available to staff working with quarantined animals. If a specific quarantine facility is not present, then newly acquired otters should be kept separate from the established collection to prohibit physical contact, prevent disease transmission, and avoid aerosol and drainage contamination.

Wildlife institutions should have zoonotic disease prevention procedures and training protocols established to minimize the risk of transferable diseases with all animals, including those newly acquired in quarantine. Keepers should be designated to care only for quarantined animals if possible. If keepers have to care for both quarantined and resident otters, they should care for the quarantined animals only after caring for the resident animals. Equipment used to feed, care for, and enrich otters in quarantine should be used only with these otters. If this is not possible, then all items should be appropriately disinfected, as designated by the veterinarian supervising quarantine before use with resident otters.

Otters should be quarantined for a minimum of 30 days (unless otherwise directed by the staff veterinarian). This allows time for the development of clinical signs of disease that may have been incubating before the animal arrived and to acclimate to potentially new food and water. During the quarantine period, the animal should be observed for signs that may be associated with disease, such as sneezing, coughing, vomiting, diarrhea, ocular or nasal discharge, etc.

If additional carnivores are introduced into adjacent quarantine areas, the minimum quarantine period should begin over again. However, the addition of mammals of a different order does not require the re-initiation of the quarantine period.

During quarantine the previously fed diet, if known should be slowly adjusted over several weeks if there is to be a diet change. All social otters (e.g. P. brasiliensis and A. cinereus) should be housed in social groupings, as far as possible, during quarantine as extended periods alone can be detrimental to these social species causing development of stereotypical or self-mutilation behaviors such as pacing or over-grooming. On occasion an isolated L. canadensis in quarantine may exhibit an exaggerated escape response manifested by pacing and hyperactivity. If this behavior persists, hyperthermia, severe foot pad abrasions, extreme stress and death may ensue. Remediation of this problem can include adjustments to the enclosure (hide areas, low levels of ambient light, noise reduction, placing another otter in quarantine in visual contact with the otter, or even administering long acting neuroleptics or tranquilizers.

During the quarantine period, specific diagnostic tests should be conducted with each otter if possible. These include:

- A complete physical, including a dental examination if applicable.
- Otters should be evaluated for ectoparasites and treated accordingly.
- Blood should be collected, analyzed and the sera banked in either a -70ºC freezer or a frost-free -20°C freezer for retrospective evaluation.
- Fecal samples should be collected and analyzed for gastrointestinal parasites and the otters should be treated accordingly. Additionally cultures for salmonella sp. or campylobacter sp. may be submitted. Periodic fecal gram stains can be performed to monitor the relative numbers of *Clostridium sp* present in the intestinal tract (G. Kollias, personal communication)

- Vaccinations should be updated as appropriate, and if the vaccination history is not known, the otter should be treated as immunologically naive and given the appropriate series of vaccinations.

Depending on the disease and history of the otters, testing protocols for animals may vary from an initial quarantine test to yearly repetitions of diagnostic tests as determined by the veterinarian. Release from quarantine should be contingent upon normal results from diagnostic testing and three negative fecal tests that are spaced a minimum of two weeks apart. Medical records for each animal should be accurately maintained and easily available during the quarantine period.

Species-specific recommendations:

P. brasiliensis and L. canadensis should be checked for *Strongyloides* species (Wünnemann 1990; C. Osmann, personal communication).

QUARANTINE FACILITIES

Ideally, quarantine facilities should be isolated from the risk of cross-contaminating other carnivores already in the collection. If this is not possible, different keepers should be used, or strict rules of personal hygiene should be adopted and resident animals should be cared for before quarantine animals. Balancing between the necessity of keeping the quarantine pen clean and the needs of the animals can be challenging. Many of the otter species do better when isolated in enclosures than when placed in hospital-type quarantine pens (Lewis 1995). If this is not practical or possible, a privacy box, climbing furniture, substrate suitable for rubbing/drying-off, and a pool or water tub suitable for swimming should be provided. Whatever type holding facility is used, care should be taken to ensure that otters cannot escape by climbing, digging, or chewing their way out.

Quarantine Exams: Two quarantine exams are recommended for otters; one performed at the beginning of the quarantine period and one performed at the end.

Initial exam: Veterinarians should visually inspect otters as soon as possible after they have arrived in quarantine.

Final exam: During the last week of quarantine, a thorough examination should be conducted, see medical examinations under Preventive Medicine.

