Otter (Lutrinae) Care Manual

Created by the
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in Association with the
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Table of Contents

Introduction

Preamble

Association of Zoos & Aquariums (AZA) accredited institutions are required to comply with all relevant local, state, and federal wildlife laws and regulations in addition to all AZA accreditation standards.

AZA accreditation standards are continuously being raised or added. Staff from AZA-accredited institutions are required to know and comply with all AZA accreditation standards, including those most recently listed on the AZA website (www.aza.org) which might not be included in this manual.

Taxonomic Classification

Table 1: Taxonomic classification for Lutrinae

Classification	Taxonomy	Additional information
Kingdom	Animalia	
Phylum	Chordata	
Class	Mammalia	
Order	Carnivora	
Suborder	Caniformia	
Family	Mustelidae	
Sub Family	Lutrinae	Some species names are still being debated

Genus, Species, and Status

Table 2: Genus, species, and status information for Lutrinae recommended for management by the AZA Otter SSP

Genus	Species	Common Name	USA Status	IUCN Status	AZA Status
Aonyx (Amblonyx)	*Cinereus (cinerea)**	Asian small-clawed otter	Not listed	Vulnerable	SSP
Lutra (Hydrictis)					
	maculicollis	Spotted-necked otter	Not listed	Least Concern	SSP
Pteronura					
	brasiliensis	Giant otter	Endangered	Endangered	SSP
Lontra (Lutra)	*canadensis*	North American river otter	Not listed	Least Concern	PMP

*Some recent resources are still using *Aonyx cinerea* (Wilson & Reeder 2005) however, the IUCN/SSC Otter Specialist Group and ITIS have switched to *Aonyx cinereus* based on input from Koepfli & Wayne (1998, 2003) and advice that the appropriate Latin gender declination is *cinereus* and not *cinerea;* we are using the OSG as our citing source (www.otterspecialistgroup.org/). ISIS uses *A. cinereus.*

General Information

Introduction: The AZA Small Carnivore Taxon Advisory Group (TAG) has designated four semi-aquatic otter species for management under the AZA Otter Species Survival Plan® (SSP). These are the Asian small-clawed otter (*Aonyx/Amblonyx cinereus*), giant otter (*Pteronura brasiliensis*), North American (Nearctic) river otter (*Lontra canadensis*), and the spotted-necked otter (*Lutra maculicollis*). The African (Cape) clawless otter (*Aonyx capensis*) is currently represented in AZA institutions but is recommended for phase-out. Because specimens are still maintained by member institutions, care information is included. These otter species exhibit varying needs and degrees of sociality. Many of their *ex-situ* population care requirements are similar but there are significant variations in some of their husbandry needs. For more detailed information please refer to the following husbandry manuals:

- Asian Small-clawed Husbandry Manual (Lombardi et al. 1998)
- International Giant Otter Studbook Husbandry and Management Information and Guidelines 2005, 2nd Edition (Sykes-Gatz 2005)
- North American River Otter Notebook, 3rd Edition (Reed-Smith 2008) and Summary of Husbandry Guidelines for North American (aka Nearctic) River Otters (*Lontra canadensis*) in Captivity (Owens et al. 2009): www.otterspecialistgroup.org/Library/TaskForces/OCT.html.
- Summary of Husbandry Guidelines for African Spotted-necked Otters in Captivity (Benza et al. 2009): www.otterspecialistgroup.org/Library/TaskForces/OCT.

Natural History/Description:

Asian small-clawed otter (*Aonyx cinereus*): This is one of five otter species found in Asia. It is one of the smallest of the world's otters, rarely weighing more than 5kg. A gregarious species, it is often seen in large groups of up to 15 animals, and *ex-situ* population studies suggest that these groups are composed of an alpha breeding pair and their offspring from successive litters. Asian small-clawed otters have unusual hand-like front paws with increased tactile sensitivity and reduced webbing, which they use to forage for their prey of crustaceans, mollusks, and small fish.

Asian small-clawed otters are found from Palawan (Philippines) through Indonesia, Southeast Asia, southern China, and westwards throughout the Himalayan foothills of Bangladesh, Bhutan, and Nepal. A disjunctive population occurs in southern India (Foster-Turley et al. 1990). Listed on CITES as Appendix II (www.cites.org) and as Vulnerable by IUCN/SSC the population is considered to be decreasing.

African (Cape) clawless otter (*Aonyx capensis*): The African clawless otter is one of four species of otters found in Africa. It is the third largest species of otter. Only the giant otter and sea otter are larger. Adults range in size from 1.15-1.5m (3.8-5ft), and weigh from 16-20kg (35.3-44.1lbs) (Foster-Turley et al. 1990). The African clawless otter has been reported as living in family groups including the male, female, and pups (Rowe-Rowe 1978), family groups consisting of the female and pups, or singly (Chanin 1985). The prevalent social grouping may vary with the habitat, which also likely influences the size and degree of overlap of home ranges. African clawless otters use their sensitive, non-webbed fingers to forage food, which consists primarily of fresh-water crabs, crayfish, and some fish. In some areas, this species is reported to occasionally raid near-by farms for young maize and cabbages (J.Reed-Smith, personal communication).

This species is distributed from Ethiopia in the east to Senegal in the west and south to South Africa, with a distributional gap in the rain forest area of the Congo basin, where the Congo clawless otter is found (Rowe-Rowe 1991). The African clawless otter is found in both fresh water streams and rivers, and along marine coastlines in South Africa.

Due to the infrequency of the holding and exhibition of the African clawless otter in zoos and aquariums, many of the standards set for this species are extrapolated from those set by the N.A. river otter and Asian small-clawed otter husbandry manuals. Listed on CITES as Appendix II (www.cites.org).

Giant otter (*Pteronura brasiliensis*): This single species in the genus *Pteronura* is one of four species of otter found in South America. The giant otter's large size (1.5-2m, 4.9-6.6ft), weight (25-32kg, 55.1-70.5lbs) (Duplaix 1980), highly social nature (multi-generational family groups), and critically endangered status make this species attractive to many facilities. However, their specific housing requirements and sensitivity to disturbances make them one of the more difficult otter species to hold successfully in zoos and aquariums. The diet of the giant otter is comprised almost completely of fish.

Although originally found in Colombia, Venezuela, Guyana, French Guiana, eastern Ecuador, Peru, Brazil, Bolivia, Uruguay, Paraguay, Suriname and northeastern Argentina, only remnant populations of giant otter are currently found throughout its former range. It is mainly found in slow moving rivers and creeks within forests, lakes, ox-bow lakes, swamps, and marshes in the tropical lowland areas of South America. With an estimated total population of only 1,000-5,000 individuals, the giant otter is considered highly vulnerable to extinction. It is classified as Endangered by the World Conservation Union (IUCN), as Endangered by the US Fish and Wildlife Service, and is listed on Appendix I of the Convention on International Trade in Endangered Species (CITES). Historically hunted for pelts, the species is now threatened by increased human colonization of tropical lowland rainforests. Other threats include habitat destruction and degradation, over-fishing, illegal hunting, mining, and water and land pollution.

North American river otter (*Lontra canadensis*): The North American river otter is one of the four new world river otter species. There are at least seven subspecies of *L. canadensis*. Adults range in size from 1-1.53m (3.3-5ft) and weigh from 4.5->16kg (9.9-35.2lbs) (Ben-David et al. 2001a,b; Reed-Smith 2001).

Although frequently solitary, except for female with pups, the North American river otter shows a great deal of social plasticity (particularly males), often forming groups of 8-15 or more animals in environments offering abundant resources (Blundell et al. 2002a,b). All male groups of up to 15 individuals have been maintained successfully in zoos and aquariums (Ben-David et al. 2000). In the wild, males do not participate in pup rearing; in zoos and aquariums males can be reintroduced to the family group once the pups are swimming well and in general interact and play with the pups. Both sexes occupy linear shaped home ranges due to their affinity for the land/water interface. Activity centers (e.g., latrines), located within home ranges, are important for both sexes. During a latrine activity study in Pennsylvania, Stevens & Serfass (2008) documented that visiting otters spent 72.7% of their time there smelling and investigating, 10.9% marking, 10.6% traveling, 4.6% rolling and rubbing, and <1% either sliding or autogrooming when visiting alone. When visiting as groups of two or more, they spent 43.6% of the time smelling and investigating, 30.7% wrestling, 7.4% traveling, 5.92% marking, 5.5% engaged in miscellaneous play behavior, 2.76% autogrooming, 2.3% sliding, 1.5% rolling and rubbing, and 0.3% allogrooming. They also found a seasonal difference in latrine visitation patterns with a visit peak occurring in spring (winter according to the Gregorian calendar) which corresponds to just before, and during breeding season (Stevens & Serfass 2008). The least number of latrine visits were recorded in summer which may reflect the tendency of females with pups defecating in the water which has been recorded in zoos and aquariums. N.A. river otters primarily feed on fish and crayfish.

This semi-aquatic species is found throughout the United States and Canada in a wide range of fresh water and marine ecosystems. Listed on CITES as Appendix II (www.cites.org).

Spotted-necked otter (*Lutra maculicollis*): This species was known as *Hydrictis maculicollis*. The spotted-necked otter is smaller than the often-sympatric African clawless otter. Their size ranges from roughly 4-6.5kg (8.8-14.3lbs) (Chanin 1985) to a maximum of ~9kg (19.8lbs) (Harris 1968), with a total length of 0.95-1.07m (3.1-3.5ft) (Chanin 1985). The spotted-necked otter has been reported to live in family groups, possibly groups of more than one family (Procter 1963), and single sex groups (IUCN 1992). During a recent study (Reed-Smith in prep.) male/female family groups were not observed. Instead the most frequent groupings observed were female(s) with young, all male groups, adolescent groups (sex undetermined), single animals, and breeding pairs. They generally forage for fish within 10m of the shore (Kruuk & Goudswaard 1990), but do forage further from shore (Kruuk & Goudswaard 1990; J.Reed-Smith, personal observation). The spotted-necked diet consists primarily of fish, supplemented at times by fresh-water crab and crayfish.

This species is found in all countries south of the Sahara, from Senegal to Ethiopia to the African Province of South Africa. It is absent only from desert areas as it lives primarily around the larger lakes (Foster-Turley 1990; IUCN/SSC 1992). Listed on CITES as Appendix II (www.cites.org) and as a species of Least Concern by IUCN/OSG (2009). However data is scarce and populations are known to be decreasing in portions of their range.

Terminology Appropriate to Taxa: Adult males are known as “dogs” but typically referred to as males; females are typically referred to as females. Young otters are referred to as pups (North America), cubs (Europe, Asia, Latin America), and in some publications as kits. The words “spraint” or “scat” are used to refer to feces and “latrine, sprainting zone, or camp site” denotes areas where otters deposit scat and urine.

Regulating Agencies: Endangered species regulations of the U.S. Fish and Wildlife Service (www.fws.gov/endangered) and Environment Canada (www.on.ec.gc.ca/wildlife/enforcement/cites-e.html) should be observed when importing or exporting the giant otter. All relevant state and federal regulatory agencies should be contacted for any current changes in permitting requirements. When shipping animals internationally, CITES permits may be required. IATA regulations should be consulted for current shipping container requirements.

Chapter 1. Ambient Environment

1.1 Temperature and Humidity

> **AZA Accreditation Standard**
>
> **(1.5.7)** The animal collection must be protected from weather detrimental to their health.

Air Temperature: All otter species should be provided with shelter from the sun and inclement weather. Indoor exhibits should offer an ambient temperature gradient within the exhibit providing otters the opportunity to select for their comfort.

<u>*A. cinereus*</u>: The ideal air temperature is between 22.2-24.4°C (72-76˚F). If Asian small-clawed otters have access to radiant heat, or a heated indoor facility, they can handle temperatures down to 10-15°C (50°F). The recommended water temperature is between 18.3-29.4°C (65-85˚F). It is recommended that warm water (29.4°C/85˚F - Lombardi 2004) be provided for swimming, since these tropical animals will spend more time in the water if it is warm (Petrini 1998), and this may have beneficial health effects.

<u>*A. capensis* and *L. canadensis*</u>: These species can tolerate a wide temperature range as long as they are offered protection from the sun and inclement weather in outdoor exhibits. Indoor exhibits should offer a thermal gradient allowing animals the selection of a comfortable temperature (10-24°C or 50-75˚F) (Reed-Smith 2004a). A temperature below 21-24°C (70-75˚F) is recommended for indoor holding/night facilities (Wallach & Boever 1983). Animals should always be provided with shelter from the sun in outdoor exhibits.

<u>*L. maculicollis*</u>: This species has been housed successfully at floor temperatures ranging from 14.4-25.5°C (58-78°F) (Schollhamer 1987). Their temperature tolerance is likely to be similar to that of *A. cinereus* and *A. capensis*; however, at this time there is insufficient information and experience to make informed recommendations. Animals should be monitored for signs of overheating and hypothermia at temperatures above 25.5°C (78°F) and below 14.4°C (58°F), respectively.

<u>*P. brasiliensis*</u>: In temperate climates, Wünnemann (1995a) recommends a minimum of 18°C (64.4°F) air temperature for dens and indoor enclosures. The suggested temperature range is 18-20°C (64.4-68°F) (Hagenbeck & Wünnemann 1992). For specific temperature recommendations for young pups, see Chapter 7. Indoor enclosures should be equipped with fans, cooling, and/or ventilation systems to prevent over-heating and provide fresh air exchange in all climates; in temperate climates a heating system is required (Sykes-Gatz 2005).

This species should be provided with the choice to use an outdoor enclosure year-around, even in temperate climates, as they are quite adaptable to colder outdoor temperatures (young pups are an exception to this, see Chapter 7), as long as they have access to heated indoor enclosures in addition to their dens (Wünnemann 1995a). Adults will not carry out their normal daily terrestrial activities in air temperatures at approximately 10°C (50°F) or below (regardless of whether the outdoor pool water is heated), but will spend limited time in these temperatures, and seem to avoid temperatures that are too cold for them. Giant otters should have access to a heated indoor enclosure at all times when seasonal daytime air temperatures regularly fall below 15°C (59°F) (Sykes-Gatz 2005; V.Gatz, personal communication). The following recommendations are provided for giant otters:

- Exposure to air temperatures at or below ~7°C (20°F) should be restricted, and otters should be carefully monitored if given access to temperatures near this range.
- Newly imported animals from tropical climates, juveniles, and sub-adults should be acclimated slowly over a period of 6-12 months to these colder temperatures.
- Shelter from the wind, rain, heat, cold, and constant direct sun should be provided in all climates (Sykes-Gatz 2005).

Water Temperature: More detailed research is required into optimal water temperature levels for the tropical otter species; however, at this time the AZA Otter SSP recommends the following temperature guidelines:

<u>*A. cinereus*</u>: The water temperature for *A. cinereus* should be maintained between 18.3-29.4°C (65-85°F), preferably at the warmer end of this scale (Petrini 1998).

A. capensis, L. canadensis: The water temperature for *A. capensis* and *L. canadensis* does not appear to be critical.

L. maculicollis: Water temperature in successful *L. maculicollis* exhibits has ranged from 8.9-15.6°C (48-60°F). Temperatures in the 15.6-21.1°C (60-70°F) range may encourage this species to spend more time in the water, however, this is has not been objectively demonstrated at this time.

P. brasiliensis: Further study into optimal pool temperatures and water temperature exposure recommendations for *P. brasiliensis* is required. Sykes-Gatz (2005) recommends this species should not be allowed to swim in unheated water when air temperatures are below 5°C (41°F). As a precaution, outdoor pools should be emptied when temperatures approach this range. Sufficient indoor swim areas are needed when seasonal daytime air temperature regularly falls below 15°C (59°F), regardless of whether outdoor water is heated. This is particularly true for family groups rearing pups that may be held indoors for 4-5 months during cold temperatures. Heating of indoor housing pools is not necessary if the ambient air temperature is maintained at recommended levels. See Appendix G for information on pool design recommendations for giant otters.

Humidity: Since otters always should have water features available to them, humidity does not seem to be a factor in their environment unless it is excessive. Excessive humidity and an inability to adequately dry off create problems for all otter species, and these conditions should be avoided. The relative humidity of indoor exhibits should range between 30-70%. Nest boxes and den sites should be provided with good ventilation and placed in locations that are not chronically humid. The AZA Otter SSP recommends the provision of sufficient dry land (see Chapter 2, section 2.1), natural substrates, and bedding material (see Chapter 2, section 2.1) to aid the otters in proper coat maintenance, and allow for adequate drying of their pelts and feet.

AZA institutions with exhibits which rely on climate control must have critical life-support systems for the animal collection and emergency backup systems available, while all mechanical equipment should be included in a documented preventative maintenance program. Special equipment should be maintained under a maintenance agreement or records should indicate that staff members are trained to conduct specified maintenance (10.2.1). Records should include daily activities required for maintenance, such as back-washing of filtration systems, ozone checks, pH, chlorine, and coliform levels as well as dates of periodic maintenance activities.

AZA Accreditation Standard

(10.2.1) Critical life-support systems for the animal collection, including but not limited to plumbing, heating, cooling, aeration, and filtration, must be equipped with a warning mechanism, and emergency backup systems must be available. All mechanical equipment should be under a preventative maintenance program as evidenced through a record-keeping system. Special equipment should be maintained under a maintenance agreement, or a training record should show that staff members are trained for specified maintenance of special equipment.

1.2 Light

Careful consideration should be given to the spectral, intensity, and duration of light needs for all otters in the care of AZA-accredited zoos and aquariums.

If otters are held in holding areas for any length of time, some natural light is recommended. Fluorescent, metal halide and mercury vapor, as well as natural light have all been used in exhibit areas. The AZA Small Carnivore TAG is unaware of any hard data on the impact of light intensity on otter health or reproduction; this should be investigated in the future. However, it is strongly suspected that otters held indoors should be provided a seasonally appropriate light cycle to promote breeding and general animal health (Bateman et al. 2009). There are no available data on possible deleterious effects of less than full spectrum light on a long-term basis.

A. cinereus: If housed indoors, these species should be kept on a 12-hour light cycle (Wilson, Tropea & Calle, unpublished data).

L. canadensis, A. capensis, L. maculicollis: The light cycle for indoor exhibits/holding should be set to mimic the natural photoperiod for the species range of origin in Equatorial Africa, for *A. capensis* and *L. maculicollis*, or the local photoperiod for N.A institutions housing *L. canadensis* (Reed-Smith 2001, Bateman et al. 2009).