PREVENTIVE MEDICINE

MEDICAL EXAMINATIONS

If possible, all animals should have at least a biennial (every other year) examination. For otters greater than 6 years of age examination should be performed annually (G. Kollias, personal communication). During examination the following procedures are performed**:**

- Transponders and/or tattoos should be checked and reapplied if they are not readable.
- Baseline physiological parameters (e.g., heart rate, weight, body temperature, respiratory rate) should be obtained & recorded.
- The oral cavity and all dentition should be examined. Teeth should be cleaned and polished if necessary. Any tooth that is fractured or in need of repair should be noted in the medical record and the condition corrected as soon as possible Radiographic changes present in affected teeth should be used to substantiate the need for treatment or extraction.
- The reproductive tract should be evaluated. Care should be taken to record any changes in the external genitalia, such as vulvar swelling or discharge, testicular enlargement, and mammary gland changes. Contraceptive hormone implants also should be checked to make sure they are in place, and not causing any local irritation.
- Radiographs taken to check for any abnormalities. If renal or cystic calculi are seen, then numbers, location, and approximate sizes should be noted in the records.
- Blood collection done, and complete blood count and chemistry profile performed. For *P. brasiliensis*, blood samples can be taken from the *V. cephalica antebrachii* of the foreleg (C. Osmann, personal communication).
- Blood serum frozen and banked when possible.
- Animals housed outside in heartworm endemic areas should be checked for heartworm disease by performing a heartworm ELISA antigen test and the animal routinely given heartworm preventative treatment (see ‘parasite control’ section).
- Urine collected whenever possible by cystocentesis for a complete urinalysis. For *P. brasiliensis*, rather than cystocentesis, which is associated with the risk of bacterial infection, it is possible to gain urine from an immobilized animal by manual squeezing of the caudal abdomen (female) or by catheterizing the urethra (C. Osmann, personal communication).
- An annual fecal examination should be performed to check for internal parasites, and anthelmintics administered if necessary (see ‘parasite control’ section). For *P. brasiliensis*, biennial fecal examinations are recommended (C. Osmann, personal communication).
- Vaccines updated if necessary (see ‘vaccination’ section).

Anesthesia/immobilizations of giant otters should only be performed when there is a medical indication for the procedure. Preventive immobilizations are of high risk for the animals, and should be substituted with regular visual examinations and testing of fecal samples, vaccinations etc. Administration of transponders, examination of the oral cavity, blood sampling etc., should be completed only when immobilizations are necessary for medical reasons. Evaluation of the reproductive tract can be performed in animals that are regularly involved in a medical training program. In well-trained animals, sonography of the uterus may be possible, as well as the visual or palpable inspection of mammary gland and testicles.

VACCINATION

The following vaccination schedule is recommended by OCT Veterinary Advisors. Vaccination product recommendations are based on clinical experience (as of 2006) in most cases, and not necessarily on controlled scientific study.

Canine distemper*:* Merial's new PureVax™ Ferret Distemper Vaccine currently on the market is a univalent, lyophilized product of a recombinant canary pox vector expressing canine distemper virus antigens. The vaccine cannot cause canine distemper under any circumstances, and its safety and immunogenicity have been demonstrated by vaccination and challenge tests. Otters should initially be given 1ml of reconstituted vaccine for a total of 2-3 injections at three-week intervals, followed by a yearly booster.

> (NOTE: annual vaccinations for domestic dogs and cats over the age of 6 years are considered unnecessary or at least controversial if they have been vaccinated on an annual basis for the first 6 years of life (an exception would be vaccination for rabies which has legal vs. medical implications for its administration; a variety of medical problems have been reported in older dogs and cats given routine vaccinations; serum neutralizing antibody titers can be requested to determine the level of immunity prior to vaccination; if the titers are sufficient to prevent infection then vaccination can be delayed until or if the titers decrease below what is considered to be a protective level (G. Kollias, personal communication).

This vaccine should be given IM instead of SQ in exotic carnivores for increased effectiveness (Merck Veterinary Manual, accessed November 2011). More information on PureVax™ Ferret Distemper Vaccine can be found at www.us.merial.com (Merial Ltd., 3239 Satellite Blvd., Duluth, GA 30096). An alternative vaccine that is available is Galaxy D (Schering-Plough Animal Heath Corporation, P.O. Box 3113, Omaha, NE 68103), a modified-live canine distemper vaccine of primate kidney tissue cell origin, Onderstepoort type (this vaccine is not available in Europe). Safety and efficacy of canine distemper vaccinations in exotic species of carnivores have been problematic. Vaccine-induced distemper has occurred in a variety of mustelids using modified-live vaccine, and killed vaccines have not provided long-lived protection and are not commercially available. However, to date there have been no cases of vaccine induced distemper in otters given the Galaxy product, and excellent seroconversion following vaccination using this product has been documented in young N.A. river otters (K. Petrini, unpublished data, Petrini et al. 2001). The use of any modified-live canine distemper vaccine in exotic species should be done with care, especially with *P. brasiliensis*, young animals, and those that have not been vaccinated previously. The use of PureVax™ Ferret Distemper Vaccine is recommended where possible.