P. brasiliensis: All indoor enclosure areas, except for the nest boxes, should be kept on a 12-hour light cycle to mimic the natural habitat conditions of giant otters. If possible, full-spectrum lighting should be provided. Giant otters are diurnal, and only the nest boxes should remain dark. If necessary, infrared lighting may be used when video cameras do not have infrared capabilities.

1.3 Water and Air Quality

AZA-accredited institutions must have a regular program of monitoring water quality for collections of aquatic animals and a written record must document long-term water quality results and chemical additions (1.5.9). Monitoring selected water quality parameters provides confirmation of the correct operation of filtration and disinfection of the water supply available for the collection. Additionally, high quality water enhances animal health programs instituted for aquatic collections.

AZA Accreditation Standard

(1.5.9) The institution must have a regular program of monitoring water quality for collections of fish, pinnipeds, cetaceans, and other aquatic animals. A written record must be maintained to document long-term water quality results and chemical additions.

Air Changes: The number of air changes per hour of non-re-circulated air needed to control odors and maintain a healthy condition for the animals and public will vary according to the number of animals in the enclosure and the size/volume of the enclosure. The initial design should be for the maximum number of animals that could be housed in that particular enclosure. Standardized rates of change for various human-occupied enclosures suggest that pet shops require a rate of air exchange of non-recirculated air equal to 1 cubic foot of air/minute/ft^2 of floor space, in order to keep odors down to a level acceptable by the public (Anon. 1981). Pupping dens may well need higher rates of air exchange in order to maintain air quality and/or low humidity. It should be noted, however, that no work has been done specifically targeting air change rates for otter exhibits or dens.

As a general rule, indoor exhibits should have a negative air pressure of 5-8 air changes per hour of non-recirculated air. Glass barriers and separate ventilation systems between indoor exhibits and visitor areas (Moore 1997) will help reduce the potential of disease transmission from the public as well as complaints due to odor.

Water Quality: See Appendix N for information on designing a life support system for otter exhibits, glossary of terms, and additional details on water treatment provided by J. Sabalones (2009) life-support systems advisor to the IUCN Otter Specialist Group’s Otters in Captivity Task Force.

Otters are semi-aquatic mammals, using bodies of water for foraging, transportation corridors, mating (typically), cleaning, and “play-type” behavior. It is recommended to monitor nutrients and perform pool water changes as needed. There are no standards yet established for pools provided to semi-aquatic otters, however, it is suggested that coliform levels be maintained at 400 per ml water or lower, which is the standard set for seal rehabilitation pools. A level of 100 per ml is considered safe for humans. All chemical additives should be monitored daily and recorded.

It is recommended that filtration be used in closed otter pools. Sand filters, pool pumps, charcoal filters, and ozone pressure sand filters have all been used effectively. Ultraviolet sterilization has proven helpful in inhibiting algae build-up, particularly when combined with regular cleaning of pool-sides. Drain outlets, filters, and skimmers should be covered or designed to prevent furnishings from blocking them, or otters from becoming stuck in them. Daily use of long-handled skimmers will help remove floating debris and keep skimmers open and flowing. Natural flow-through systems also work well in otter exhibits, as long as the water is determined to be clean and free of heavy pollutants. In general, otters should be kept in fresh water systems; however, Ben-David et al. (2000, 2001a,b) successfully kept a group of 15 males in sea water that was changed daily. In this case, fresh water was provided in tubs for the animals to bathe in. Regardless of water treatment method used, an additional source of fresh, potable drinking water should be available at all times.

Accreditation Standards and Related Policies

The provision of fresh potable water is a requirement of USDA Animal Welfare Regulations (AWR 2005) as stated: "If potable water is not accessible to the animals at all times, it must be provided as often as necessary for the health and comfort of the animal. Frequency of watering shall consider age, species, condition, size, and type of the animal. All water receptacles shall be kept clean and sanitary" (AWR 2005). Considering the needs of otters, the AZA Small Carnivore TAG states that otters should be given fresh water daily if their pools are not filtered or dumped and filled on a daily basis. AZA Accreditation Standards require that institutions abide by relevant federal laws and regulations: "The institution must comply with all relevant local, state, and federal wildlife laws and regulations. It is understood that, in some cases, AZA accreditation standards are more stringent than existing laws and regulations. In these cases the AZA standard must be met" (AZA 2008).

Water quality should be maintained at a level sufficient to control bacterial counts and organic load, and to allow clear underwater visibility of animals for health inspections. Clarity and color maintained to provide a perceived color of clear and/or blue water is preferred by most facilities. This water clarity is an aesthetic requirement only, as long as the water quality is maintained, and the presence of floating algae or other material is not harmful to the otters. Otters can be messy eaters and will track a lot of particulate debris into their pools. All food remains should be removed from pools daily to prevent consumption of spoiled items. The filtration system needs an effective means of skimming (from top to bottom) particulate matter. Turn over rate using rapid sand filtration is suggested to be once/hour; meaning that the total exhibit water volume should be turned over at least one time per hour when using rapid sand filtration. When using rapid sand filtration couple with Ozone; the turnover rate can be extended to once every 1.5 hours.

Chemical treatment such as ozone applied to foam fractionation is recommended for marine systems. Ozone applied through a contact chamber in conjunction with a low dosage of chlorine is an effective treatment for freshwater systems. A large surface area biological filter bed should be incorporated if possible. This will allow a natural nutrient removal system to establish itself, which will provide system stability. It also will help reduce organic loading as well as reduce colonization of undesirable bacteria species. The tank effluent should be pre-filtered before it is sent to the sand filters and foam fractionators.

At least bi-weekly water quality tests are recommended for bacterial counts and daily tests of chemical additive levels. Records should be maintained and available for APHIS inspection and reference if problems arise.

<u>Coliform bacteria</u>: Coliform bacterial counts are used to monitor filtration system efficiency and keep track of potentially harmful bacteria. Coliform counts should be done at least every other week and more often if there are multiple animals using the pool (a policy regarding coliform testing should be set by the institution). Often a MPN (Most Probable Number) per 100ml is given as an acceptable limit. However, a more accurate measure is the total or fecal coliform count (NOAA 2006). There are no standards yet established for fresh-water otter pools. At this time, it is suggested that coliform levels be maintained at or lower than levels established for rescued pinnipeds by NOAA. These are:

- Total coliform counts should not exceed 500 per 100ml water, a MPN of 1000 coliform bacteria per 100ml water.
- Fecal coliform count should not exceed 400 per ml.

If animal caretakers are routinely exposed to pool water, an institution may establish a higher standard of 100 per ml, which is the level considered safe for humans; this should be based on institutional policy.

<u>Chlorine</u>: Many municipalities add chlorine to their water, and readings from tap water of 1ppm or higher are possible. While otters generally show no adverse effects from these levels, it is not known what the overall impact is to their health and the water repellency of their coats. For this reason, the AZA Otter SSP recommends that otters should not be exposed to chlorine levels higher than 0.5ppm for prolonged periods, and ideally chlorine should be kept at a non-detectable level. The addition of sodium thiosulfate will neutralize any residual chlorine (see below and Appendix M).

Algae control: Algae control is a continuing problem in otter pools, particularly those exposed to significant sunlight. There are several techniques that have been used with varying success (see Appendix M).

- Liquid copper sulfate: Liquid copper sulfate can be added directly to the pool water without harm to the animals. While this does not get rid of algae, it will inhibit algal growth.
- UV sterilization: Ultra violet sterilization has proven helpful in inhibiting algae build-up, particularly when combined with regular cleaning of pool sides.
- Chlorine: Chlorine can be used, if necessary, when the otters are not present. In this case, sodium thiosulfate can be added and run through the system for an hour before the otters are allowed access again. A 5% concentration of sodium thiosulfate added has been successful. To obtain the amount needed for a particular system, multiply 0.53 by the volume of the pool in gallons; this provides the amount of sodium thiosulfate required in millimeters (C. Harshaw, personal communication).
- Barley straw: Hanging a bag of barley straw in the water stream assists in filtering out algae. This should be hung where the otters cannot get to it (e.g., at the top of a waterfall, etc.). Reports of its efficacy vary and may be dependent on design and location of pool.

All of these techniques should be accompanied with routine scrubbing of the pool sides to inhibit algae growth, and discussed with life-support professionals.

Drinking Water: Clean drinking water should be available at all times. Drinking water should be provided in bowls small enough that the otters do not swim in them, or via lixits or similar devices. Animals should be introduced to the use of lixits (or other drinking fixtures) and monitored by staff until they are certain they are proficient in their use.

Further research is needed into the impact, if any, of pH on otters. The deleterious effects of chlorine on otters specifically is anecdotal, e.g. stripping of water repellency; presumed, e.g. potential carcinogenic by-products of chlorine break-down; and, unknown, e.g. impact on overall health. While research is desirable it is not recommended on these wildlife species and instead caution should be exercised when using chlorine in otter pools.

1.4 Sound and Vibration

Consideration should be given to controlling sounds and vibrations that can be heard by otters in the care of AZA-accredited zoos and aquariums. While there is no evidence that low level background noise is disruptive to otters, loud noises can be frightening to them, and high-pitched, long-term noise should be avoided. Parturient females should not be subjected to loud or unusual noises; this is particularly true with primiparous females and giant otters. Females about to give birth and with very young pups should not be subjected to close proximity of the public, loud or unfamiliar voices, construction noise, sudden loud noises such as sirens, unfamiliar ambient noises, or vibrations to which they are not already accustomed. For the giant otter, both minor and loud sounds that they have been accustomed to before parturition, including familiar noises from zoo staff during daily caretaking routines, can also cause significant stress after parturition. All precautions should be taken to eliminate these during the last two weeks prior to birth (time margin is estimated based on difficulty of predicting parturition dates in these species) and during roughly the first month after parturition for primiparous females and for giant otters until pups emerge from the den or until roughly 75 to 120 days post parturition when giant otters have demonstrated more noise tolerance in the past.

Otters' hearing is considered to be good but nothing is known definitively about their hearing acuity or frequency ranges heard. Both of these are areas needing further research.

Chapter 2. Habitat Design and Containment

2.1 Space and Complexity

Careful consideration should be given to exhibit design so that all areas meet the physical, social, behavioral and psychological needs of the species. Animals should be displayed, whenever possible, in exhibits replicating their wild habitat and in numbers sufficient to meet their social and behavioral needs (1.5.2).

> **AZA Accreditation Standard**
>
> **(1.5.2)** Animals should be displayed, whenever possible, in exhibits replicating their wild habitat and in numbers sufficient to meet their social and behavioral needs. Display of single specimens should be avoided unless biologically correct for the species involved.

Important factors to consider when creating successful otter exhibits include: exhibit land area size, design and complexity; pool size--design and complexity (including shoreline length and complexity); substrate materials and depths; water quality; climbing surfaces; digging areas; and denning sites (location and construction).

Land/Water Ratio: Suggested optimal land/water ratios will change as an exhibit size increases or decreases. The ratios offered here are for the recommended minimum exhibit size. Smaller exhibits will require a higher land area proportion within the ratio. Larger exhibits and *P. brasiliensis* exhibits 600m² (6,458ft²) in size or more, may have a somewhat lower land proportion and still be successful. .

L. canadensis, L. maculicollis, and A. capensis: The recommended land/water ratio for *L. canadensis*, *L. maculicollis*, and *A. capensis* is 3:1 to 4:1 (3:1 is the absolute minimum land area proportion and considered adequate only if the exhibit is large, vertically complex, and offers hard-surface features within the pool, such as logs, islands, etc) (Duplaix-Hall 1975; Reed-Smith 2001 2004a).

A. cinereus: For *A. cinereus*, the recommended ratio is 5:1 or 6:1 (Duplaix-Hall 1975; Lombardi et al. 1998).

P. brasiliensis: *P. brasiliensis* indoor and outdoor enclosures between 240m² to 600m² (2,583ft² to 6,458ft²) in size should be provided with an absolute minimum of 60% land area. (Sykes-Gatz 2005; Duplaix-Hall 1972 & 1975). As exhibits and any living areas that may include water features decrease in size below 240m² (2,583ft²), a greater land proportion within the ratio is recommended. E.g. a 150m² (1,615ft²) enclosure should provide an absolute minimum of 69% land area and a 75m² (807ft²) enclosure needs at least 76.5% land area. Sykes-Gatz (2005) (also see Appendix G) offers a simple formula that should be used for guiding land-water calculations.

Exhibit Complexity – Terrestrial: Otters are land mammals that swim; they are semi-aquatic or amphibious, not aquatic animals. Behaviorally healthy otters kept in appropriate enclosure conditions spend more of their daytime hours on land than in the water. As instinctively avid diggers (*P. brasiliensis* and *A. capensis* in particular) and groomers (all species), otters dig and groom extensively in soft loose natural substrates. They groom when wet or dry by rubbing, scratching, and digging into soft loose dry substrates, often covering their body fur with the freed particles. (See photo-N.A. river otter, Jennifer Potter, Calgary Zoo) These behaviors are among the most favored and frequently performed terrestrial activities in zoos and aquariums, and otters will use the entire expanse of their land area to carry them out. Together with foraging, exercising, and frequent play bouts on land, these terrestrial behaviors constitute a significant proportion of otters' natural, daily goal-oriented activities (*P. brasiliensis* do not forage on land). These behaviors are considered essential to maintaining the otters' physical and behavioral health, as well as to the promotion of successful pup-rearing practices (Reed-Smith 2001; Sykes-Gatz 2005). Additionally, the ability to carry out all of these behaviors is considered important for an otter's healthy adjustment to new or unusual situations. Digging and grooming are among the most important activities required by *P. brasiliensis* in particular to prevent or reduce stress and to maintain health of the animals (Sykes-Gatz 2005).

The AZA Otter SSP recommends that exhibits should be constructed of a variety of natural substrates to accommodate these activities. If artificial surfaces like concrete are used, these should be kept to a minimum. Digging pits and grooming areas with soft, loose substrates should always be included in otter exhibits, both indoor and outdoor enclosures. (*P. brasiliensis* although require additional substrate conditions to keep sufficiently dry and healthy; see Appendix G and below in 2.1.) Adequate land area and substrates on which otters can groom are considered key to the care of *ex-situ* populations of otters. In order to maintain healthy thermal properties of their coats, otters have to frequently groom their fur, replacing the air layer trapped within the under-fur (Dunstone 1998, Weisel et al. 2005).

A variety of live plants can be used in exhibits, as well as log piles, large tree stumps or root systems, hollow logs, hills, etc., all of which can provide visual complexity to the exhibit and offer otters excellent foraging, playing, and shelter opportunities. All of these features can be placed to allow for visitor viewing. However, accommodation giving very shy animals the ability to hide should be made.

As with any species, otter exhibits should be "redecorated" periodically. However, it is advisable that preferred denning or hiding spots not all be changed simultaneously. All exhibits should be constructed with a means of accomplishing re-fitting of exhibit furniture, including the introduction of large deadfall. Self-dug dens, particularly those of *P. brasiliensis* should be allowed to remain as long as they are deemed safe.

The minimum exhibit size (including land and water surface area) suggested for otters is as follows:

- *A. capensis* and *L. canadensis*: 150m^2 (1615ft^2) for two animals. An extra 25m^2 (269ft^2) of useable land surface and 10m^2 (108ft^2) water surface should be provided for each additional animal (Duplaix-Hall 1975; Reed-Smith 2004a).
- *A. cinereus*: 60m^2 (646ft^2) for 2-4 animals (Duplaix-Hall 1975); 93m^2 (1,000ft^2) for more than 4 animals.
- *L. maculicollis*: 100m^2 (1076.5ft^2) land and water surface for two animals. An extra 20m^2 (215.3ft^2) of useable land and 5m^2 (54ft^2) water surface should be provided for each additional animal.
- *P. brasiliensis*: In North America, the minimum enclosure size (land and water surface) that has housed a family of giant otters is 121m^2 (1300ft^2). Duplaix-Hall (1972, 1975) recommends a minimum size of 240m^2 (2,584ft^2) for a pair. Sykes-Gatz (2005) also cites 240m^2 (2,584ft^2) for a pair with additional space provided for offspring. A recommended additional size for family groups is not offered but stress is placed in the quality of the enclosure space and land/water ratio appropriate for the exhibit size. Please refer to Sykes-Gatz (2005) for ratio formulas and more information.

Recommended minimums are based on the species natural history, observations of *in-situ* populations of otters, and authors experience with otters in *ex-situ*.

Exhibit Complexity – Aquatic: The water portion of an otter exhibit should include areas of varying depths, and some portion of shoreline that allows for easy access to and from the shore for both old and young animals. Shorelines also should be complex and designed to allow for the periodic change of features (e.g., logs, rock piles, pebble pockets, etc.). For more specific suggestions for *P. brasiliensis* enclosure shoreline design, see Sykes-Gatz (2005). Streambeds or shallow wading pools with rocky bottoms offer good enrichment and foraging opportunities, but can cause footpad abrasions if otters are forced to walk on them too often without access to natural substrates for drying their feet. To aid in minimizing the transference of debris into pool water, the shoreline can be constructed of raised flat/sloping rocks or logs positioned to hold natural substrates; Sykes-Gatz (2005) recommends a ~10cm (4-inch) curb. Furnishings should allow otters, especially pups, females carrying pups, and geriatric otters easy and safe ingress and egress to/from pools. Pools should be designed with several skimmers that can capture large particle debris before it reaches the filtration system. All pool openings such as skimmers, drains, filters, etc. to which the otters have access should be securely covered with sturdy wire fencing to prevent curious otters from getting their heads/feet stuck. Ample, extensive dry land areas should be provided to allow all otter species to dry off completely.

Exhibit pools should have varying depths, offering opportunities for animals to forage in the shallow water and swim/dive in deeper water. Shorelines should be complex and curving, as opposed to straight lines and uninterrupted. Additionally, the shore should be periodically interrupted with shade structures (e.g., bushes, trees, etc.), and climbing or laying-out structures, such as logs, boulders, log jams, etc. Otters should not be forced to swim continuously next to public viewing windows as this frequently leads to stereotypic activities such as flip-swimming, tail-sucking, and back-flips (Reed-Smith 2004b). Instead, exhibit pools should offer swimming alternatives that allow the animals to access deep and shallow areas that are not next to the public viewing areas. A narrow pool design has not caused these negative effects on *P. brasiliensis*. Instead insufficient land vs. water area and/or inappropriate substrate conditions have been the primary cause of behavioral problems including; stereotypic swimming, swimming excessively and aimlessly, and parents or older siblings overexposing pups to pools or mistreating them there (Sykes-Gatz 2005). See Appendix M for additional information regarding designing and maintaining pools in otter enclosures.