Parvovirus*:* The efficacy of feline and canine parvovirus vaccines has not been proven in otters. Otters should initially be given 1ml of vaccine IM for a total of 2-3 injections at three-week intervals followed by a yearly booster. Parvocine™ (Biocor Animal Health Inc., 2720 North 84th Street, Omaha, NE 68134) is a killed univalent parvovirus vaccine that has been used in otters. Using a univalent product such as Parvocine™ reduces the risk of vaccine allergic reactions.

Rabies*:* The efficacy of rabies vaccines has not been proven in *Lontra canadensis* or other exotic mustelids. Vaccinated otters that bite humans should not be considered protected from rabies. Only killed rabies products should be used in otters. One commonly used product is Imrab® 3 (Merial Ltd., 3239 Satellite Blvd., Duluth, GA 30096), which is a killed rabies vaccine that has been used extensively in small carnivores without apparent adverse effects. Otters should be given 1ml of vaccine IM once at 16 weeks of age followed by a yearly booster. PureVax™ Feline Rabies (Merial Ltd., 3239 Satellite Blvd., Duluth, GA 30096) is a live canarypox vectored, non-adjuvanted recombinant rabies vaccine that is currently being used at some institutions for small carnivores. Dosage and route are the same as for Imrab® 3, but this vaccine can be given once at age 8 weeks or older, then annually.

Leptospirosis: The susceptibility of river otters to leptospirosis is debated in the literature, and the benefit of vaccination is unknown. Killed *Leptospira* bacterins are available and can be administered in areas where leptospirosis has been problematic. Initially two doses should be given at 3-4 week intervals. Vaccine efficacy and duration of immunity has not been studied in the otter and is an area where further study should be conducted.

PARASITE CONTROL

Otters should have fecal examinations performed regularly (annual exams are advised, but at least biennially; some advisors prefer them to be done twice per year). The frequency of these examinations depends on the incidence of parasitism in the geographic region and the likelihood of exposure. Animals also should be screened for parasites before shipment to another facility, release, and during quarantine. Fecal testing should include a direct smear examination and a fecal flotation, as well as sedimentation techniques, if possible. Baermann fecal examination techniques help identify certain parasites, such as lungworms, that are otherwise difficult to detect.

Heartworm ELISA antigen tests should be conducted annually in animals exposed to mosquitoes in heartworm endemic areas and animals maintained on a heartworm preventative (e.g. North America). External parasites (e.g., mites, fleas, ticks) can be detected during physical examinations.

Dr. George Kollias, Cornell University School of Veterinary Medicine, states: "*Dirofilaria immitis*, the cause of heartworm disease in dogs, cats and some other carnivores, has been found in the hearts of otters in and from Louisiana. This filarial worm has to be differentiated from *Dirofilaria lutrae*, the microfilaria of which can be found in the blood and adults in the subcutaneous tissues and coelomic cavity of river otters. *D. lutrae* generally does not cause disease. Newly acquired otters should be screened for microfilaria (via the Knott's test on blood) and for adults, via the ELISA antigen test on serum. *D. immitis* can be differentiated from *D. lutrae* by the morphological appearance of the microfilaria and by the antigen test. Thoracic radiographs should also be taken as part of routine health screening and definitely if an otter is Knott's test positive and/or antigen positive". See also Snyder et al. (1989), Neiffer et al. (2002), and Kiku et al. (2003) for reports of heartworm in otters.

In heartworm endemic areas, otters can be given ivermectin (0.1mg/kg orally once/month year around) as a preventative. Although it is still uncertain whether or not *D. immitis* causes progressive heartworm disease, as in the dog and cat, prevention is safest approach. If used at the proper dose, ivermectin has proven safe in otters. Mortality has been associated with Melarsomine dihydrochloride administration to North American river otters and a red panda for heartworm disease (Neiffer et al. 2002). In another report of otter deaths after treatment with Melarsomine, adult heartworms were found in the hearts of three out of the four animals during necropsy (G. Kollias, personal communication).

- **Parasite Testing***:* Recommendations and protocols for parasite testing in otters are provided in the following table:

Table 1: Otter parasite testing protocols

Parasite	Testing protocol
External parasites	Regular inspections during any physical examinations
Internal parasites	Annual fecal examinations: direct smear, fecal flotation, & sedimentation or Baermann techniques. Pre-shipment fecal examinations*:* direct smear and flotation Quarantine fecal examination: 3 negative direct smear results & 3 negative fecal flotation results before release from quarantine. Heartworm ELISA antigen tests: conducted annually in animals exposed to mosquitoes in heartworm endemic areas (test will not detect all male infections or infections with < 3 female nematodes). If infection is suspected, positively identify the microfilaria as pathogenic before instituting treatment.

- **Parasite Treatment***:* The following table (Table 2) provides a list of anthelmintic products that have been used safely in a variety of mustelids:

Table 2: Recommended anthelmintic treatments for otters

Treatment	Dose
Fenbendazole	50mg/kg orally for 3-5 days. In *P. brasiliensis* there was a complete elimination of a *Strongyloides spp*. infestation after treatment with 10-20mg/kg over 3 days
Pyrantel pamoate	10mg/kg orally
Ivermectin	0.1mg/kg orally, once monthly for heartworm prevention 0.2-0.4mg/kg subcutaneously or orally for treatment of intestinal nematodiasis (G.Kollias, personal communication)

Praziquantel	5mg/kg SC or orally

Medical Management of Neonates

Otter cubs can develop health issues suddenly, and they should be carefully watched for any change in behavior. Some problems that have developed in young hand-reared cubs are listed below with suggested first-step solutions or treatments, and neonatal examination and monitoring guidelines are also provided. See Table 3.

Table 3: Neonatal examination & monitoring guidelines (from Read & Meier 1996)

Vital signs	Temperature, include activity level Pulse, rate and character Respiration, rate and character
Organ systems	---
Weight	---
Hydration	Skin tone and turgor
Mucous membranes	Color and capillary refill
Vitality	Response to stimulation, activity levels: type, frequency, duration
Physical condition	---
Laboratory values (optional)	Complete blood count White blood cell count Serum chemistries, including blood glucose & blood urea nitrogen Urinalysis and urine specific gravity (recommended)
Urination	Frequency, amount, and character
Defecation	Frequency, amount, and character
Condition of umbilicus	---
Total fluid intake	Amount in 24 hours Parenteral fluids, amount, frequency, and type Oral fluids, amount, frequency, type, nipple
Housing temperature	---

Dehydration/Emaciation: Give subcutaneous or oral (only if sucking well) electrolytes. Lactated Ringers Solution (LRS) with 2.5% dextrose or sodium chloride (0.8% NaCl) are recommended. Oral fluids are given at the dose of 5% body weight per feeding. The dose for subcutaneous fluids is determined by the level of dehydration and should be determined by a veterinarian.

P. brasiliensis: Parenteral volume substitution can be made with Amynin® (full electrolyte, glucose, amino acid solution offered by Merial), a mixture of 50% Ringer Lactate (Lactated Ringer's solution), + a 50% solution of 5% Dextrose, for a total dosage of about 10% of body weight/day. Oral fluids for children, such as Humana Elektrolyt®, are a good option to use in otter cubs. Weak cubs may be gavage-fed by someone experienced in inserting a tube into the stomach and injecting formula directly into the digestive tract. This is a risky endeavor, as the stomach-tube can be accidentally inserted into the trachea resulting in milk infusion directly into the lungs. In general, if cubs are too weak to suckle, their gastrointestinal tracts are too compromised to digest food and they require immediate veterinary care. The administration of Ringer Lactate solution before attempting to feed a weak cub is advised. Administration of paramunity inducers, such as Zylexis® (Pfizer - www.pfizer.com/pfizer/main.jsp), is recommended in weak and less vital cubs (C. Osmann, personal communication).

Diarrhea/Constipation: Digestive upset is a common issue with hand-reared neonates, and may be associated with several factors (Meier 1985): inappropriate milk formula, feeding frequency, overfilling the stomach which can cause bloating, and rapid changes in the diet. When digestive upset occurs, characterized by diarrhea, bloating, inappetance, and/or extreme disorientation, it is recommended that one factor is analyzed and/or changed at a time. The veterinarian should be consulted immediately in the case of diarrhea, as the condition of very young animals can deteriorate rapidly.