P. brasiliensis: *P. brasiliensis* pools should have a deep area of at least 100cm (3.28ft), as well as shallow areas; the otters make frequent use of waters of differing depths. Depths of 150-200cm (5-6.5ft) and deeper are highly recommended allowing a wider range of swimming and diving behaviors.

Exhibit Design and Species-appropriate Behaviors: In addition to a pool, all otter enclosures should be enhanced with a variety of furnishings. The quality of space for these animals is just as important as exhibit size. Logs, trees, tree roots, stumps, grasses, boulders, dens, caves, climbing structures, bushes, deadfall (positioned so animals cannot use them to climb out of the enclosure), waterfalls, floating log piles, rafts, islands, varied exhibit levels, and a variety of substrates are all important elements of a complex and successful otter exhibit (for all species). On-exhibit sleeping and hiding places should be provided; these sites should be of varying sizes to allow the group to sleep together or to allow for individual seclusion. Animals should be allowed to dig, roll, climb, and slide within their exhibit. Enclosure designers should take all of these activities into consideration when designing the land/water interface, public viewing, substrates, and pool filtration systems. Pools should be designed so that the animals are not always forced to swim in close proximity to the public (this requirement does not appear to be as crucial for the giant otter). Public viewing should be provided from various angles while maintaining one side without public access as a secure zone for the animals. Sykes-Gatz (2005) and Hancocks (1980) are two of the many resources available with information on naturalizing older exhibits.

All pool shorelines should be provided with lounging logs, shaded rest areas, and sandy banks to be used as latrine sites. Large flat/sloping rocks and logs can be used along the shoreline to hold back the substrate as well as provide good sunning areas.

A variety of substrates should be incorporated into otter exhibits. These substrates include: grass, mulch, sand, clay, soil, rocks, boulders, pebbles, leaves, bark, concrete, and gunite (the latter two are not recommended and should be limited to small areas, or should be covered with soft pebble-free sand or tree bark mulch when their use is unavoidable). The substrate recommendations for *P. brasiliensis* are slightly different to other otter species, and species-specific recommendations are provided below. Exhibits with artificial substrates should offer areas of grass, dirt, sand, pebbles, etc. for exploration and adequate grooming. Hard-pack soils, abrasive sands, and sharp rocks should not be used in otter exhibits. Recent research into the structure of sea otter (*E. lutris*) and river otter (*L. canadensis*) hair structure shows guard hairs can suffer damage (Weisel et al. 2005). While unclear at this time, it is possible that extensive damage to guard hairs can impact the insulative ability of the otter's coat. Problems with chronically wet surface areas or overexposure to hard surfaces should be addressed immediately to prevent injuries to the animals (e.g., foot pad abrasions) or health issues (e.g., fungal infections, pneumonia) from developing.

All exhibits should offer bedding material; products used successfully include: grasses, leaves, hay, straw, wood wool, sedges, pine needles, towels, burlap bags, indoor/outdoor carpeting, natural fiber mats, and wood shavings (Reed-Smith 2001). Bedding material recommendations for *P. brasiliensis* are different than other otter species and these species-specific recommendations are provided below. Some facilities have successfully used fleece and blankets (Ben-David et al. 2000, 2001a and b; J. Reed-Smith, personal experience). However, as with all bedding, these should be monitored to ensure the otters are not chewing on or eating them. If animals are chewing on these items, they should be removed immediately. Some wood shavings (from conifers) contain residues that can strip the water proofing from the coat of semi-aquatic species, and/or may cause sneezing. Cedar contains aromatic phenols that are irritating to the skin and respiratory system. Several studies indicated that close, chronic contact with cedar shavings contributed to infant mortality (Burkhart & Robinson 1978), respiratory disease (Ayars et al. 1989), and liver damage (Vesell 1967) in rodents. The impact of these products on otters is unknown; if used, caution should be exercised.

Some facilities use paper products such as shredded paper, cardboard boxes, paper bags, and cardboard rolls. These products should be monitored carefully to ensure the animals are not ingesting them or taking them in the water where they could become plastered over an animal's mouth and nose, or become impacted in their teeth. In most cases the AZA Otter SSP advises against using these products with otters.

Indoor/outdoor carpeting and natural fiber mats also have been used for the animals to roll and groom themselves on in concrete holding areas. All materials used for bedding should be monitored in case an animal consumes them excessively, or in the case of towels, etc., shreds or eats them. 'Wood wool' sticks to fish or other moist foods, and so should not be used near feeding areas to prevent its ingestion. When it is used in nest boxes, caution should be exercised if any females become pregnant as pups can become entangled in it.

L. canadensis: Typically *L. canadensis* shed their under-fur between May and August (This "…under-fur produces a dense, matted, felt-like layer, which forms an efficient insulating layer by trapping air next to the skin…'[Dunstone 1998]), and replace their guard hair between August and November (northern latitudes, there may be some variation in timing at southern latitudes) (Ben-David et al. 2000; J.Reed-Smith, personal observation). Animals' health requires ample grooming opportunities and surfaces on which they can rub to prevent matting, and aid in this annual coat replacement process. Grooming and drying opportunities also are important for the maintenance of healthy foot condition, with damp or excessively humid conditions leading to footpad abrasions.

P. brasiliensis: Field (Duplaix 1980) and *ex-situ* population studies (Sykes-Gatz 2005) of *P. brasiliensis* have shown that grooming consumes a great deal of time, and plays a vital roll in group cohesion and home range identification. Duplaix (1980) states that this species spends fully half of its time on land with a great deal of this dedicated to grooming.

Sykes-Gatz (2005) stresses the importance of providing at least the minimum land area proportion within the recommended land/water ratio and the recommended substrate conditions as primary to maintaining sufficient dry land area, nestboxes, and bedding materials in indoor and outdoor giant otter enclosures. The following additional designs are secondary for the same purposes, but will not be effective if insufficient land vs. water area or inappropriate substrate conditions (esp. hard surfaces) exist. The smaller the land area provided giant otter and the more land area exposed to the water's edge, the harder it is to keep dry.These large otters carry significant amounts of water from their pools onto land during their frequent exits from the pool. Water areas should be bordered by at least 5m (16ft) of land area extending back from the shoreline of the pool for all exhibit sizes. For exhibits smaller than 240m² (2,583ft²) and up to 600m² (6,458ft²) Sykes-Gatz (2005) should be consulted for specific recommendations. Additionally, long water area contour lines, or land bordered by water on more than two sides, are not recommended for this species as this will result in the land becoming saturated as otters enter and exit the pool along the length of the shoreline. Dens and nest boxes should be located at least 3m (10ft) and preferably 5m (16ft), away from the water's edge. Also, plentiful land area should extend laterally from the den/nest box entrances to offer sufficient and conveniently located areas where otters can dry off before entering their dens/nest boxes. These design features will help eliminate continuously damp or wet land surfaces and nestbox conditions, which can lead to health problems (see Chapter 6).

Existing exhibits that do not meet these guidelines can be relatively easily and inexpensively modified. Sykes-Gatz (2005) and Hancocks (1980) provide useful information on naturalizing older exhibits. All enclosure surfaces can be naturalized with the methods and substrates described below. Sykes-Gatz (2005) stresses the importance of covering the enclosure land area and holding surface to a depth of 10-20cm (4-8") with soft natural substrates (soft pebble-free sand or tree bark mulch) that are deep and loose enough to allow adequate drainage and remain dry, so that otters can easily dig in it, and adequately groom themselves upon it. The use of other mulch products is not considered advisable with this species. Finely pieced mulch easily becomes saturated and does not dry sufficiently, creating unhealthy conditions for the otters. New mulch or sand should be added on top of the existing layer as needed to maintain no less than the minimum recommended depth, and/or to cover broken-down or compacted mulch. If this is not monitored closely the substrate may become too wet or damp, or too hard and unusable for digging or grooming by the otters. Additionally, indoor and outdoor enclosures should each have an area (20m² (215ft²) is recommended by Sykes-Gatz, 2005) with sand or mulch at least 40-60cm (16-24") deep, or soil hillsides, to allow for deep digging. See Appendix G and Sykes-Gatz (2005) for additional information on appropriate substrates and depths for giant otters.

Animal shift and keeper doorframes raised roughly 10cm (4") above the desirable substrate surface height will help prevent substrates from blocking all doors. A log border placed a little behind an existing keeper door or placed against the front and back of an animal shift doorframe, or a wooden lip for the door's track, serves the same purpose (Sykes-Gatz 2005).

Nest boxes can be provided with sand or mulch that is 10cm (4") in depth. When young pups are not present, other bedding materials such as wood wool, hay, straw, or leaves also may be provided, but these materials should be removed prior to birth of any litters and can be replaced with sand or tree bark mulch. Care should be exercised to ensure that all nest box substrates remain dry. Damp conditions may contribute to otter pup mortality. After parturition, otters may dig out all bedding but can still successfully raise pups. Pine needles, towels, burlap bags, indoor/outdoor carpeting, and natural fiber mats should not be offered to this species (Sykes-Gatz 2005).

The recommended substrates are inexpensive, effective, easy to maintain and acquire, and they remain sanitary with dry spot cleaning. When more land area is needed for an appropriate land/water ratio, a portion of an artificial (e.g., concrete) or natural pool can be divided with a waterproof barrier, or one or more of multiple pools can be emptied. These can be filled-in with the recommended substrates to create enough land.

This species in particular appears to like digging underneath large tree stumps with long root systems, logs, etc., and so these furnishings are advisable, especially in deep digging areas. A variety of vegetation and furnishings of this type, in addition to grasses, bamboo stands, and leaf piles, should be contoured into all exhibits allowing for exploration, visual barriers, and privacy when pups are born. Other effective furnishings include large hollow logs, large logs on land and lying into-over the water, and boulders (Sykes-Gatz 2005). In climates where otters can be outside year around, some zoos have successfully provided soil hillsides to allow otters to dig underground dens. The hillsides should be at least 2m (6.5ft) high, have an angle not more or less than 40-45°, and be located behind and near the water area shoreline (otters cannot dig deep enough in flat terrain or shallow substrates) (Sykes-Gatz 2005). Trees, large bushes, or tree stumps with long extended root systems may help prevent den cave-ins but this is always a danger. If there is any doubt about the safety of a den it should be refilled with substrate. See Chapter 2 for enclosure barrier considerations to prevent escape by digging. See Appendix G for additional information on giant otter exhibit furnishings, substrates, etc.

Sensory Barriers: Visual barriers are important to allow animals to avoid one another, when necessary. All individuals, particularly paired otters, will go through times when they exhibit a tendency to stay by themselves. Vegetation, exhibit topography, denning sites, and deadfall should be strategically placed to allow for this. While there is no evidence that low level background noise is disruptive to otters, loud noises can be frightening to them, and high-pitched, long-term noise should be avoided.

Otters can be odiferous; facilities with indoor exhibits may want to provide olfactory barriers for the comfort of the viewing public. If exhibiting more than one breeding otter group, or permanently separating animals from a group and housing them within the same institution, it may be very important to have visual, olfactory, and audio separation to avoid intra-specific aggression or abnormally elevated levels of stress and/or frustration. In these cases, it is advisable to plan for this in advance rather than trying to accommodate it if a problem arises.

L. canadensis: Parturient females generally become very aggressive towards the male several days before giving birth and while the pups are quite young. Pairs housed in large naturalistic exhibits can be maintained together if sufficient visual barriers are provided to allow the male to remain out of the female's line of sight. In all other cases, it is important that the pair is separated. The pair can be left at the exhibit if one animal is held in the holding dens/off-exhibit area while the other is on exhibit. In these instances, the animals should not have to pass in view of one another to shift into alternate areas for cleaning or feeding.

P. brasiliensis: Visual and acoustic isolation from human disturbances (staff and visitors) is necessary during parturition and early pup-rearing. All human sounds and disturbances should be minimized to ensure successful pup rearing, as this species is highly sensitive to human interference. See Chapter 7 for further information.

In the rare cases when bonded pairs or other group members have to be separated, they should be held in facilities distant enough to prevent visual, acoustic, or olfactory communication. *P. brasiliensis* is highly vocal and their calls carry great distances.

Enclosure Cleaning: Otters are scent-oriented animals; therefore, their entire exhibit or holding area should not be cleaned at the same time. Enclosures should be raked and spot cleaned daily, with appropriate disinfecting as necessary. Indoor or hard surface floors should be cleaned with detergent daily. Due to their natural scent marking behavior, exhibit furniture should not be cleaned as frequently. It may actually be stressful to some otters if their territory is totally cleared of their markings. All detergents should be thoroughly rinsed, as any residue left on the floor, mats, or furnishings can strip their coats of its natural oils (Schollhamer 1987).

Food and water containers should be cleaned and disinfected daily. All food remains should be removed before it can become spoiled; in some climates this may require removal more than once a day. A safe and effective control program for insects, ectoparasites, and bird and mammal pests also should be maintained.

The same careful consideration regarding exhibit size and complexity and its relationship to the otter's overall well-being must be given to the design and size all enclosures, including those used in exhibits, holding areas, hospital, and quarantine/isolation (10.3.3).

A holding area connected to the exhibit is recommended; ideally this should include a pool with clean water available at all times, proper lighting, appropriate lighting cycle, a sleeping or den box, enough floor space for grooming and drying areas, an outdoor play area (if extended stays are routine), and at least one nest box that is heavily bedded to allow excess moisture to be removed from the animals' coats (Lombardi et al. 1998; Reed-Smith 2001). Each holding den should be large enough to at a minimum allow the animal to turn around (when used only for holding during cleaning) but at least 122cm (4 ft.) by 122cm (4 ft.) if an otter is to be held over-night. Best practices would include larger night dens than this minimum. Holding pens should have non-climbable sides, access one to another (allowing animals to have more room during extended stays), and multiple ingress and egress to the exhibit. Maternity dens should be isolated so exhibit mates do not have to pass in front of this den to enter or exit the exhibit. If chain-link barriers are present, the sides should be covered with lexan or similar material to prevent animals from climbing too high and falling.

Quarantine facilities should offer the maximum space available. They should be furnished with natural items to include logs, stumps, and outdoor carpeting that can be periodically disinfected/changed as well as sand/mulch/straw/soil boxes that can be changed when they become soiled or saturated.

> **AZA Accreditation Standard**
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> **(10.3.3)** All animal enclosures (exhibits, holding areas, hospital, and quarantine/isolation) must be of a size and complexity sufficient to provide for the animal's physical, social, and psychological well-being; and exhibit enclosures must include provisions for the behavioral enrichment of the animals.

2.2 Safety and Containment

Otters are not appropriate for free-ranging environments. Animal exhibits and holding areas in all AZA-accredited institutions must be secured to prevent unintentional animal egress (11.3.1). Exhibit design should be considered carefully to ensure that all areas are secure and particular attention must be given to shift doors, gates, keeper access doors, locking mechanisms and exhibit barrier dimensions and construction.

Otters can climb, and will take advantage of anything provided. Trees, bushes, etc., should be placed away from exhibit perimeters. Walls should be non-climbable, and fences should be strong and inhibit climbing. The containment walls/fence should be at least 1.52m (5ft) high for *A. cinereus* and at least 1.83m (6ft) high for *L. canadensis*, *L. maculicollis*, *P. brasiliensis*, and *A. capensis*. While these heights should contain most otters, Ben-David (personal communication) reported a *L. canadensis* scaling a 3m (9.8ft) fence. Animals that are known to be climbers may require additional containment height or features. It has been shown that *Lutra lutra* can clear 1.3m (4.27ft) when leaping from the ground to a platform, 1.6m (5.25ft) when jumping from one platform to another, and 0.92m (3ft) when jumping from the water onto a platform, if they are able to push off from the pool bottom (Reuther 1991). If containment barriers are mesh, they should be topped with an un-climbable, inward-facing overhang of 80cm (2.7ft) (Duplaix-Hall 1975; Foster-Turley 1990). Hot wire can be used, but should not be accessible to an animal in the water, and should be placed at a height that will not cause injury to an animal if they fall as a result of touching the wire.

> **AZA Accreditation Standard**
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> **(11.3.1)** All animal exhibits and holding areas must be secured to prevent unintentional animal egress.

> **AZA Accreditation Standard**
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> **(11.3.6)** Guardrails/barriers must be constructed in all areas where the visiting public could have contact with other than handleable animals.

> **AZA Accreditation Standard**
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> **(11.2.3)** All emergency procedures must be written and provided to staff and, where appropriate, to volunteers. Appropriate emergency procedures must be readily available for reference in the event of an actual emergency. These procedures should deal with four basic types of emergencies: fire, weather/environment; injury to staff or a visitor; animal escape.

Otters also dig proficiently (particularly *A. capensis*, *L. canadensis*, and *P. brasiliensis*). When designing exhibits, perimeter walls and fences should be buried or mesh linked. Sinking perimeter fences/walls at least 80cm (2.6ft) is advisable for most species, however, Sykes-Gatz (2005) recommends sinking perimeter fences deeper than 1m (3.28ft) for *P. brasiliensis*, because this species easily digs down that far. Holes along perimeter containment should be promptly refilled. To provide additional safety, secondary containment areas should be constructed at all enclosure entrances.

Exhibits in which the visiting public may have contact with animals must have a guardrail/barrier that separates the two (11.3.6). Barriers should be high enough to prevent visitors from reaching over or into the exhibit.

In case of an otter escaping institutional and AZA policies for containment should be followed. Typically all otters will run and hide if they are not able to return to their exhibit. Staff should notify the appropriate chain of command, keep the animal in sight, ask the public to leave the area, and attempt to slowly encourage the animal into a building or other confined area. Nets, push boards, and gloves should be availbe in case of these emergencies. Tranquilizer guns should not be used on these species, particularly the smaller ones. Instead, if anesthesia is required a blow pipe should be used. In the case of severe weather (i.e. hurricanes, tornados, thunderstorms animals should be confined to indoor holding. Policies regarding other emergencies such as fire or earthquake should be developed by each institution based on the species and enclosure design. For all of these situations, an institutional policy should be developed in advance and all emergency safety procedures should be clearly written, provided to appropriate staff and volunteers, and readily available for reference in the event of an actual emergency (11.2.3).

Staff training for emergencies must be undertaken and records of such training maintained. Security personnel must be trained to handle all emergencies in full accordance with the policies and procedures of the institution and in some cases, may be in charge of the respective emergency (11.6.2).