Diarrhea related to diet changes may be treated with Kaopectate® with veterinary approval. It should be noted that Kaopectate® now contains salicyclic acid (aspirin), as does Pepto-Bismol®, and gastrointestinal bleeding may result from frequent doses. Persistent diarrhea, or loose stool accompanied with inappetance, requires continuous veterinary care. Bacterial infections or parasites, such as coccidia may be the cause of the problem and require specific medication. Osmann (personal communication) recommends the administration of *Lactobacillus spp*. into the formula for *P. brasiliensis* cubs with diarrhea or after antibiotic treatment. Veterinarians should consider this for all otter species.

Constipation may be treated by diluting the formula to half-strength for 24 hours, and gradually increasing back to full-strength over a period of 48 hours. The cub also can be given oral electrolyte fluids at the rate of 5% body weight in between feedings and 1-2 times over a 24-hour period. The cub's back end can be soaked for a few minutes in warm water (make sure to dry off completely) accompanied by gentle stimulation, but care should be exercised that the anal area is not irritated.

Upper Respiratory Infections: Cubs that have been eating normally and suddenly start chewing on the bottle or seem uninterested in the bottle may have an upper respiratory infection. They cannot nurse properly when congested. Upper respiratory infections need to be treated immediately. Newborn cubs can die within 24 hours of the first symptom. Antibiotics should be started at the first sign of infection.

Antibiotics can be given orally or injected. Care should be taken with the location of injections to avoid the sciatic nerve in their rear limbs (in two cases where limb mobility was affected due to injection site, the lameness/paralysis was resolved over time). Cubs on antibiotics may also develop GI problems and/or get dehydrated, and this should be treated accordingly. Antibiotics that have been used successfully for upper respiratory infections are listed below. Antibiotics should not be given without consulting a veterinarian first.

- Enrofloxacin: injectable at 5mg/kg BID IM or SC
- Amoxicillin: 20mg/kg BID PO
- Amoxicillin (long-acting): 15mg/kg IM every 48 hours (*P. brasiliensis*)

- Penicillin G Procaine: 40,000-44,000 IU/kg q24 hr IM or SC
- Chloramphenicol: administered orally at 30-50mg/kg/day (*P. brasiliensis*)
- Trimethoprim/sulfonamide combination: given parenteral at 15mg/kg/day (*P. brasiliensis* – C.Osmann, personal communication)

Bloat: Some otter cubs have developed bloat. Care should be taken to ensure that there is no air in the formula or any leaks in the bottles. The amount of formula fed at each feeding should be re-evaluated as the cub may be receiving too much. Reducing the amount fed per feeding and adding another feeding should be considered. Watch for respiratory distress as respiration may become labored with severe abdominal distention. Treatment options for bloat include passing tubing to decompress, or the use of over-the-counter medication. Infant gas drops have been tried with no effect. Care should be taken with attempting the use of over-the-counter medications, such as certain gastric coating agents (e.g. bismuth subsalicylate (Pepto-Bismol®), as some ingredients may create more problems.

Fungal Infections: Caretakers should look for hair loss and discoloration of skin, and should pull hair samples and culture for fungus using commercially available fungal culture media. At first appearance, fungal infections can be treated with shampoos and creams, and shaving the affected areas can also help. Severe infections may need to be treated with oral/injectable medication.

Parasites: Fecal samples should be taken regularly from otter cubs (specifically hand-reared cubs), even if they are previously negative. Cubs should be dewormed as needed and treatment started immediately to avoid any weight loss.

Bite/Puncture Wounds: Any bite or puncture wounds should first be cleaned and flushed with fluids, and then treated with topical antibiotic and systemic antibiotics if necessary.

CAPTURE, RESTRAINT, AND IMMOBILIZATION

The need for capturing, restraining and/or immobilizing an otter for normal or emergency husbandry procedures may be required. All capture equipment should be kept in good working order and available to authorized and trained animal care staff at all times.

It is recommended that anesthesia be given to otters intramuscularly (IM) in the cranial thigh (quadriceps), caudal thigh (semimembranosus-tendinosus), or paralumbar muscles (Spelman 1999). Animals should be kept as quiet as possible. Generally, restraint is accomplished using a net, squeeze cage, or capture box. If possible, training animals to receive injections minimizes stress prior to all anesthesia events. Immobilization of *P. brasiliensis* using a blowpipe has proven to be relatively easy and minimizes stress to the animals involved. Osmann (personal communication) recommends darting the animal in the M. biceps femoris/semimembranosus/semitendinosus. A variety of agents have successfully been used in otter species for immobilization. These include Ketamine alone (not recommended), Ketamine with midazolam, Ketamine with diazepam, and Telazol®.