Emergency drills should be conducted at least once annually for each basic type of emergency to ensure all staff is aware of emergency procedures and to identify potential problematic areas that may require adjustment. These drills should be recorded and evaluated to ensure that procedures are being followed, that staff training is effective and that what is learned is used to correct and/or improve the emergency procedures. Records of these drills should be maintained and improvements in the procedures duly noted whenever such are identified. AZA-accredited institutions must have a communication system that can be quickly accessed in case of an emergency (11.2.4).

AZA-accredited institutions must also ensure that written protocols define how and when local police or other emergency agencies are contacted and specify response times to emergencies (11.2.5). AZA-accredited institutions which care for potentially dangerous animals must have appropriate safety procedures in place to prevent attacks and injuries by these animals (11.5.3). Animal attack emergency response procedures must be defined and personnel must be trained for these protocols (11.5.3).

Otter attack emergency drills should be conducted at least once annually to ensure that the institution's staff know their duties and responsibilities and know how to handle emergencies properly when they occur. All drills need to be recorded and evaluated to ensure that procedures are being followed, that staff training is effective, and that what is learned is used to correct and/or improve the emergency procedures. Records of these

AZA Accreditation Standard

(11.6.2) Security personnel, whether staff of the institution, or a provided and/or contracted service, must be trained to handle all emergencies in full accordance with the policies and procedures of the institution. In some cases, it is recognized that Security personnel may be in charge of the respective emergency (i.e., shooting teams).

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(11.2.4) The institution must have a communication system that can be quickly accessed in case of an emergency.

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(11.2.5) A written protocol should be developed involving local police or other emergency agencies and include response times to emergencies.

AZA Accreditation Standard

(11.5.3) Institutions maintaining potentially dangerous animals (sharks, whales, tigers, bears, etc.) must have appropriate safety procedures in place to prevent attacks and injuries by these animals. Appropriate response procedures must also be in place to deal with an attack resulting in an injury. These procedures must be practiced routinely per the emergency drill requirements contained in these standards. Whenever injuries result from these incidents, a written account outlining the cause of the incident, how the injury was handled, and a description of any resulting changes to either the safety procedures or the physical facility must be prepared and maintained for five years from the date of the incident.

drills must be maintained and improvements in the procedures duly noted whenever such are identified (11.5.3).

If an otter attack occurs and injuries result from the incident, a written account outlining the cause of the incident, how the injury was handled, and a description of any resulting changes to either the safety procedures or the physical facility must be prepared and maintained for five years from the date of the incident (11.5.3).

Chapter 3. Transport

3.1 Preparations

Animal transportation must be conducted in a manner that adheres to all laws, is safe, and minimizes risk to the animal(s), employees, and general public (1.5.11). Safe otter transport requires the use of appropriate conveyance and equipment that is in good working order.

When transporting otters there always should be at least two people present; if animals have been anesthetized the veterinarian always should be present. Staff involved in transports should understand their duties and have a clear idea of the institutions policies regarding transports. The Otter SSP has no specific recommendations regarding staff roles in transports but does recommend procedures and policies be clearly defined and understood in advance by all participating staff.

> **AZA Accreditation Standard**
>
> **(1.5.11)** Animal transportation must be conducted in a manner that is safe, well-planned and coordinated, and minimizes risk to the animal(s), employees, and general public. All applicable local, state, and federal laws must be adhered to.

The equipment must provide for the adequate containment, life support, comfort, temperature control, food/water, and safety of the otters(s). Safe transport also requires the assignment of an adequate number of appropriately trained personnel (by institution or contractor) who are equipped and prepared to handle contingencies and/or emergencies that may occur in the course of transport. Planning and coordination for animal transport requires good communication among all affected parties, plans for a variety of emergencies and contingencies that may arise, and timely execution of the transport. At no time should the otter(s) or people be subjected to unnecessary risk or danger.

> The transport of wild animals is regulated by the International Air Transport Association (IATA). The standards of care provided within this chapter are based on IATA regulations (e.g., IATA 2007), best practice recommendations from the AZA Otter SSP, and AZA Accreditation Standards.

Pre-shipment Exams: All otters should receive a thorough pre-shipment physical examination (for more information, see Chapter 6). Ideally, a copy of the pre-shipment physical exam findings and laboratory work should be sent to the veterinarian at the receiving institution before the animal is transferred. If an otter has a current medical condition requiring ongoing treatment, the case should be discussed between the veterinarians at the shipping and receiving institutions before the animal is moved. All animal shipments should be accompanied by a hard copy of the medical record, as well as a health certificate and the USDA acquisition, disposition, or transport form (form #7020) (APHIS 1997) as well as any other paperwork required by IATA, carrier, or regulatory agency. Institutions using MedARKS should provide the receiving institution with electronic copies of the medical records. Dietary, enrichment, and training records should be sent prior to shipping the animal

Crate Requirements: All possible relevant regulatory agencies should be checked for shipping, health, and permit requirements before transporting animals (USFW, state regulations, CITES, etc.). The International Air Transport Association (IATA) publishes specific guidelines for transport containers used for animal shipments. These guidelines are available from the Publication Assistant, IATA, 2000 Peel Street, Montreal, Quebec, Canada, H3A 2R4 (Ott Joslin & Collins 1999). An alternate address for IATA is International Air Transport Association, 800 Place Victoria, P.O. Box 113, Montreal, Quebec, Canada, H4Z 1M1. The Live Animals Regulations document is available in print or CD ROM format, and can be ordered from sales@iata.org. IATA regulations change periodically, and so the most recent publication or website should be consulted. It is very important to adhere closely to these requirements as airlines may refuse to fly animals in containers that do not conform to the guidelines. In general, IATA regulations require the following:

All shipping crates should allow for adequate ventilation. Ventilation apertures should be small enough to prevent the escape of the animal and small enough that the animal cannot get any part of its body through the opening.

Generally, a Vari Kennel® may be used for *A. capensis*, *A. cinereus*, *L. canadensis*, and *L. maculicollis*, with the following modifications. The grill door should be covered with securely fixed weld mesh and all ventilation openings covered with wire mesh. The door should have secure fasteners at the top and the bottom. A curtain that can be raised and lowered and does not block ventilation should be fixed over the door to reduce light inside the container. A dropping tray should be fixed to the floor and filled with absorbent material; the container should be leak-proof. There should be ventilation openings on the rear of the container; extra ventilation openings may have to be added in order that the total ventilation area is at least 20% of the four sides. The container should be correctly labeled. If the container has wheels, they should be removed or rendered inoperable. Airlines also may require a wire mesh cage be fitted to the inside of the Vari Kennel®.

Shipping crate doors should be secured with additional fasteners; *A. cinereus* has been known to pull doors inwards when they could not push them out and escape from containers that were not fastened securely. This proviso applies to all otter species.

Otters should be transported separately. *A. cinereus* have been transported successfully in groups when the animals are less than six months of age. IATA states that "the height of the container must allow the animal to stand in a natural position with its head extended and the width must permit it to turn around and lie down comfortably" (IATA 2009). Sykes-Gatz (2005) reports successful shipments of *P. brasiliensis* in containers measuring 140cm (55.2in) in length, 60cm (23.6in) in width, and 57cm (22.5in) tall. However, all IATA and airline regulations should be checked prior to selecting or constructing shipping containers.

P. brasiliensis: Sykes-Gatz (2005) states that *P. brasiliensis* should not be transported in hard plastic containers in situations other than within an institution. Airlines and IATA regulations should be consulted for appropriate transport containers for this species. It is advisable to ship this species without a wire mesh lining, as it could be harmful to their exceptionally sensitive footpads and webbing between the toes (Sykes-Gatz 2005). Lining of this type has not been proven necessary for safety when transporting this species. However, IATA requires that giant otter be shipped in containers with an interior of heavy-gauge wire mesh or sheet metal lining (IATA 2009). IATA regulations require the floor to be solid-metal, leak proof and covered with a thick layer of absorbent material (e.g. shavings). It is very important that the relevant IATA and airline regulations be checked prior to constructing all shipping containers and complied with to ensure acceptance by the transport carrier.

3.2 Protocols

Transport protocols should be well defined and clear to all otter care staff. Institutional protocols for planning and carrying out transports should be used for otters. There are no specific requirements. However, the AZA Otter SSP does encourage facilities to train their animals to voluntarily enter crates for transport.

Food and Water:

IATA requires that the crate allows for feeding and watering of the animal if needed. The food and water ports should be clearly marked on the outside of the crate. In case of delay on long flights, provisions should be made for feeding in transit (this may necessitate shipping food with the animal).

Otters do not normally require additional food and water during transport for periods up to 24 hours. If unforeseen delays occur, canned dog food or cat food may be offered. Adequate moisture is present in these foods to take care of water needs during short periods. A metal bowl may be attached to the corner of the crate; this should be accessible from the outside. Always check for any specific IATA or shipping carrier regulations or requirements.

Bedding and Substrate: Bedding, such as straw or shavings, should be placed in the transport container for the animal's comfort and absorption of feces and urine. Types of bedding material allowed should be checked with the airline. In order to separate urine and feces from the animal, IATA requires a drop tray be fixed to the floor of the crate and filled with absorbent material.

Temperature: Temperatures of 7.2-26.7°C (45-80°F) that are permitted by the airlines for transport are not appropriate at the high end for otter species. Otters should not be exposed to 21.1°C (70°F) temperatures for periods longer than 15 minutes when contained in a shipping crate. These animals can easily overheat at elevated temperatures (21.1°C or 70°F and higher), especially when stressed and/or contained in a shipping crate. While this is a more restrictive temperature range than previously recommended, past experience has shown that otters quickly overheat and succumb to hyperthermia. Animals in unheated vehicles should not be exposed to temperatures less than 7.2°C (45°F).

A. cinereus: *A. cinereus* should not be exposed to drafty 7.2°C (45°F) temperatures for extended periods. This also may be true for *L. maculicollis*, but it is not known at this time and requires further research.

L. canadensis: *L. canadensis* exhibit a low heat tolerance and should not be shipped when temperatures are forecasted to exceed 21.1°C (70°F) at transfer locations.

P. brasiliensis: *P. brasiliensis* have been shown to have low heat tolerance. It is recommended that this species should not be transported in temperatures below 15.5°C (60°F) or above 26.6°C (80°F) (Sykes-Gatz 2005).

Light and Sound: As far as possible, noise should be kept to a minimum (including sudden loud noises, constant high-pitch noises, or anything considered uncomfortable to people), and the animal kept in low light conditions. Mesh doors or side windows (i.e., as in air kennels) should be covered with a breathable, opaque material to allow for ventilation and privacy for the animal (Ott Joslin & Collins 1999). These basic noise reduction precautions will help reduce stress from sudden, frightening sounds, and low light will provide a minimal sense of security for species that hide in small, dark spaces (e.g., dens) when frightened.

Animal Monitoring: Otters should not be maintained in shipping containers for longer than 24 hours without food and water. Directions on what should be done in the case of an emergency that necessitates animal treatment should accompany all animal shipments. If access to an animal is required due to illness or extended shipment delays, the shipping/receiving institutions should be notified. The crate should be transferred to the nearest zoo or veterinary clinic prepared to handle the animal.

Emergencies: In case of emergencies contact information for the shipping and recipient zoos should be included on the paper work and the crate. If an otter escapes, the animals should be closed into a confined space (if possible) and both institutions notified as quickly as possible. At a minimum, visual contact (from a distance) should be maintained until one or both institutions have been contacted and professionals have arrived.

Post-transport Release: Upon arrival at their destination, shipping crates should be placed inside the quarantine holding pen, the door opened, and the animal left to exit at will. All holding pens should be provided with food, water, alternate hiding places, appropriate bedding, and enrichment structures.

Chapter 4. Social Environment

4.1 Group Structure and Size

Careful consideration should be given to ensure that otter group structures and sizes meet the social, physical, and psychological well-being of those animals and facilitate species-appropriate behaviors.

A. cinereus: In zoos and aquariums, Asian small-clawed otters are monogamous, with both members of the pair helping to raise the offspring. Unlike *L. canadensis*, the otter parents and offspring should be housed together. Older siblings help raise the younger ones, and a family group or breeding pair can produce two litters a year with up to seven pups in each litter. Similar sized groups are sometimes found in the wild (Foster-Turley 1990). It is recommended that these otters be held in adult pairs, adult pairs with offspring, or single sex groups. The size of the group will depend on the size of the exhibit and the compatibility of the individual animals (Lombardi et al. 1998). The formation of single sex groups should be accomplished at a very young age to avoid aggression.

A. capensis: *A. capensis* have been observed in multiple social situations. In fresh water inland systems, the male stays with the female and offspring (Rowe-Rowe 1978). Clans up to 10-15 individuals have been observed. In marine ecosystems, it has been noted that the male does not stay with the family group (Estes 1989). Females with pups are the typical family group (Chanin 1985), but this species is often seen alone (Chanin 1985). In zoos and aquariums, pairs have been separated during parturition and early pup rearing. This species has been held in pairs and in groups of 1.2 in zoos and aquariums (Reed-Smith & Polechla 2002).

L. canadensis: *L. canadensis* are believed to be more social than most other mustelids (but not as social as some of the other otters), based on the findings of a number of researchers (Beckel 1982; Rock et al. 1994; Testa et al. 1994; Johnson & Berkley 1999; Blundell et al. 2002a,b; Gorman et al. 2006, M. Ben-David, personal communication; S. Shannon, personal communication). For example, Stevens & Serfass (2008) documented that out of 172 film documentations of otters using latrines, 59.3% were by single otters, 19.2% were by two otters, 17.4% were by three otters, and 4.1% were by four otters.

A variety of social groupings have been documented, but in general, the following are most typical: female with offspring; lone male; group of males; lone female; group of males with sub-adult females; pair (during mating season only); or two females with offspring. Male groups have been reported by Blundell et al. (2002a, 2002b, 2004) and Hansen (2004); female groups were recorded by Gorman et al. (2006)

Otter associations may vary with the habitat in which the animals are found. Blundell et al. (2002a,b) found otters living in areas rich in resources seem to show more of a tendency to socialize, particularly the males and sub-adult females. Discounting female and pup associations, the Blundell et al. (2002a,b) telemetry study found that females were asocial in 47% of their locations, while males were asocial during only 24% of their locations. Further, they determined that among the "social" otters, males were social in 46% of their locations and 63% of that time was spent in all male groupings. "Social" females were located in social groupings only 26% of the time, and 78% of that time they were located in mixed-sex groupings (Blundell et al. 2002a,b).

Gorman et al. (2006) found in Minnesota that annual home ranges for males were 3.2 times greater than for females, and annual core areas for males were 2.9 times greater than for females. Core areas frequently overlapped with interactions between overlapping resident females and males most common (51.7%), while male-male (27.6%) and female-female (20.7%) interactions were slightly less frequent. Female core areas overlapped those of other females 22.2% of the time and males 15.8% of the time; male core areas overlapped with those of other males on average 15.7% of the time, on average. They concluded that river otters living in the upper Mississippi watershed "exhibited clear evidence of space sharing, suggesting that individuals in this population were neither solitary nor territorial….[otters] appeared to socialize to some degree with any individual for which they had an encounter opportunity."

Zoos and aquariums would qualify as "habitats rich in resources," thus providing the opportunity to keep more social groupings of this species as found in some portions of their range. Ben-David et al. (2000, 2001a, and b) maintained 15 unrelated males in one enclosure for 10 months with little to no problem.

There are no minimum or optimum group sizes for this species. To the contrary, this species' behavioral plasticity allows the formation of social groups not normally associated with a typically solitary species. Preferred groupings include: multiple males, a male-female pair, one male and multiple females

(1.2), multiple pairs (2.2). The only social grouping not recommended is all female, unless they are related (or introduced at a very young age) and have been together continuously (Reed-Smith 2001). This does not mean it is impossible to introduce adult females or house unrelated females together, just that it is very difficult and frequently unsuccessful (Reed-Smith 2001, 2004b). See Introductions/Reintroductions and Appendix N for additional information. There are some indications that pairs raised together do not breed well; in situations where breeding is desired, one of these young otters may need to be switched with an unfamiliar animal.

Multiple pair groupings should be monitored closely for signs of stress in subordinate animals. Groupings of multiple males with one female are not recommended, but can be maintained if monitored closely or separated during breeding season to prevent the males from fighting over the female or causing harm to the female with overly attentive advances (Reed-Smith 2001).

L. maculicollis: This species is best housed as pairs or family groups. All male groups also may be a good option but not enough is known yet to determine how this natural social pattern works in a confined setting. While several field researchers have reported seeing large groups (10-20 individuals) of *L. maculicollis* (Proctor 1963; Kruuk & Moorhouse 1990; Kruuk 1995, J. Reed-Smith in prep), it is not known what role, if any, older siblings play in caring for younger pups or how often family groups join. Proctor (1963) reported observing groups of about five otters most frequently, a size believed to be consistent with a single family. Ongoing studies in Lake Victoria cite frequent observation of groups of 6-8 animals, at times constituted of animals of varying sizes (J.Reed-Smith, personal observation).

It is considered unwise to introduce adult males and difficult to introduce adult females. In the latter case, some females show a marked preference for certain females over others. Compatible groups of females may show aggression towards one another at times, particularly if a female is in estrus (Scollhamer 1987). In one instance, older pups have been maintained in an exhibit with the adult pair and younger pups (R.Willison, personal communication); the female kept the older pup and adult male at bay as parturition neared. However, there have been reports of adult females ostracizing female offspring which required the young female to be moved (Benza et al. 2009). Reports from the wild that this species typically live as pairs, with both parents participating in pup rearing, have come under some doubt (Reed-Smith in prep). This has not been the case in zoos and aquariums, at least during the early phase of pup rearing; instead, the female raises the pups alone until they are active and swimming at which time the male is again allowed into the group (R.Willison, personal communication). Recent field studies in Lake Victoria, Tanzania (Reed-Smith in prep) recorded single animals, mother with young, two or more adult females with young, male groups/pairs and adolescent groups as the social configurations sighted.

Optimal group size will vary with the size of the exhibit and compatibility of individuals involved, as with all otter species.

P. brasiliensis: There is a high degree of pair bonding and group cohesiveness in *Pteronura* (Duplaix 1980). In the wild, a mated pair normally bonds for life, and all family members, including offspring (one and two year olds) from previous litters, care for new pups (Schenck & Staib 1994). In zoos and aquariums, this species should be housed as mated pairs with young up to about two years of age. All male groups (e.g., 2-3 animals) can be exhibited as an alternative. Females can be kept together, but these are generally only successful as related duos or animals introduced at an early age. There are a few reports of adult females living together *ex-situ*, but it is not known if these animals were related. There are no reports of the successful introduction of adult females and this grouping is not recommended.