Otters have a large respiratory reserve, and so using gas induction chambers is often very time consuming (this can take up to 10 minutes in *A. cinereus*), but has been done successfully. Despite the method of induction, anesthesia can be maintained by intubating the animal and maintaining it on Isoflurane (Ohmeda Pharmaceutical Products Division Inc., P.O. Box 804, 110 Allen Rd., Liberty Corner, NJ 07938). Halothane (Fort Dodge, 9401 Indian Creek Parkway, Ste. 1500, Overland Park, KS 66210) is <u>no longer recommended for use in otters as it may cause liver failure</u> (G. Meyers, personal communication). Otters are relatively easy to intubate, and this method is preferred when it is necessary for an animal to be immobilized for a lengthy procedure (>30 minutes). Intermittent positive pressure ventilation must be provided for lengthy procedures. This is essential because otters do not adequately spontaneously ventilate when inhalation anesthetic agents are employed. Hypoventilation and subsequent hypoxemia may be a primary cause for mortality in anesthetized otters. (Kollias & Abou-Madi 2007).

Careful monitoring of anesthetic depth and vital signs is important in any immobilization. Body temperature, respiratory rate and depth, heart rate and rhythm, and mucous membrane color and refill time should be assessed frequently. Pulse oximetry sites include the tongue, the lip at the commissure of the mouth, or in the rectum. Oxygen supplementation should be available and administered when indicated.

<u>A. cinereus:</u> For A. cinereus, ketamine hydrochloride can be used alone or in combination with midazolam hydrochloride (Versad®, Roche Labs, 340 Kingsland St., Nutley, NJ 07110-1199) or diazepam to improve muscle relaxation (Petrini 1998). Telazol® (Fort Dodge, 9401 Indian Creek Parkway, Ste. 1500, Overland Park, KS 66210) is another good immobilizing agent for this species. Generally, it provides smooth, rapid induction and recovery along with good muscle relaxation. Doses of Telazol® required for adequate immobilization vary considerably between individuals. Ranges for some injectable drug combinations are listed below:

- Telazol: 5.5-9.0mg/kg IM
- Ketamine: 12-15mg/kg & midazolam: 0.5-0.75mg/kg IM
- Ketamine: 9-12mg/kg & diazepam: 0.5-0.6mg/kg IM

Muscle rigidity is common with these injectable drug combinations at the lower end of the dosages. Initial apnea and low oxygen saturation readings, as measured by pulse oximetry, often accompany higher doses. All three combinations produce a relatively short duration of anesthesia time, approximately 15-30 minutes. Administering an additional 5mg/kg ketamine IM when needed can prolong anesthesia time. Alternatively, the animal can be intubated and maintained on gas anesthesia.

Combining ketamine with medetomidine hydrochloride (Domitor®, Pfizer Animal Health, 812 Springdale Dr., Exton, PA 19341) may provide a slightly longer duration of anesthesia and may give better myorelaxation; it also has the added advantage of being reversible with atipamezole hydrochloride (Antisedan, Pfizer Animal Health, 812 Springdale Dr., Exton, PA 19341). Vomiting may occur during induction, and initial apnea and low oxygen saturation readings are common. Supplemental oxygen should be available for administration if necessary. Dosages that have been used successfully are:

- Ketamine: 4-5.5mg/kg & medetomidine: 0.04-0.055mg/kg IM; reversed with atipamezole: 0.200-0.275mg/kg IM. Caution: medetomidine administered to hypothermic can result in mortality (G. Kollias, personal communication).

L. canadensis: For short-term anesthesia (25-30 minutes) of L. canadensis, Spelman (1998) recommends the following:

- Ketamine: 10mg/kg & midazolam: 0.25mg/kg
- Ketamine: 15 mg/kg & midazolam 0.3 mg/kg or diazepam 0.5 mg/kg (for otters greater than 4.5 kg bw) or Ketamine: 20 mg/kg & midazolam 0.3mg.kg or diazepam 0.5 mg/kg (for otters less than 4.5 kg bw) (Kollias and Abou-Madi 2007,IBID)
- Ketamine: 2.5-3.5mg/kg, medetomidine: 0.025-0.035mg/kg & atipamezole: 0.125mg/kg (respiratory depression is more likely at higher dosages).
- Telazol®: 4mg/kg (Spelman 1998), 9mg/kg (Blundell et al. 1999; Bowyer et al. 2003), 8mg/kg (Petrini et al. 2001); reversed by Flumazenil: 0.08mg/kg to prevent a prolonged recovery time.
- Ketamine: 10mg/kg. Muscle rigidity and variable duration should be expected.
- Ketamine: 5-10mg/kg & diazepam: 0.5-1mg/kg. Prolonged recovery compared to ketamine with midazolam.

P. brasiliensis: Due to their large size, a deep IM injection is recommended for good anesthesia. The breathing of the animals should be carefully monitored, and the temperature tested frequently to avoid hyperthermia (L. Spelman, personal communication, 2007). The following anesthesia protocols have been used with giant otters:

- Ketamine at 7.5mg/kg (5-10mg) in combination with xylazine at 1.5mg (1-2mg/kg). Combining Ketamine with xylazine (Rompun® 2%, BayerVital GmbH, 51368 Leverkusen) gives a short-term anesthesia with good muscle relaxation and analgesia. Xylazine may be reversed with atipamezole (Antisedan®, Pfizer GmbH, Pfizerstraße 1, D-76139 Karlsruhe) (C. Osmann, personal communication).
- Give xylazine at 2.5mg/kg, wait 15 minutes and give ketamine at 2.5mg/kg; when done, reverse with yohimbine (L. Spelman, personal communication, 2007).
- For a single injection, use medetomidine 0.030mg/kg and ketamine 3mg/kg, and reverse with atipamezole 0.125mg/kg. Although easier to use, this regimen can lead to poor breathing at the start of the procedure (L. Spelman, personal communication, 2007).
- Supplemental oxygen should always be available for administration, if necessary. For longer procedures, animals should be maintained on Isoflurane.

MANAGEMENT OF DISEASES, DISORDERS, INJURIES AND/OR ISOLATION

Wildlife institutions should have a veterinary program that manages animal diseases, disorders, or injuries and has the ability to isolate these animals in a hospital setting for treatment if necessary. Staff should be trained for meeting the animal's dietary, husbandry, and enrichment needs, as well as in restraint techniques, and recognizing behavioral indicators animals may display when their health becomes compromised. Protocols should be established for reporting these observations to the veterinary department. Hospital facilities should have x-ray equipment or access to x-ray services, contain appropriate equipment and supplies on hand for treatment of diseases, disorders or injuries, and have staff available that is trained to address health issues, manage short and long term medical treatments and control for zoonotic disease transmission. Institutions also should have a clear process for identifying and addressing animal welfare concerns within the institution; this process should identify the protocols needed for animal care staff members to communicate animal welfare questions or concerns to their supervisors. Protocols should be in place to document the training of staff about otter welfare issues, identification of any otter welfare issues, coordination and implementation of appropriate responses to these issues, evaluation (and adjustment of these responses if necessary) of the outcome of these responses, and the dissemination of the knowledge gained from these issues.

All living animals will die at some point. As care givers for the animals residing in our zoos and aquariums, it is vital that we provide the best care possible for them until the time their health deteriorates to a point where euthanasia is the most humane treatment, or the animal dies on its own. Necropsies should be conducted on deceased otters to determine their cause of death and the subsequent disposal of the body must be done in accordance with any local, state, or federal laws. Necropsies should include a detailed external and internal gross morphological examination and representative tissue samples from the body organs should be submitted for histopathological examination if possible. Selected tissues should be frozen at -20F for bacterial isolation or -70C for virus isolation if supported by the histopathological findings.

Illnesses commonly seen in otters: Little information on common diseases and disorders for *A. capensis* and *L. maculicollis* is available, and more research is required for these species. Urolithiasis is the most common illness, and renal calculi are the most frequent cause of death in *A. cinereus*. The cause and reversal of this condition is the subject of ongoing research.

Poor Coat Quality: Otters are amphibious mammals reliant on trapping air within their coats rather than a layer of blubber for thermal insulation (Tarasoff 1974). Studies have shown that otter fur is far denser than that of other mammal species, with an average of 26,000 hairs/cm^2 (foot) to 165,000 hairs/cm^2 (foreleg) (Weisel et al. 2005). Sea otter pelts are roughly twice as dense as the fur of a river otter, and the river otters' fur is twice as dense as that of a mink (Weisel et al. 2005).