There are some indications that pairs reared together from a very young age, or introduced well before they reach sexual maturity, will not breed successfully (Sykes-Gatz 2005). Therefore, it is recommended that breeding pairs be introduced after they have matured sexually.

In the wild, one litter of 1-6 pups is born annually. The pups are dependent upon the other family members for care, socialization, learning life skills, etc. In zoos and aquariums, the pair should be allowed to rear their pups together. Preventing such activities often causes significant problems, such as litter loss, and inhibits the socialization of older young who have to learn pup care from their parents. Both parents will take care of the pups, teach them how to swim, feed them, groom them, etc. On occasion, both will move them to different nest boxes. In the wild (Staib 2002) and in zoos and aquariums (Sykes-Gatz 2005), the sire may sometimes take pups from the den only to have them immediately returned there by the dam. This behavior is considered normal and should not be viewed with alarm unless it is accompanied by signs of agitation or excessive stress (Sykes-Gatz 2005). See Chapter 7 for additional information on pup rearing in this species.

Single-sexed Groups: The following recommendations are provided for the formation and maintenance of single-sexed otter groups:

A. cinereus: Single-sexed groups are not seen in the wild, but all male groups are found in zoos and aquariums. Groups should be formed as early and at as young an age as possible; extra care should be given at feeding time. Extra dens should be provided so that every animal has its own area to be apart from the others (Lombardi et al. 1998).

While it is possible to keep single-sex sibling groups, it is not a preferred grouping and should be attempted only at the recommendation of the AZA Otter SSP. Generally, the group structure begins to break down beginning at sexual maturity, leading to increasing aggression toward subordinate animals. Typically, all male groups have been more successful than female groups and hinge on introduction of non-sibling animals at a very young age.

In one case, six females housed together all of their lives began fighting at the ages of 3-5 years, when the dominant female began harassing the most submissive animal. This behavior was not apparently associated with estrus, and reduction of the aggression was originally handled through training (the animals had been conditioned to separate into two groups and station singly for brief periods) and positive reinforcement (e.g., the aggressive and subordinate animals were reinforced for looking calmly at one another – this was not successful). However, with time, the aggressive behavior focused on expelling the subordinate female spread throughout the group and previously friendly otters began to have problems as well. The situation eventually resulted in the need to permanently separate the dominant and subordinate animals, at first rotating them in with different group members and then sending them out to different institutions (S.Duncan, personal communication).

A. capensis: Information on the success of single-sexed groups is unknown at this time, and further research is required.

L. canadensis: All male groups do very well together and have been repeatedly documented in the wild, particularly in resource rich environments (Blundell et al. 2002a and b; G.Blundell, personal communication; S.Shannon, personal communication, Gorman et al. 2006). Fifteen unrelated adult males were successfully housed together for 10 months at the Alaska Sea Life Center (Ben-David et al. 2000, 2001a,b), and five males have been successfully housed together at the Virginia Aquarium (C.Harshaw, personal communication). All male groupings are suggested for non-breeding situations.

Sub-adult females (older than one year but less than two years of age or prior to first estrus) and females without pups (this occurs less often) have been known to associate with all male groups (Blundell et al. 2004). All female groups generally do not fare well, however these groups have been reported in the wild by Gorman et al. (2006). In zoos and aquariums, sisters or females introduced at an early age may be compatible for years. However, if one animal has to be separated, they may not re-establish this social equilibrium upon reintroduction (Reed-Smith 2001, see Appendix N for introduction/reintroduction protocol). In one other *in-situ* study, an all female clan was identified (S.Shannon, personal communication). All female groupings are not generally recommended.

L. maculicollis: Adult males introduced before the age of four months have been kept together successfully, as long as they are not exposed to the scent or sight of estrus females (Schollhamer 1987).

Adult females can be housed together, but their introduction may be difficult. Some females show a marked preference for certain females and a dislike of others. Females that have been previously housed together can be reintroduced after giving birth, once their pups are eating solid food at about three months of age (Schollhamer 1987). Aggression has been shown towards younger females but documented only in mixed-sex situations (Benza et al. 2009). Further research is required to determine the most compatible social groupings for this species.

P. brasiliensis: Single sex groups, typically two males, have been kept together successfully (rarely have groupings of three males been successful). However, these groupings should be raised together or introduced slowly to monitor any signs of potential aggression. There are a few reports of adult females living together but no additional information is available. The introduction of adult females to one another is not advised (Sykes-Gatz 2005).

4.2 Influence of Other Species and Conspecifics

Compatible otters typically do not require specific inter-individual distances. All animals should be provided with denning choices, allowing them the opportunity to sleep together or separately. Breeding pairs of *A. capensis*, *L. canadensis*, and *L. maculicollis* will require separation during the early stages of pup rearing or an exhibit environment that allows for physical and visual separation of the male and female.

While it is not recommended to house Asian small-clawed otter groups within visual or auditory range of each other (Lombardi et al. 1998), different groups of spotted-necked otter (R.Willison, personal communication), Eurasian otter (cited in Reed-Smith 2004b), and North American otters (Reed-Smith 2004b) have been successfully housed within auditory and visual range of one another. Care always should be taken that these groups are not experiencing undue stress; all animals should be monitored for any signs of stress or agitation (such as excessive grooming, screaming, pacing, or squabbling through containment wire). When animals are separated from a group for any reason (all species), care should be taken when reintroducing them; at times, all of the steps of an initial introduction should be followed (see 4.3 Introductions and Reintroductions). Some of these reintroductions, particularly with females, may not be successful, so prolonged separation, particularly of Asian small-clawed otters and giant otters, should be avoided except in cases of medical treatment, aggression, or parturition (this last condition is not applicable to Asian small-clawed and giant otters because they should be maintained within their groups for parturition).

A. cinereus: If a group of Asian small-clawed otters have to be split for some reason (e.g., aggression between two individuals), experience has shown that the non-aggressing animals should be rotated between the two groups to prevent aggression between additional animals. In these cases, the social bond between highly compatible animals appeared to break down if the animals were left separated from any particular individuals for more than about two weeks. These temporary groups were rotated between off-exhibit holding and the exhibit on a daily basis, allowing all animals time in each location, but essentially keeping the problem animals separated visually and physically (S.Duncan, personal communication).

Mixed-species Exhibits: All individuals housed in mixed-species exhibits should be routinely monitored for stress, injuries, and to ensure they are getting adequate food and water. Typically, otters are exhibited with species that focus behaviorally on other exhibit features than those used by the otters (e.g., arboreal species). Animals within mixed exhibits should be monitored for stress, and management plans should be made to accommodate older animals, special nutritional needs, impending births, etc.

A. cinereus: This species has been exhibited successfully with barbirusa, binturong, black hornbills, butterflies, peafowl, gibbons, giant hornbill, muntjac, Prevost squirrels, proboscis monkeys, slender-nose crocodiles, giant Asian squirrels, and Rodriguez fruit bats. Water monitors were tried, but were not successful.

A. capensis: Guenons were housed with *A. capensis* unsuccessfully at one facility; another facility housed them successfully with DeBrazza guenon, with occasional interspecific aggression (R.Willison, personal communication).

L. canadensis: *L. canadensis* have been successfully exhibited with beaver. There are unsubstantiated reports of exhibiting them with deer, fox, and possibly porcupine in large naturalistic exhibits, but these have not been confirmed. Any attempt at mixed species exhibits with the North American river otter should take into account their natural inquisitiveness, their semi-aquatic nature, their inclination to climb and dig, and their carnivorous diet.

L. maculicollis: Spotted-necked otters were housed successfully for an extended period with Schmidt's spot-nosed guenon, Allen's swamp monkey, and François langur. While food was placed in species appropriate locations, the otters did eat some of the monkeys' food. There were some reports of intermittent interspecific aggression, generally initiated by the otters in these groupings. An unusual aggressive event by a young otter in one of these mixed-exhibits lead to the death of a newborn monkey and several days later, the death of the aggressing otter. In this case, the exhibit was re-evaluated and discontinued. It should be noted that the otters and monkeys had been exhibited together for several years, but the unexpected birth of the guenon infant altered an un-easy truce established between the otters and monkeys.

P. brasiliensis: This species should not be kept in mixed-species exhibits due to their highly territorial and aggressive nature. There is a record of one unsuccessful attempt at housing giant otter with caiman, which was abandoned when a caiman attacked an injured otter and was subsequently removed.

4.3 Introductions and Reintroductions

Managed care for and reproduction of animals housed in AZA-accredited institutions are dynamic processes. Otters born in or moved between and within institutions require introduction and sometimes reintroductions to other animals. It is important that all introductions are conducted in a manner that is safe for all otters and humans involved.

Introductions may take anywhere from a few hours to weeks or months in some cases. There are some introductions that will never be successful. In general, these documented, unsuccessful cases involved multiple female *L. canadensis* or *P. brasiliensis*, male-male introductions in otter species other than *L. canadensis*, and some *L. canadensis* male-female pairings. However, there is always a risk that an introduction will not be successful so all attempts should be monitored closely.

Otters are very social animals; introductions should be carried out in the standard way, beginning with auditory and olfactory introductions, and then moving onto visual and limited tactile contact. Once affiliative behavior is observed (e.g., chuckling, rubbing, grunting, "friendly" pawing, and rolling – for additional behavior descriptions see Rostain 2000; Reed-Smith 2001; Rostain et al. 2004, Sykes-Gatz 2005), physical introduction can be attempted, preferably in neutral territory (see exceptions for giant otter below). New animals should be allowed to familiarize themselves with their new holding and exhibit prior to any physical introductions to other otters/future exhibit-mates. Introducing adult females to other adult females is difficult and is generally not recommended for any of the otter species (see Appendix N for suggested protocol if female/female introductions are tried). Groups of females that are compatible may exhibit aggression towards one another during estrus or be impossible to reintroduce after even a short separation (*L. canadensis* in particular). In cases of aggression during estrus, the females should be separated (Schollhamer 1987; J.Reed-Smith, unpublished data).

Auditory, visual, and olfactory introduction should be successfully completed before attempting physical introduction. Successful introductions have been reported as early as one day and have taken as long as several months or more. Training animals to station may be beneficial when attempting introductions, but there are limited data on its use with otters.

Reintroductions may need to follow the same steps, particularly if animals have been separated for any length of time. When animals are reintroduced to one another, even after short separations, staff should carry out each step (auditory, olfactory, visual, limited tactile, then full contact) to ensure animals do not experience undue stress or injury.

L. canadensis: Some introductions will never be successful with *L. canadensis*, particularly adult female to adult female introductions (Reed-Smith 2001) and some adult female to sexually immature male introductions. In general, the latter introductions should not be attempted while the female is in estrus; it is possible that immature males may be regarded as "female" by adult females leading to aggression on her part. Unsuccessful introductions should never progress beyond the screaming, lunging, or fighting stage, or result in stressful stand-offs where the animals stay away from one another, scream if the other approaches, and may refuse to go on exhibit if the other animal is there. Introductions of breeding pairs should be handled as any other, but may move more quickly, particularly during estrus. Introductions of all other *L. canadensis* pairings/groups should be conducted as indicated above.

Hand-reared *L. canadensis* may require some time to transfer their focus from humans to otters. The easiest way to accomplish this is to have their primary caregiver perform the introductions and stay with the animal (free contact situations) or on the other side of the caging (protected contact situations) for the initial introductions. Once the hand-reared animal is comfortable with the other otter, their attention often transfers immediately to that animal and away from the human caregiver. Hand-reared otters should be introduced to other otters as soon after weaning as possible. It is also recommended to raise more than one pup together, if possible (Reed-Smith 2004b). Fostering of pups to another nursing female was successful following an attempt (Columbus Zoo and Beardsley Zoo).

L. maculicollis: For male-female introductions, estrus female spotted-necked otters that have previously been with a male are easily introduced again to the same male or to a different, experienced male. When introducing a sexually mature, but inexperienced male and female, less aggression occurs if the pair is introduced when the female is not in estrus (Schollhamer 1987).

P. brasiliensis: *P. brasiliensis* that are unfamiliar with each other, and those that have been temporarily separated (i.e., animals that were previously housed together), should be introduced or reintroduced to each other in a gradual, cautious, and closely monitored manner. It is necessary that visual-acoustic-olfactory introductions be conducted before potentially dangerous, full-contact physical introductions are attempted. Reports of significant injury and death during improperly conducted giant otter introductions have not been uncommon. During introductions/reintroductions, females are more commonly reported to have injured/killed males, and even other females, than have males. However, males have been known to occasionally injure other males. Females appear to be the more dominant animal during typical introductions (this generally holds true even once pairing has been accomplished), and they seem to initiate fights more often (Sykes-Gatz 2005). Some giant otters, after 1-2 weeks of visual introduction, have been successfully introduced after their first full-contact day; other pairings have taken up to 8 weeks.

Initial positive reactions during the first few days of visual and full-contact introductions can be misleading. Otters may initially be compatible for some period of time, but show serious aggression later. Tolerance, stress, tiredness, aggression, affiliation levels, hunger, etc. should be considered when determining timing of initial full-contact sessions, when to increase contact time, and when to decrease or stop contact. Some tension and minor, non-harmful fights should be allowed, but temporary separation is required before serious fighting develops. During introductions, it is advisable to use fence barriers with ~2cm x 2cm (1" x 1") mesh size or cover existing fences with material of this mesh size. This will prevent fence fighting and/or injury to body parts. It is advisable to conduct introductions in areas of sufficient size, allowing for proper interaction between the otters; Sykes-Gatz (2005) recommends an introduction fence of at least 5m (16.4ft) in length. During introductions, the otters should be provided with sufficient living space to allow for their privacy and isolation if desired. The International Giant Otter Studbook Husbandry and Management Information and Guidelines (Sykes-Gatz 2005) provides greater detail on the recommended introduction procedure for this species.

Chapter 5. Nutrition

5.1 Nutritional Requirements

AZA Accreditation Standard

(2.6.2) A formal nutrition program is recommended to meet the behavioral and nutritional needs of all species and specimens within the collection.

A formal nutrition program is recommended to meet the behavioral and nutritional needs of all animals (2.6.2). Diets should be developed using the recommendations of veterinarians as well as AZA Taxon Advisory Groups, Species Survival Plans®, and Nutrition Advisory Groups (www.nagonline.net/feeding_guidelines.htm). Diet formulation criteria should address the otters' nutritional needs, feeding ecology, as well as individual and natural histories to ensure that species-specific feeding patterns and behaviors are stimulated.

The target nutritional values for otters are based on several sources. The cat is typically used to establish nutrient guidelines for carnivorous animals. The National Research Council (NRC) (1986, 2006), Association of American Feed Control Officials (AAFCO) (1994), and Waltham Center for Pet Nutrition (Earle & Smith 1993) have provided recommendations for cats. A limited amount of information is provided by the NRC publication for mink and foxes (1982), which represents the requirements of another mustelid species. The target nutrient values presented (Maslanka & Crissey 1998) are a range of values reported from various references. As new information becomes available, these ranges will change to reflect knowledge gained. Table 3 lists dietary nutrient ranges for otters.

Table 4 contains updated information on feline nutritional requirements based on NRC recommendations published in 2006. The original target values have been retained for comparison. See Dierenfeld et al. (2002) for information on nutrient composition of whole vertebrate prey. Appendix H provides a description of the nutrients listed in these tables.

Diet formulation should account for animal preferences, body weight, exercise, physical condition, environmental/seasonal changes, behavioral considerations, diet item availability, gastrointestinal tract morphology, and actual nutrient requirements.

Primarily piscivorous, otters have high metabolic rates, rapid digestion, and have been found to spend 41-60% of their time involved in feeding or foraging activities (Hoover & Tyler 1986; Davis et. al 1992; Kruuk 1995; J.Reed-Smith, unpublished data). Duplaix-Hall (1975) found that otters (unidentified species) in the wild rarely ate more than about 500g of food at a time and that they consumed approximately 20% of their own body weight daily. Kruuk (1995) reviewed his and other study results indicating that *ex-situ* populations of *Lutra lutra* consuming between 11.9-15% of their body weight maintained a healthy weight. Ben-David et al. (2000, 2001a and b) reported success using 10% of a *L. canadensis'* (*ex-situ* population) body weight as a guide for the basis of their maintenance diet. See section 5.2 for sample diets for the various otter species.

Table 3: Target dietary nutrient ranges for otters (dry matter basis).

Item	Target nutrient range*
Energy, kcal/g	3.6-4.0
Crude Protein, %	24-32.5
Fat, %	15-30**
Vitamin A, IU/g	3.3-10***
Vitamin D. IU/g	0.5-1.0
Vitamin E, mg/kg	30-120 [a]
Thiamin, mg/kg	1-5 [a]
Riboflavin, mg/kg	3.7-4.0
Pantothenic Acid, mg/kg	5-7.4
Niacin mg/kg	9.6-40
Pyridoxine, mg/kg	1.8-4.0
Folic acid, mg/kg	0.2-1.3
Biotin, mg/kg	0.07-0.08
Vitamin B-12, mg/kg	0.02-0.025
Choline, mg/kg	1000-3000
Calcium, %	0.6-0.8 [b]
Phosphorus %	0.6 [b]
Potassium, %	0.2-0.4
Sodium, %	0.04-0.06
Magnesium, %	0.04-0.07
Zinc, %	50-94
Copper, mg/kg	5-6.25
Manganese, mg/kg	5.0-9.0
Iron, mg/kg	80-114
Iodine, mg/kg	1.4-4.0

* Target nutrient ranges expressed on a dry matter basis derived from requirements for domestic cats (NCR 1986), AAFCO recommendations (1994), Waltham Center for Pet Nutrition recommendations (Earle & Smith 1993), and requirements for mink and foxes (NCR 1982).

** The fat content of fish commercially available in N.A. typically ranges from 5–40% (Maslanka & Crissey 1998), and N.A. river otters have been maintained on diets containing 24-30% fat (Reed-Smith 1994), thus an appropriate range for fat appears to fall between 15-30%.

*** The vitamin A requirement for cats is 10 IU/g (dry matter basis; NRC 1986), which represents the upper bound of the range. However, free-ranging N.A. otters consume a higher proportion of fish and may have a higher tolerance for vitamin A due to the high levels, which occur in their natural diet.