Weisel et al. (2005) determined, via the use of scanning and polarizing light microscopy, that otter guard and underhairs are characterized by the presence of fins, petals, and grooves that allow adjacent hairs to fit together forming an interlocking structure. Trapped within this interlocking structure are bubbles of air forming an insulating layer between the skin and water. Air is trapped in the fur when the otter shakes upon emerging from the water, via piloerection of the hairs (including grooming and rubbing), and muscular pleating of the skin (Weisel et al. 2005). Thus, behavioral actions combined with the density and complexity of the underfur structure essentially prohibits water from touching the skin. Weisel et al. (2005) also determined that the outer and inner hairs of an otter's coat are, "… hydrophobic due to the presence of a thin layer of body oil from the sebaceous glands of the otter."

This recent work documented that the long, outer hairs do guard the more fragile inner fur from damage and that they can become damaged, reducing their effectiveness. At this time it is not possible to say what damage is done to the otters' guard hairs by gunite or other abrasive surfaces within *ex-situ* exhibits, but many professional zoo and aquarium organizations recommend that those surfaces be avoided in otter exhibits as far as possible.

Poor coat quality and other factors (e.g. chronic hypothermia and subsequent immunosuppression) can lead to bacterial, viral or protozoal pneumonia. Poor coat quality is of concern when its water repellency is affected. If water does not form droplets and cannot be easily shaken off the guard hairs (i.e., dark brown fur), the otters' guard hairs clump together resulting in a coat that looks slick and saturated; this is an indication of poor coat quality. Poor quality leads to water penetrating the guard hairs and exposure of the under-fur (gray/white under coat), which can then become waterlogged. An otter in this condition may not swim in an effort to remain as dry as possible. If the otter does swim, and it cannot keep dry, its body temperature will drop rapidly leading to observable shivering, even during sleep. Enteritis can develop in cases of extreme chilling. If measures are not taken, death can follow in a matter of days through pneumonia and/or gastro-intestinal complications (Duplaix-Hall 1972). Insufficient land area compared to water area, and/or inappropriate enclosure substrates causing overly damp/wet conditions, were historically most often the reason for poor coat condition and the resulting health problems in river otters (Duplaix-Hall 1972, 1975). For the giant otter, this is still the most frequent cause of poor fur condition and related health problems; no other environmental or physical conditions have been reported to cause these coat problems except in one case of an unrelated serious illness (Sykes-Gatz 2005, unpublished data).

COMMON DISEASE ISSUES

Dr. Gwen Myers, AZA Otter SSP and OCT Veterinary Advisor conducted a review of all submitted necropsy reports for *L. canadensis*. Her findings (Table 4) indicate that the most frequent causes of *L. canadensis* deaths (excluding neonatal deaths).

Table 4: Common causes of deaths in *L. canadensis.*

Cause of death	Causal factors
Heart disease	Heartworm/death from heartworm treatment; Acute myocarditis; Myofiber degeneration
Renal failure	Etiology unknown; Amyloidosis; Pyelonephritis
Hepatic lipidosis	
Adenocarcinoma	
Transitional cell carcinoma (bladder)	
Peritonitis	Secondary to intestinal perforation from foreign body; Secondary to GI perforation from ulcers
Diarrhea	Unknown etiology; Clostridial endotoxin; *Helicobacter* (also causing vomiting, weight loss); *Salmonella*
Gastric dilatation with volvulus	
Pneumonia	Often without identifying underlying cause; etiology for pneumonia often involves bacterial infection which leads to their death.
Anesthetic death	

P. brasiliensis: Causes of death have included: leptospirosis, parvovirus, bronchopneumonia/pneumonia, internal bleeding, gastroenteritis, intussusceptions, severe inbreeding resulting in inherited thyroid malfunction in cubs, parental or older sibling neglect of, or mistreatment towards cubs due to stress from human disturbances or inappropriate insufficient land vs. water area and/or enclosure substrate conditions, inappropriately conducted introductions of unfamiliar or temporarily separated otters, heart failure, kidney failure, pyrometra, and exposure to continually very damp or wet conditions (Osmann & Wisser 2000; Sykes-Gatz 2005, unpublished data; C. Osmann, personal communication).

Common medical problems in this species include skin lesions, particularly on the tail and hind legs. These often become infected with *Staphylococcus spp*. and typically respond well to topical and/or systemic antibiotics (C. Osmann, personal communication). Progressive walking difficulties involving the lower back or hind legs also are reported in this species, particularly in animals aged 4 years and over. Again, the causal agent appears to be continued overexposure to hard surfaces. Other physical problems caused by overexposure to hard or continuously wet or very damp conditions include foot pad abrasions, irritation of the foot's webbing, and poor coat condition (Sykes-Gatz 2005). These conditions can be caused or exacerbated by exposure to coarse substrates.

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