[a] When mostly fish diets are offered, the presence of unsaturated fatty acids and thiaminases causes the breakdown of these vitamins. Thus, dietary levels of 400 IU vitamin E/kg of dry diet and 100-120mg thiamin/kg of dry diet or 25-30mg thiamine/kg fresh weight as fed basis are recommended (Engelhardt & Geraci 1978; Bernard & Allen 1997).

[b] The recommended Ca:P ration is between 1:1 and 2:1

Table 4: Target nutrient ranges for carnivorous species (dry matter basis)

Nutrient	NRC 1986 Cat[1]	NRC 2006 Cat[2]			Arctic fox[3]	Mink[4]	Carniv[5]
	Maintenance	Growth	Maintenance	Gestation Lactation	Maintenance	Maintenance	All
Protein (%)	24-30	22.5	20	21.3-30	19.7-29.6	21.8-26	19.7-30
Fat (%)	9.0-10.5	9.0	9.0	15.0	--	--	9-15
Linoleic Acid (mg/kg)	0.5	0.55	0.55	0.55	--	--	0.5-0.55
Vitamin A (IU/g)	3.3-9.0	3.55	3.55	7.5	2.44	5.9	2.44-9
Vitamin D (IU/g)	0.5-0.75	0.25	0.25	0.25	--	--	0.25-0.75
Vitamin E (mg/kg)	27-30	38.0	38.0	38.0	--	27.0	27-38
Vitamin K (mg/kg)	0.1	1.0	1.0	1.0	--	--	0.1-1
Thiamin (mg/kg)	5.0	5.5	5.6	5.5	1.0	1.3	1-5.6
Riboflavin (mg/kg)	3.9-4.0	4.25	4.25	4.25	3.7	1.6	1.6-4.25
Niacin (mg/kg)	40-60	42.5	42.5	42.5	9.6	20.0	9.6-60
Pyridoxine (mg/kg)	4.0	2.5	2.5	2.5	1.8	1.6	1.6-4
Folacin (mg/kg)	0.79-0.8	0.75	0.75	0.75	0.2	0.5	0.2-0.8
Biotin (mg/kg)	0.07-0.08	0.075	0.075	0.075	--	0.12	0.07-0.12
Vitamin B_{12} (mg/kg)	0.02	0.022	0.022	0.022	--	0.032	0.02-0.032
Pantothenic acid (mg/kg)	5.0	6.25	6.25	6.25	7.4	8.0	5-8
Choline (mg/kg)	2400	2550	2550	2550	--	--	2400-2550
Calcium (%)	0.8-1.0	0.8	0.29	1.08	0.6	0.3-0.4	0.29-1.08
Phosphorus (%)	0.6-0.8	0.72	0.26	0.76	0.6	0.3-0.4	0.26-0.8
Magnesium (%)	0.03-0.08	0.04	0.04	0.06	--	--	0.03-0.08
Potassium (%)	0.4-0.6	0.4	0.52	0.52	--	--	0.4-0.6
Sodium (%)	0.05-0.2	0.14	0.068	0.132	--	--	0.05-0.2
Iron (mg/kg)	80.0	80.0	80.0	80.0	--	--	80
Zinc (mg/kg)	50-75	75.0	75.0	60.0	--	--	50-75
Copper (mg/kg)	5.0	8.4	5.0	8.8	--	--	5-8.8
Manganese (mg/kg)	5.0	4.8	4.8	7.2	--	--	4.8-7.2
Iodine (mg/kg)	0.35-0.42	2.2	2.2	2.2	--	--	0.35-2.2
Selenium (mg/kg)	0.1	0.4	0.4	0.4	--	--	0.1-0.4

[1] NRC (1986), Legrand-Defretin and Munday (1993), AAFCO (1994). All numbers are based on requirement set for maintenance.
[2] Dog and Cat NRC (2006).
[3] NRC (1982). Protein is range of growth and maintenance; vitamins are for growth, and minerals for growth and maintenance.
[4] NRC (1982). Protein is for maintenance, vitamins are for weaning to 13 weeks and minerals are a range of growing and maintenance.
[5] Combination of cat, mink, and fox

Changing Nutrient Requirements – Age: An animal's diet should be developed to maintain optimal weight or weight gain and normal physical development for a young animal. Diets for young or senescent adults should take into account their activity level, dental development and/or condition.

P. brasiliensis: In an *ex-situ* population study, Carter and Rosas (1997) determined that an adult consumed roughly 10% (range 6-16%) of their body weight daily and a sub-adult consumed 13.4% (range 8-18.9%). Earlier studies (Zeller 1960; Best 1985) reported similar findings with adults and sub-adults daily consuming 7-9.6% and 12.9% of their body weight, respectively. Amounts eaten can vary with air temperature and activity level changes, but if food is refused for one day, this could be a sign of sickness. Excess weight gain or loss and daily amounts and food types eaten should be monitored and recorded (Sykes-Gatz 2005).

Changing Nutrient Requirements – Reproduction: There is an increased need for energy during lactation. Tumanov & Sorina (1997) supported the use of high-energy diets for lactating female mustelids. Fat is the most concentrated source of energy in the diet. For lactating females, fat levels in the diet may be increased to support lactation (see below for exceptions) and also to provide increased energy to minimize mobilization of body stores and metabolic stress associated with milk production. Diet increases for lactating otters should be based on past experiences with individual otters and/or observed body weight loss (mobilization of tissue to support lactation). To date, institutions have typically increased the amount of fish offered a lactating female versus simply increasing the fat content by switching the type of food offered. An increase of 10-30% is the accepted rule.

P. brasiliensis: Hagenbeck and Wünnemann (1992) reported that lactating females at the Hagenbeck Tierpark generally increased their food consumption from 4.41-6.61 lbs/day to 13.23lbs/day (2-3kg/day to 6kg/day). They also reported increasing vitamin supplements during pregnancy/lactation and calcium supplementation during lactation (Sykes-Gatz 2005).

The energy requirements of a pair of otters, including a pregnant female, at the Philadelphia Zoo also increased during pregnancy and lactation. At this time, the energy intake of the pair increased to 246kcal/kg $BW^{0.75}$ (~2.75kg fish/animal fed at a ratio of 1:2 low- to high-fat fish). Fifty days postpartum and with one surviving pup, the intake of the pair was 236kcal/kg $BW^{0.75}$ (~3kg fish/animal fed at a ratio of 1:4 low- to high-fat fish). The female exhibited a preference for herring, trout, and catfish (K.Lengel, personal communication). It appears that feeding behaviors of *ex-situ* populations of reproductive *P. brasiliensis* mimic those of their wild counterparts. Rosas et al. (1999) found that during the birthing season, the diet of wild otters included a higher proportion of fish in the order Siluriformes (catfishes), which are higher in fat (37-41% fat DMB – Silva 1993) than fish in the order Percoidei (perch) (22-31% fat DMB – Twibell & Brown 2000),which are commonly fed on in the wild. Siluriformes are also higher in fat than Chiclidae (tilapia) (21-32% fat DMB – Toddes 2005-2006 analysis), which are the low-fat fish commonly fed to otters at the Philadelphia Zoo.

Seasonal Changes in Nutritional Needs: An animal's weight should be monitored regularly and diets adjusted accordingly. Some institutions report seasonal changes in appetite of some otters, but not in the majority of animals. Further research in this area is required. An animal's weight should be regularly monitored and diets adjusted accordingly. At this time, further research into seasonal nutritional requirements is required.

P. brasiliensis: The energy needs of *P. brasiliensis* are very dependent on their life stage, social grouping, and the ambient temperature of the environment. At the Philadelphia Zoo, an average energy intake of 173kcal/kg $BW^{0.75}$ (~2kg of fish fed at a ratio of 3:1 low- to high-fat fish) was adequate to maintain a single adult otter within a target weight range during the warmer months of the year (K.Lengel, personal experience).

When maintained with a mate, the same animal required an increased energy intake from 173kcal/kg to 201kcal/kg $BW^{0.75}$ (2.75kg of fish fed at a ratio of 3:1 low- to high-fat fish) during warmer months and went as high as 243kcal/kg $BW^{0.75}$ (2.75kg of fish fed at a ratio of 2:1 low- to high-fat fish) during cooler months (K.Lengel, personal experience).

A group of two adults, an 18-month old juvenile male, and three 6-month-old pups were successfully maintained during the summer season on an average energy allotment for the group of 545kcal/kg $BW^{0.75}$ (~6kg fish/animal fed at a ratio of 1.25:1 low- to high-fat fish). This energy allotment exceeded that of previous intake studies by almost double. However, the group was extremely active and primarily comprised of growing adolescent animals.

Weight Loss: While otters should carry some body fat and not be kept artificially thin, they are prone to gaining excessive weight in zoos and aquariums. Tarasoff (1974) reported subcutaneous fat deposits primarily at the base of the tail and caudally on the rear legs, with smaller deposits around the genitalia and in the axillary regions. There are several ways to approach formulating a weight loss diet for otters. Depending on the food items available, the feeding situation (fed alone or in a group), and the amount of weight loss desired, one or more of the following approaches may be appropriate.

Feed less total food: By reducing the amount of total food offered, weight loss may occur. This practice is confounded by the aggression observed in most otters, and particularly *A. cinereus* and *P. brasiliensis* groups, around feeding time and the potential for this to increase when less food is offered.

Add more water to the diet: By providing a diet that contains more moisture, the total calories in the diet are diluted and this may allow for weight loss. The otter can consume the same amount of total diet, but will actually be consuming fewer calories.

Increase the "bulk" of the diet: By adding indigestible or lower calorie items to the diet, the total "bulk" of the diet can be increased, effectively diluting the calories in the diet. The otter can consume the same amount of total diet, but will actually be consuming fewer calories.

Offer lower calorie items: Lower calorie items can be substituted in the diet. For example, fish varies in energy content from species to species. If weight loss is desired, a leaner fish, such as Pollock, could be substituted for a fattier fish, such as herring or capelin, to reduce total calories in the diet. This would be the preferred method for all otter species fed fish.

5.2 Diets

The formulation, preparation, and delivery of all diets must be of a quality and quantity suitable to meet the animal's psychological and behavioral needs (2.6.3). Food should be purchased from reliable, sustainable, and well-managed sources. The nutritional analysis of the food should be regularly tested and recorded.

Food preparation must be performed in accordance with all relevant federal, state, or local regulations (2.6.1). Meat processed on site must be processed following all USDA standards.

If browse plants are used within the animal's diet or for enrichment, all plants must be identified and assessed for safety. The responsibility for approval of plants and oversight of the program should be assigned to at least one qualified individual (2.6.4). The program should identify if the plants have been treated with any chemicals or near any point sources of pollution and if the plants are safe for the species. If otters have access to plants in and around their exhibits, there should be a staff member responsible for ensuring that toxic plants are not available. Before any plants are placed in or near otter exhibits, they should be vetted through relevant management staff (e.g. curator, veterinarian, grounds keeper, county poison control officer, etc.). Plants considered toxic to humans or other animals should be considered toxic to otters. Loquat (Weber and Garner 2002) consumption has proven fatal to Asian small-clawed otters (see useful Veterinary References). Otters are obligate carnivores but they will eat some vegetative matter such as berries and/or consume vegetation or other foreign material out of boredom or while exploring their environment.

AZA Accreditation Standard

(2.6.3) Animal diets must be of a quality and quantity suitable for each animal's nutritional and psychological needs. Diet formulations and records of analysis of appropriate feed items should be maintained and may be examined by the Visiting Committee. Animal food, especially seafood products, should be purchased from reliable sources that are sustainable and/or well managed.

AZA Accreditation Standard

(2.6.1) Animal food preparations must meet all local, state/provincial, and federal regulations.

AZA Accreditation Standard

(2.6.4) The institution should assign at least one person to oversee appropriate browse material for the collection.

Sample Diets: The one best diet for any of the otters of *ex-situ* populations has not been found and requires further research. However, current recommendations for all but *A. cinereus* are that a variety of fish species should be offered 3-4 times a week, preferably daily. Currently the AZA Otter SSP recommends a specific diet for *A. cinereus* (see below) and that *P. brasiliensis* should be offered fish daily as their main diet. The AZA Small Carnivore TAG chair should be contacted for specifics regarding the *A. cinereus* diet.

A. cinereus: The following food items represent the recommended daily diet per animal for *A. cinereus* (AZA Otter SSP recommendation 2006). The items are given as percentage of diet fed:

- 54.5% commercial canned diet designed to meet the nutrient requirements for domestic cats and control occurrence of calcium oxalate uroliths (e.g., Hill's x/d®, IAMS Moderate pH/O®).
- 2.5% commercial dry food to meet nutrient requirements for domestic cats and control occurrence of calcium oxalate uroliths (e.g., Hill's x/s®, IAMS Moderate pH/O®).
- 17.4% capelin
- 24.6% lake smelt
- 1% cricket and meal worms
- 100 IU vitamin E per kg of fish offered
- 25-35mg thiamin per kg of fish offered

A. capensis: The following food items represent a sample daily diet per animal for *A. capensis* in *ex-situ* environments.

- 182g Dallas Crown Carnivore Diet
- 908g fish (herring, capelin, sardines, two choices daily)
- 1.5 dozen large crayfish a week
- 30g lamb & rice dry dog food
- 90 IU vitamin E
- 6.25mg thiamin

L. canadensis: The amounts of food items in the sample diet below are based on achieving a target weight for otters. The diet should be fed at least three times a day and 4-5 times if possible. These additional feedings can consist of the fish, rib bones, and enrichment/training feeds.

- 155g commercially prepared feline diet, 2 x day, 7 days a week
- 112g capelin, 1 x day
- 120g smelt, 1 x day
- 135g trout, 1 x day
- ½ medium carrot, 1 x day scattered
- 2 rib bones, ox tail, or similar 3 x week
- 25-35mg thiamin per kg of fish offered
- 100 IU vitamin E per kg of fish

Only good quality, mostly fresh water fish, low in thiaminase and fat should be offered (Wünnemann 1995a). The fish source(s) and/or vendor(s) should be examined closely to assess their handling practices, ensure that HACCP guidelines are being met, and that the fish is considered human grade. Historical use of a type of fish by zoos and aquariums does not ensure it is an adequate diet ingredient, and only careful inspection of handling practices and the fish itself ensures consistent safety and quality. Most diets currently include horsemeat products, or alternative beef-based products which are available in addition to nutritionally complete dry and wet cat foods. The following sample diet is recommended for *L. canadensis*:

- 13.5% capelin
- 14.5% smelt
- 16.3% herring
- 18.2% carrots
- 37.5% nutritionally complete cat food or beef-based product (IAMS® cat food used for analysis)
- 2 bones, 3 per week (rib, ox/horse tail, or similar)
- 25-35mg thiamin and 100 IU vitamin E per kg of fish fed

L. maculicollis: The following food items represent a sample daily diet per animal for *L. maculicollis*. The animals should be fed at least twice per day:

- 50g Iams® Less Active Cat Kibble
- 150g Natural Balance zoo carnivore diet
- 150g trout (3 x week)
- 120g squid (3 x week)
- Yams and carrots offered in small amounts.

P. brasiliensis: A variety of good quality, fresh-water fish low in thiaminase and fat should be offered as the main diet (Wünnemann 1995a). Saltwater fish, high in fat, should only be offered occasionally. This species should be fed 3-5 times daily. Typically, 2-3kg (4.4-6.6lbs) of fish should be fed daily to each adult. Results of a survey of facilities housing this species indicate that all of these institutions offer fish daily (thawed, frozen, live, and/or freshly caught) as the main diet. Fish species offered include the following: rainbow trout (*Salmo gairdneri*), carp (*cyprinus carpio*), river fish (unidentified), tilapia, redeye (*Rutilus rutilus*), common bream (*Abramis brama*), herring* (*Culpea harengus*), mackerel* (*Scomber scombrus*), felchen (*Coregonus albula*), and channel catfish (*Ictalurus punctatus*). Fish species marked with an asterisk (*) can be used as a training reward or for vitamin delivery. If thawed frozen fish constitute the bulk of the diet the otters should be given supplementary B_1 (thiamine) and vitamin E. Supplements should be fed separately from the main feedings by at least 2 hours (Sykes-Gatz 2005).

For all otters: Fish types containing high thiaminase and/or high polyunsaturated fat levels should be avoided as they can cause malnutrition, sickness, and even death (Merck 1986). Diets containing fish high in thiaminase can lead to thiamin (vitamin B_1) deficiency in the otters fed this diet (Merck 1986). The process of fish storage (freezing), thawing, and preparation, can lead to fish nutrient loss, particularly vitamins B_1 and E, and especially in fish with high fat and/or high thiaminase content (Crissey 1998; Merck 1986). Vitamin supplements, especially vitamin B_1 (thiamin), vitamin E, and a multivitamin, should be added when fish is the main diet. The recommended vitamin supplementation regime for fish eating animals is as follows:

- Thiamin: 25-30mg/kg fish fed, fresh weight as fed basis (Bernard & Allen 1997)
- Vitamin E: 400 IU/kg dry weight basis (Engelhardt & Geraci 1978)

Based on the information above, the following food items represent a sample diet for giant otters (Sykes-Gatz 2005):

- 2-3kg (4.4-6.6lbs) fish/day/adult
- 400 IU vitamin E daily
- 100mg vitamin B_1 daily
- Multi-vitamin/mineral supplement 3 x week

Feeding Schedule: Due to their naturally nutrient dense diet, reliance on fat as a source of energy, rapid transit time of food through the intestinal tract, feeding style of frequent, small amounts, and generally high activity level – it is recommended that otters be fed at least twice a day and preferably three or more times daily (including enrichment or training feeds). *P. brasiliensis* should be fed 3-5 times per day. Frequent feeding prevents consumption of spoiled food, accommodates their rapid digestion (Ormseth & Ben-David 2000), and can stimulate increased activity in these generally active and curious species.

In addition to feeding smaller amounts frequently, it is recommended that a portion of the daily diet be fed as part of enrichment or husbandry training activities. At least one of the daily feedings, or part of a feeding, should be scattered to encourage foraging (except for giant otter). Timing of foraging opportunities and items offered should be varied to prevent habituation. All uneaten food should be removed before it spoils; this may be daily or more frequent in warm climates or seasons.

P. brasiliensis: Food for *P. brasiliensis* should not be scatter fed, as they do not forage on land and non-living food left uneaten in pools can be difficult to find. A portion of the daily diet can be used for daily training sessions with this species.

Food Variability: Otters should routinely be offered a variety of fish either as part of their diet or as enrichment. Reliance on multiple fish species, versus one or two, will prevent animals from developing strong preferences and help in switching them over to new sources if one fish type becomes unavailable.

With the exception of *P. brasiliensis* (see below), otters will sample a variety of food groups, especially if introduced to them at an early age; cat kibble, worms, crickets, vegetables, berries, mice, chicks, etc., can all be added to the diet as enrichment. Due to the possible formation of uroliths, foods high in calcium oxalates should be avoided (e.g., beans, celery, leafy greens, sweet potato, berries, peanuts, among others), particularly for *A. cinereus*. The use of these items for enrichment scatter feeds for North American river otter is acceptable on a limited basis, but the overall nutrient and caloric intake, body weight of the animal(s), and condition of the animal(s) should be taken into consideration.

All otters will benefit from receiving live fish/crayfish (from approved sources), at least as enrichment on a weekly basis. Whole fish should be the sole dietary item offered to *P. brasiliensis* and should comprise a portion of the daily diet of all other species.

A. cinereus: The AZA Otter SSP has specific diet recommendations for this species that should be obtained from the AZA Otter SSP Chair

A. capensis, *L. canadensis*, and *L. maculicollis*: It is recommended that fish constitute at least a portion of the daily diet offered these species. Hard dietary items should be routinely incorporated for dental health. These can include: hard kibble, crayfish, crabs, chicken necks, ox/horse tails, partially frozen fish, bony fish, day-old chicks, mice, rib bones, canine dental bones, or similar items.

P. brasiliensis: Staib (2002) reports that wild giant otters almost exclusively eat fish. In the wild, fish from the suborders Characoidei (characins), Percoidei (perch), and Siluroidei (catfish) make up the majority of the giant otter diet (Carter & Rosas 1997). A variety of good quality, fresh-water fish low in thiaminase and fat should be offered as their main diet (Wünnemann 1995a). Saltwater fish, high in fat, should only be offered occasionally. Gravid fish have caused diarrhea and appetite loss, and fish eggs should be removed before feeding (V.Gatz, personal observation). This species should be fed 3-5 times daily (typically 2-3kg [4.4-6.6lbs] fish/day/adult) (Sykes-Gatz 2005). A small amount of left over food is common and desirable to ensure all members of the group receive their portion and to avoid fights over fish. Uneaten fish should be regularly removed to prevent the otters from consuming spoiled food (Sykes-Gatz 2005). The strategy of feeding animals multiple times per day and using at least some feedings as training sessions has been successful at maintaining animal weights and maintaining low levels of food aggression in the group (Toddes 2005-2006).

Species-appropriate Foraging and Feeding: Live fish and crustaceans can and should be provided, if possible, on a regular basis. However, due to the risks of live fish or crayfish transmitting disease or parasites, policies regarding the feeding of live prey should be established by each facility. If these items are used, they should be obtained only from known, institutionally approved sources. Where live prey are used, provisions in the exhibit should be made to allow these prey species a place to hide from the otters, thus forcing the otters to use their hunting skills and extending the time of activity.

There also are a variety of puzzles and other feeding devices described in the literature that can be adapted for use in river otters. Alternatively, feeding tubes can be built into exhibits that randomly release live prey or food items into the exhibit. See Chapter 8, section 8.2 for other enrichment items used, including non-food items.

5.3 Nutritional Evaluations

The Otter SSP is currently beginning work on a body-condition matrix that can be used to help assess proper weight and condition for otters (See Appendix O for working matrix draft). At this time there are no known tools for performing clinical nutritional evaluations of otters; this would be a useful area for future research.

Chapter 6. Veterinary Care

6.1 Veterinary Services

AZA General Policies: Veterinary services are a vital component of excellent animal care practices. A full-time staff veterinarian is recommended, however, in cases where this is not practical, a consulting/part-time veterinarian must be under contract to make at least twice monthly inspections of the animal collection and to any emergencies (2.1.1). Veterinary coverage must also be available at all times so that any indications of disease, injury, or stress may be responded to in a timely manner (2.1.2). All AZA-accredited institutions should adopt the guidelines for medical programs developed by the American Association of Zoo Veterinarians (AAZV) (www.aazv.org/associations/6442files/zoo_aquarium_vet_med_guidelines.pdf). The current Veterinary Advisor for the AZA Otter SSP is Dr. Gwen Myers.

Protocols for the use and security of drugs used for veterinary purposes must be formally written and available to animal care staff (2.2.1). 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.

Animal recordkeeping 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 (1.4.6). Recordkeeping must be accurate and documented on a daily basis (1.4.7). Complete and up-to-date animal records must be duplicated and retained in a fireproof container within the institution (1.4.5) as well as be duplicated and stored at a separate location (1.4.4). Giant otter are a USFWS and IUCN listed endangered species, all relevant record keeping regulations should be followed.

AZA Accreditation Standard

(2.1.1) A full-time staff veterinarian is recommended. However, the Commission realizes that in some cases such is not practical. In those cases, a consulting/part-time veterinarian must be under contract to make at least twice monthly inspections of the animal collection and respond as soon as possible to any emergencies. The Commission also recognizes that certain collections, because of their size and/or nature, may require different considerations in veterinary care.

AZA Accreditation Standard

(2.1.2) So that indications of disease, injury, or stress may be dealt with promptly, veterinary coverage must be available to the animal collection 24 hours a day, 7 days a week.

AZA Accreditation Standard

(2.2.1) Written, formal procedures must be available to the animal care staff for the use of animal drugs for veterinary purposes and appropriate security of the drugs must be provided.

AZA Accreditation Standard

(1.4.6) A staff member must be designated as being responsible for the institution's animal record-keeping system. That person must be charged with establishing and maintaining the institution's animal records, as well as with keeping all animal care staff members apprised of relevant laws and regulations regarding the institution's animal collection.

AZA Accreditation Standard

(1.4.7) Animal records must be kept current, and data must be logged daily.

AZA Accreditation Standard

(1.4.5) At least one set of the institution's historical animal records must be stored and protected. Those records should include permits, titles, declaration forms, and other pertinent information.

AZA Accreditation Standard

(1.4.4) Animal records, whether in electronic or paper form, including health records, must be duplicated and stored in a separate location.

6.2 Identification Methods

AZA Policies: Ensuring that animals are identifiable through various means increases the ability to care for individuals more effectively. Animals must be identifiable and have corresponding ID numbers whenever practical, or a means for accurately maintaining animal records must be identified if individual identifications are not practical (1.4.3).

AZA Otter SSP Identification Recommendations: The AZA Otter SSP recommends that all otters be identified as soon as possible after birth with a transponder chip. Chips have been placed subcutaneously over the bridge of the nose/forehead area, SQ/IM in the intrascapular area at the base of the ears, and many institutions have placed them between the scapulas (recommended placement for giant otter). Placement in all of these areas has been met with success and failure (migration, loss, unable to read them as planned). At this time, the AZA Otter SSP recommends the forehead area as the preferred area of placement for all but the giant otter (see below); this location should make the chip easy to read when the animal comes to the front of the cage. The intrascapular area should be used as an alternative (this is the most frequently used location reported by member institutions). However, transponders placed in the intrascapular area can migrate and may be broken or lost during fighting and breeding attempts. Placement location of the transponder chip should be recorded in the animal's medical record.

AZA Accreditation Standard

(1.4.3) Animals must be identifiable, whenever practical, and have corresponding ID numbers. For animals maintained in colonies or other animals not considered readily identifiable, the institution must provide a statement explaining how record keeping is maintained.

P. brasiliensis: For this species it is recommended that transponder chips be placed in the neck behind the left ear or the intrascapular area. Transponders should be placed deeply under the skin or intramuscularly to decrease the risk of lost or damaged transponders (C.Osmann, personal communication).

AZA Acquisition/Disposition Policies: AZA member institutions must inventory their population at least annually and document all animal acquisitions and dispositions (1.4.1). Transaction forms help document that potential recipients or providers of the otters should adhere to the AZA Code of Professional Ethics, the AZA Acquisition/Disposition Policy (see Appendix B), and all relevant AZA and member policies, procedures and guidelines. In addition, transaction forms must insist on compliance with the applicable laws and regulations of local, state, federal and international authorities. All AZA-accredited institutions must abide by the AZA Acquisition and Disposition policy (Appendix B) and the long-term welfare of animals should be considered in all acquisition and disposition decisions. All species owned by an AZA institution must be listed on the inventory, including those animals on loan to and from the institution (1.4.2). Standard institutional and AZA approved acquisition and disposition policies and forms are used for otters. The AZA Otter SSP and relevant species coordinator should be notified when otters are acquired or transferred between institutions.

AZA Accreditation Standard

(1.4.1) An animal inventory must be compiled at least once a year and include data regarding acquisitions and dispositions in the animal collection.

AZA Accreditation Standard

(1.4.2) All species owned by the institution must be listed on the inventory, including those animals on loan to and from the institution. In both cases, notations should be made on the inventory.

6.3 Transfer Examination and Diagnostic Testing Recommendations

AZA Policies: The transfer of animals between AZA-accredited institutions or certified related facilities due to SSP or PMP recommendations occurs often as part of as concerted effort to preserve these species. These transfers should be done as altruistically as possible and the costs associated with specific examination and diagnostic testing for determining the health of these otters should be considered.

AZA Otter SSP Recommendations: See Section 6.4 Quarantine.

AZA Accreditation Standard

(2.7.1) The institution must have holding facilities or procedures for the quarantine of newly arrived animals and isolation facilities or procedures for the treatment of sick/injured animals.

6.4 Quarantine

AZA Policies: AZA institutions must have holding facilities or procedures for the quarantine of newly arrived animals and isolation facilities or procedures for the treatment of sick/injured animals (2.7.1). All quarantine, hospital, and isolation areas should be in compliance with AZA standards/guidelines (2.7.3; Appendix C). All quarantine procedures should be supervised by a veterinarian, formally written and available to staff working with quarantined animals (2.7.2). 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. If the receiving institution lacks appropriate facilities for quarantine, pre-shipment

AZA Accreditation Standard

(2.7.3) Quarantine, hospital, and isolation areas should be in compliance with standards or guidelines adopted by the AZA.

AZA Accreditation Standard

(2.7.2) Written, formal procedures for quarantine must be available and familiar to all staff working with quarantined animals.

quarantine at an AZA or AALAS accredited institution may be applicable. Local, state, or federal regulations that are more stringent than AZA Standards and recommendation have precedence.

AZA institutions must have zoonotic disease prevention procedures and training protocols established to minimize the risk of transferable diseases (11.1.2) 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 of the same class, 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.

> **AZA Accreditation Standard**
>
> **(11.1.2)** Training and procedures must be in place regarding zoonotic diseases.

Quarantine durations span of a minimum of 30 days (unless otherwise directed by the staff veterinarian). If additional carnivores are introduced into their corresponding quarantine areas, the minimum quarantine period should begin over again. However, the addition of mammals of a different order will not require the re-initiation of the quarantine period.

During the quarantine period, specific diagnostic tests should be conducted with each otter if possible or (see Appendix C). A complete physical, including a dental examination if applicable, should be performed. 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 -20ºC freezer for retrospective evaluation. Fecal samples should be collected and analyzed for gastrointestinal parasites and the otters should be treated accordingly. 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.

A tuberculin testing and surveillance program must be established for animal care staff as appropriate to protect both the health of both staff and animals (11.1.3). 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. TB testing is not routinely performed on otters. Animals should be permanently identified by their natural markings or, if necessary, marked when anesthetized or restrained (e.g., tattoo, ear notch, ear tag, etc.). Release from quarantine should be contingent upon normal results from diagnostic testing and two 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.

> **AZA Accreditation Standard**
>
> **(11.1.3)** A tuberculin testing and surveillance program must be established for appropriate staff in order to ensure the health of both the employees and the animal collection.

Although every living animal dies at some point, otters which die during the quarantine period should have a necropsy performed to determine the cause of death and the subsequent disposal of the body must be done in accordance with any local or federal laws (2.5.1). Necropsies should include a detailed external and internal gross morphological examination and representative tissue samples form the body organs should be submitted for histopathological examination.

> **AZA Accreditation Standard**
>
> **(2.5.1)** Deceased animals should be necropsied to determine the cause of death. Disposal after necropsy must be done in accordance with local/federal laws.

Otter SSP Quarantine Recommendations: All animals should undergo a 30-day quarantine stay at the receiving institution before incorporation into the rest of the collection. This allows time for the development of clinical signs of disease that may have been incubating before the animal was shipped. 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. Three fecal examinations for parasites should be performed and negative results obtained before release into their permanent enclosure. *P. brasiliensis* should be checked for *Strongyloides* species (Wünnemann 1990; C.Osmann, personal communication). The diet should be slowly adjusted over several weeks if there is to be a diet change. *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.

AZA Accreditation Standards and Related Policies

See Appendix F for specific animal care and management recommendations for small carnivore quarantine, which are included in the AZA Accreditation Standards and Related Policies (2008).

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 mustelid 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. If a pre-shipment physical examination has not been done before the animal was transferred, it would be prudent to perform a complete examination during the first week of quarantine (see section 6.2).

Final exam: During the last week of quarantine, a thorough examination should be conducted as outlined in section 6.2. It is extremely important to take radiographs of the animal during this time even if they were done at the previous institution (see note on *P. brasiliensis* below). This gives the new institution its own baseline film from which to compare future radiographs. This is especially important since radiographic techniques vary from facility to facility.

As giant otters are an endangered species that are very rare in zoos, the zoo animal population is highly valuable. The zoo veterinarian of the receiving institution should bear in mind that every anesthesia may be of high risk for each animal. Basic radiographs may be of importance from the medical point of view, but should only be taken if the otter is anesthetized for a special reason. Regular visual examinations of the otters' health status during quarantine should be performed, as well as parasitological and microbiological testing of fecal samples (C.Osmann, personal communication).

6.5 Preventive Medicine

AZA Accreditation Standard

(2.4.1) The veterinary care program must emphasize disease prevention.

AZA Accreditation Standard

(11.1.2) Training and procedures must be in place regarding zoonotic diseases.

AZA Policies: AZA-accredited institutions should have an extensive veterinary program that must emphasize disease prevention (2.4.1). The American Association of Zoo Veterinarians (AAZV) has developed an outline of an effective preventative veterinary medicine program that should be implemented to ensure proactive veterinary care for all animals (www.aazv.org/associations/6442/files/zoo_aquarium_vet_med_guidelines.pdf).

As stated in Chapter 6.4, AZA institutions must have zoonotic disease prevention procedures and training protocols established to minimize the risk of transferable diseases (11.1.2) with all animals. Keepers should be designated to care for only healthy resident animals, however if they need to care for both quarantined and resident animals of the same class, they should care for the resident animals before caring for the quarantined animals. Care should be taken to ensure that these keepers are "decontaminated" before caring for the healthy resident animals again. Equipment used to feed, care for, and enrich the healthy resident animals should only be used with those animals. Standard institutional policy regarding disinfection of equipment used should be followed when working with otters. Care should be taken to ensure all disinfectant is washing from all surfaces before otters are reintroduced into an area.

Also stated in Chapter 6.4, a tuberculin testing and surveillance program must be established for animal care staff, as appropriate, to protect the health of both staff and animals (11.1.3). Depending on the disease and history of the otters, testing protocols may vary from an initial quarantine test, to annual repetitions of diagnostic tests as determined by the veterinarian. To prevent specific disease transmission, vaccinations should be updated as appropriate for otters. TB testing is not routinely done in otters.

AZA Accreditation Standard

(11.1.3) A tuberculin testing and surveillance program must be established for appropriate staff in order to ensure the health of both the employees and the animal collection.

AZA Otter SSP Recommendations for Medical Management

Medical Examinations: It is recommended that all animals have at least a biennial examination and, if possible, an annual examination (see below for exception for *P. brasiliensis*), during which 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.
- 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. Contraceptive hormones should not be used in giant otters because of side effects such as endometritis and pyometra, and the potential result of future breeding inhibition (C.Osmann, personal communication)

Animal Records: Thorough and accurate medical records are essential to learn and understand more about the medical problems of species in *ex-situ* populations. Medical records should be systematic and entries should identify the history, physical findings, procedures performed, treatments administered, differential diagnosis, assessment, and future plans for treatment. A computerized medical record system, which can help track problems and be easily transmitted from one institution to the next, is extremely beneficial. The AZA Otter SSP encourages the use of Med ARKS (International Species Information System, 12101 Johnny Cake Ridge Road, Apple Valley, MN 55124, U.S.A.) as a universal medical record program. Many institutions already use this program, making it easy to transfer information between them. At this time the ZIMS product is being developed to replace current zoological record keeping systems and is considered a suitable substitute.

Vaccination: The following vaccination schedule is recommended by the AZA Otter SSP Veterinary Advisor. 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. This vaccine should be given IM instead of SQ in exotic carnivores for increased effectiveness. 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. 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). 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 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. 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 5: Otter parasite testing protocols

Parasite	Testing protocol
External parasites	Regular inspections during any physical examinations
Internal parasites	<u>Annual fecal examinations</u>: direct smear, fecal flotation, & sedimentation or Baermann techniques. <u>Pre-shipment fecal examinations</u>*:* direct smear and flotation <u>Quarantine fecal examination</u>: 3 negative direct smear results & 3 negative fecal flotation results before release from quarantine. <u>Heartworm ELISA antigen tests:</u> 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 6) provides a list of anthelmintic products that have been used safely in a variety of mustelids:

Table 6: 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 pups 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 pups are listed below with suggested first-step solutions or treatments, and neonatal examination and monitoring guidelines are also provided. See Table 7.

Table 7: 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 volumen substitution can be made with Amynin® (full electrolyte, glucose, amino acid solution offered by Merial), Ringer Lactate, and Glucose 5%, 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 pups (C.Osmann, personal communication). The AZA Otter SSP recommends veterinarians contact one of these companies for their advice on suitable products. Weak pups 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 pups are too weak to suckle, their gastrointestinal tracts are too compromised to digest food and they require immediate veterinary care. 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* pups 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 pup 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 pup'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: Pups 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 pups 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). Pups 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
- 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
- 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 pups 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 pup 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 the use of certain gastric coating agents, such as 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 pups (specifically hand-reared pups), even if they are negative. Pups 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.

6.6 Capture, Restraint, and Immobilization

> **AZA Accreditation Standard**
>
> **(2.3.1)** Capture equipment must be in good working order and available to authorized, trained personnel at all times.

The need for capturing, restraining and/or immobilizing an otter for normal or emergency husbandry procedures may be required. All capture equipment must be in good working order and available to authorized and trained animal care staff at all times (2.3.1).

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. The AZA Otter SSP recommends training animals to receive injections to minimize 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 Isofluorane (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 no longer recommended for use in otters as it may cause liver failure (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).

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.

A. cinereus: 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

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: 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 Isofluorane.

6.7 Management of Diseases, Disorders, Injuries and/or Isolation

AZA Policies: AZA-accredited institutions should have an extensive 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 (2.4.2). 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 (2.3.2), contain appropriate equipment and supplies on hand for treatment of diseases, disorders or injuries, and have staff available that are trained to address health issues, manage short and long term medical treatments and control for zoonotic disease transmission.

> **AZA Accreditation Standard**
>
> **(2.4.2)** Keepers should be trained to recognize abnormal behavior and clinical symptoms of illness and have knowledge of the diets, husbandry (including enrichment items and strategies), and restraint procedures required for the animals under their care. However, keepers should not evaluate illnesses nor prescribe treatment.

> **AZA Accreditation Standard**
>
> **(2.3.2)** Hospital facilities should have x-ray equipment or have access to x-ray services.

AZA-accredited institutions must have a clear process for identifying and addressing animal welfare concerns within the institution (1.5.8) and should have an established Institutional Animal Welfare Committee (AWC). This process should identify the protocols needed for animal care staff members to communicate animal welfare questions or concerns to their supervisors, their Institutional Animal Welfare Committee or if necessary, the AZA Animal Welfare Committee. 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.

> **AZA Accreditation Standard**
>
> **(1.5.8)** The institution must develop a clear process for identifying and addressing animal welfare concerns within the institution.

> **AZA Accreditation Standard**
>
> **(2.5.1)** Deceased animals should be necropsied to determine the cause of death. Disposal after necropsy must be done in accordance with local/federal laws.

As stated in Chapter 6.4, 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 (2.5.1). Necropsies should include a detailed external and internal gross morphological examination and representative tissue samples form the body organs should be submitted for histopathological examination.

AZA Otter SSP Disease Management Recommendations: 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 the AZA Otter SSP recommends that those surfaces be avoided in otter exhibits as far as possible.

Poor coat quality and other factors can lead to 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 Veterinary Advisor conducted a review (G.Myers, personal communication) of all submitted necropsy reports for *L. canadensis*. Her findings (Table 8) indicate that the most frequent causes of *L. canadensis* deaths (excluding neonatal deaths).

Table 8: 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
Anesthetic death	

P. brasiliensis: Causes of death have included: leptospirosis, parvovirus, bronchopneumonia/pneumonia, internal bleeding, gastroenteritis, jejunum invagination, severe inbreeding resulting in inherited thyroid malfunction in pups, parental or older sibling neglect of, or mistreatment towards pups 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).

Illnesses seen 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 made worse by exposure to coarse substrates.

For additional information on how to deal with separated animals, see pages 29 (*A. cinereus*), 31 (*P. brasiliensis*) and 59. If one of the social otters (e.g. *A. cinereus* or *P. brasiliensis*) has to be separated for an extended period, it may be best to house them with another individual. Prolonged isolation for this species is not considered desirable. Extended separation of female *L. canadensis* may lead to reintroduction issues; the reintroduction of these animals should proceed cautiously and follow the standard introduction process (See Introduction/Reintroduction section).

Useful Veterinary References:

Baitchman, E.J. DVM, G.V. Kollias, DVM, PhD. 2000. Clinical anatomy of the North American river otter (*Lontra canadensis).* Journal of Zoo and Wildlife Medicine 31(4):473-483.

Kimber, K. DVM, G.V. Kollias DVM, PhD. 2005. Evaluation of injury severity and hematologic and plasma biochemistry values for recently captured North American river otters (*Lontra canadensis*). Journal of Zoo and Wildlife Medicine 36(3):371-384.

Neiffer, D.L. VMD, E.C. Klein VMD, P.P. Calle VMD, M. Linn DVM, S. P. Terrell DVM, R. L. Walker DVM, D. Todd AHT, C. C. Vice DVM, S. K. Marks DVM. 2002. Mortality associated with melarsomine dihydrochloride administration in two North American river otters (*Lontra canadensis*) and a red panda (*Ailurus fulgens fulgens).* Journal of Zoo and Wildlife Medicine 33(3):242-248.

Rothschild, D.M., T.L. Serfass, W.L. Seddon, L. Hegde, R.S. Fritz. 2008. Using fecal glucocorticoids to assess stress levels in captive river otters. Journal of Wildlife Management 72(1):138–142.

Schettler, E. DVM, K. Müller DVM, G. Fritsch DVM, S. Kaiser DVM, L. Brunnberg, DR., K. Frölich PhD, DVM, G. Wibbelt DVM. 2007. Progressive ataxia in a captive North American river otter (*Lontra canadensis*) associated with brain stem spheroid formation. Journal of Zoo and Wildlife Medicine 38(4):579-582.

Swenson, J. DVM, J.W. Carpenter, DVM, K.S. Janardhan PhD, C. Ketz-Riley DVM, E. Brinkman DVM. Paresis in an Asian small-clawed otter (*Aonyx cinereus*) associated with vertebral and ischial osteolysis caused by a malignant lymphangiosarcoma. Journal of Zoo and Wildlife Medicine 39(2):236-243.

Tocidlowski, M.E. DVM, L.H. Spelman DVM, P.W. Sumner MS, M.K. Stoskopf DVM, PhD. 2000. Hematology and serum biochemistry parameters of North American river otters (*Lontra canadensis*). Journal of Zoo and Wildlife Medicine 31(4):484-490.

Weber, M.A. DVM, M.Garner DVM. 2002. Cyanide toxicosis in Asian small-clawed otters (*Amblonyx cinereus*) secondary to ingestion of loquat (*Eriobotrya japonica*). 2002. Journal of Zoo and Wildlife Medicine 33(2):145-146.

Weisel, J.W., C. Nagaswami, R.O. Peterson. 2005. River otter hair structure facilitates interlocking to impede penetration of water and allow trapping of air. Canadian Journal of Zoology 83(5):649-655.

Chapter 7. Reproduction

7.1 Reproductive Physiology and Behavior

It is important to have a comprehensive understanding of the reproductive physiology and behaviors of the otters in our care. This knowledge facilitates all aspects of reproduction, artificial insemination, birthing, rearing, and even contraception efforts that AZA-accredited zoos and aquariums strive to achieve.

***A. cinereus*: **These otters are non-seasonal and thought possibly to be spontaneous ovulators (Bateman et al. 2009). The estrous cycle lasts 30-37 days, with breeding occurring year round. Some facilities report this cycle extending to every few months with older animals. Estrus lasts from 1-13 days. Behavioral signs of the onset of estrus may include increased rubbing and marking. Sexual behavior has been observed in pups as young as 6 months, with breeding behavior having been noted in animals (males and females) as young as 1½ years. Successful breeding has been reported for 2.1-year-old females and 2.8-year-old males. There do not appear to be any significant environmental cues that are involved with the onset of estrus. Breeding pairs have been introduced at various ages and have been together for varying lengths of time before successful breeding occurs. It has been reported that pups from previous litters have interfered with copulation, but their presence had no bearing in any other way (Lombardi et al. 1998).

Recent work has shed light on litter intervals; Bateman et al. (2009) report: "In one female having three consecutive pregnancies during [their] study, the interval between the first parturition and subsequent progesterone increases owing to the next pregnancy was 169.25±11.15 days. This female was observed nursing her pups from the first pregnancy for the first 122 days of this intergestational time period."

Breeding pairs need to establish a bond for successful reproduction. The male pursues the female in courtship and most breeding occurs in shallow water. A single copulation can last from 5-25 minutes. Courtship behavior has been recorded from 1-3 days, at one-month intervals. Gestation is roughly 60-74 days (67-77 range, average 71.17±1.49 days reported by Bateman et al. 2009).

Pseudopregnancies do occur in this species, including in females housed in single sex groups (5 of 6 females housed together exhibited pseudopregnancy) (Bateman et al. 2009). Bateman et al. (2009) report: "…a mean duration of pseudopregnancy of 72.45±1.37 days (range: 62–84 days). The average interval length between sequential pseudopregnancies and/or pregnancies was 39.86±3.86 days (range: 17–92 days) in paired females and 134.50±48.94 days (range: 62–279 days) in the single gender group."

The sire plays a very active role in rearing the pups and should not be removed prior to their birth. Male behaviors include nest building, carrying pups, and bringing food to the pups during weaning.

Table 9: *Ex-situ* population breeding parameters of *Aonyx cinereus* in North American zoological facilities 1980's and 1990's (Reed-Smith & Polechla 2002, Bateman et al. 2009)

Aonyx cinereus	
Estrus cycle	30-37 days. Polyestrual with breeding occurring year around
Estrus length	1-10 days
Ovulation	Likely spontaneous ovulators (Bateman et al. 2009)
Copulation frequency	Several times a day.
Copulation duration	1-30 minutes, varied.
Copulation position	Dorsal/Ventral most common, also ventro/ventral.
Copulation location	In the water and on land
Copulation initiated by	Varies amongst groups; in some it is initiated by male only, in others both initiate.
Age at 1st breeding	Unknown
Breeding behavior	Increased rubbing, marking.
Gestation	Gestation ranges between 67 and 74 days; pseudopregnancies lasted 62 – 84 days (Bateman et al. 2009). No delayed implantation (Bateman et al. 2009)
Pair management	Most facilities started out with a pair when 1st litter born. Pair left together all of the time
Group management	Some reports of harassment of new pups by older pups (too much play), one report reported cannibalism of pups by dam. Most leave all animals together.
Signs of parturition	Some weight gain, more time spent in nestbox.
Pupping boxes	Wooden boxes, hollows under logs & burlap bags have been used.
Contraception	MGA implants in females.

A. capensis**:** This species does not appear to have a specific reproductive season (Mead 1989). Breeding in the northern hemisphere has been observed in November, January, March, and April (R.Meyerson, personal communication), with pups born in January-March and June-September (R.Meyerson, personal communication). Gestation length ranges from 63-80 days depending upon the source (Estes 1989; Reed-Smith & Polechla 2002; R.Meyerson, personal communication). In one *ex-situ* population breeding situation, receptivity by the female lasted one day (Personal communication); in another, breeding occurred for 2-3 days (R.Meyerson, personal communication). Generally, 1-2 days before a female is receptive, the male will start following her around. All *ex-situ* population pairs have shown an increase in the level of interactive play behavior for several days before and after breeding. All observed copulations have occurred in the water.

Table 10: *Ex-situ* population breeding parameters of *Aonyx capensis* in North American zoological facilities 1980's and 1990's (Reed-Smith & Polechla 2002)

Aonyx capensis	
Estrus cycle	Breeding occurred in Jan, Apr, Nov & Dec for 4 litters produced.
Estrus length	Peak receptivity lasted 1 day; day before & after consisted of play & close following
Copulation frequency	Several times during the 2nd day of breeding behavior
Copulation duration	Not documented.
Copulation position	Not documented.
Copulation location	In the water.
Copulation initiated by	Male, females would only cooperate for 1 day.
Age at 1st breeding	Males from 2 yrs.10 mos.; Females from 4 yrs. 2 mos.
Breeding behavior	Females fought when one was in heat. Signs of estrus shown by male behavior
Gestation	80, 103 days
Pair management	Breeding was opportunistic. Male kept away from pups by female or separated 1 week prior to expected parturition.
Group management	1.2 housed together on exhibit during the day; 0.2 given 2 dens at night
Signs of parturition	Females gained weight particularly in fold between foreleg and body.
Pupping boxes	Females did not want bedding in their boxes. Given only one box, no problems.
Contraception	Two males have been vasectomized due to small gene pool. Females medicated for contraception.

L. canadensis: These otters are seasonal breeders. Females mature reproductively as early as 12-15 months (rare reports of successful breeding at this age) to two years of age (typical). They are believed to be induced ovulators and experience delayed implantation (Chanin 1985; Reed-Smith 2001, personal observation). Recent evidence suggests that this species also may be capable of spontaneous ovulation (Bateman et al. 2009).

There is evidence that breeding season varies somewhat with latitude (Reed-Smith 1994, 2001; Bateman et al. 2005, 2009) and also may be influenced by seasonal availability of food resources (Crait et al. 2006); however, the authors of the one study (Crait et al. 2006) speculating on the influence of food availability acknowledge there could have been other things occurring, and their sample size was small. In general, breeding occurs in late spring (March-June) at northern latitudes and between November-February at more southern latitudes, with a gradient in between (Reed-Smith 2001). The estrus period lasts approximately 42-46 days, unless mating occurs (Chanin 1985). Bateman et al. (2009) found that "…peaks in fecal estrogen values occurred only during the defined breeding season from December to March". They also report, "…the estrus phase of their cycles [N=11] was observed just once per year with an average duration of 15.33±1.98 days (range: 6–54 days). The average duration of estrus elevation coincident with observed breeding (n=4) was 22.00±1.22 days (range: 19–24 days)." During this time, observations of *ex-situ* populations suggest peaks of maximum receptivity are roughly 3-6 days apart with intervals of only mild receptivity during which the female may completely reject the male (Liers 1951; Reed-Smith 2001). The work done by Bateman et al. calls into question the estrus duration of 42 to 46 days traditionally cited; this is an area that should be researched further. Worth noting is the slightly longer estrus (21-23 days) reported in breeding versus non-breeding females (14-17 days); also the widely varying range of estrus duration (6-54 days) reported in the Bateman et al. study.

More recently, Bateman et al. (2009) have reported some additional interesting results from fecal hormone studies:

> "In the observed pregnancies and pseudopregnancies (n= 12), the date of initial progesterone increase ranged from September 4 to January 14, and the timing was not correlated (r=0.53, P>0.05) with the female's geographic latitude (range: 27–41°N) at the time of the pregnancy or pseudopregnancy. However, the date of the

progesterone increase was correlated (r=0.66, P<0.05) with the female's geographic latitude at the time of her own birth (range: 27–42°N)."

The finding of a possible genetic component tying the timing of progesterone elevation in pregnant and pseudopregnant females to the female's birth latitude (instead of to their geographic location) is significant and requires further study. This impacts *ex-situ* population management practices (transfers of animals to create breeding pairs) and the outcome of wild otter translocation/reintroduction projects (females may be giving birth too early or too late).

Males mature sexually at about two years; the production of male spermatozoa begins at this age. The male's testes begin to enlarge and spermatozoa production begins sometime in October/November, (or earlier at more southern latitudes) and testes remain distended until the end of the breeding season (Liers 1951; Reed-Smith 2001). Bateman et al. (2009) have shown that male testosterone levels increase seasonally to coincide "…with the increasing amount of daylight occurring after the winter solstice." Testosterone levels peaked at different times of the year corresponding with the male's latitudinal location; "As latitude increased, peak testosterone values appeared to occur later in the calendar year" (Bateman et al. 2009). They found that "for all NARO males, testosterone levels were elevated above baseline for an average of 101.8±78.97 days with peak levels being maintained for 25.50±7.51 days."

Females may show some, all, or occasionally none, of the following signs of estrus: increased marking of their territory, vulvular swelling, a slight pinking of the vulva area, increased rubbing, rolling and allo-grooming, increased interest in the male or the male's quarters, increased interaction between the female and the male to include chasing, tumbling, mutual grooming, sleeping together (obviously will only be apparent in pairs that do not normally do this), chuckling to the male, genital sniffing by the male of the female and vice-versa, and copulation. If male and female *L. canadensis* are housed alone, the pair should be introduced for breeding after the first signs of estrus appear or when the female shows unusual interest in the male's scent or enclosure. Absent any obvious signs, introductions should begin roughly four weeks prior to typical onset of estrus at the facilities latitude. Some facilities have reported a small amount of estrus-associated bleeding from the vulva, while others have not seen this. This is an area that requires further research. Female river otters also appear to be prone to urogenital infections, which frequently cause a milky, milky-blood-tinged, or slightly off colored discharge, which has been interpreted as a possible indicator of estrus or imminent parturition. If this kind of discharge is seen, the female should be closely observed and the condition monitored by a veterinarian.

Although copulation generally takes place in the water, it also can take place on the land. The copulatory act is vigorous, noisy, and can be lengthy with intromission lasting up to 60 minutes or more. A pair will copulate repeatedly over a period of an hour or two, then rest or forage for several hours before starting again. Copulation generally occurs several times over successive days. Copulations should be at least several minutes in duration to be successful. Short and/or infrequent copulatory bouts are generally not successful (Reed-Smith 2004b). Breeding activity may resume after a lull of 3-6 days throughout the course of a female's receptive period. During copulation, the male holds the female's scruff with his teeth, and positions the posterior part of his body around and below the female's tail (Liers 1951; Toweill & Tabor 1982). If the female is not receptive, or interested, she may roll on her back and paw at the male, nip and scream at him, or bite him and then run away.

Total gestation lasts from ~317 to 370 days reported by Liers (1951) and Reed-Smith (2001) or 302-351 (average 333.3 ±15.7) days reported by Bateman et al. (2009); actual gestation is about 68-73 days (average 71.67±1.48) (Bateman et al. 2005, 2009). The parturient female may exhibit a number of different signs including: increased 'nest' building, swollen mammae, aggression towards exhibit mates or keepers, depressed appetite, frequent floating in the pool, refusal to leave the nest box, restlessness or lethargy. No pre-partum discharge has ever been noted. Parturition may occur from November through May, however, the peak time appears to be March through May in the northern latitudes (40-60˚N) and late December through February at more Southern latitudes (23-30˚N).

Estrus occurs soon after parturition lasting the same 42-46 days (recent hormonal work sets estrus as 15 to 22 days [Bateman et al. 2009]). Hamilton and Eadie (1964) record estrus as occurring not long after parturition. Most zoos that have observed postpartum estrus see behavioral signs 1-2 weeks after parturition.

The AZA North American River Otter PMP recommends that facilities interested in breeding should exchange one individual if the pair has been together since a young age and have not been successful at breeding. Based on previous hormone monitoring, the time of year in which each river otter's breeding season occurs is highly influenced by its geographic location/latitude (Bateman et al. 2005, 2009).

Consequently, a possibility exists that some animals moved between widely varying latitudes may be physiologically out of synchrony and would require at least one breeding season to adapt physiologically to their new environmental cues, which are important for signaling the start of breeding season. This should be factored in when making transfer recommendations, but should not limit transfer options when creating new breeding pairs. The North American River Otter Husbandry Notebook (Reed-Smith 2001) provides greater detail on the breeding strategy and reproductive physiology of this species, and the AZA Otter SSP reproductive advisor (Helen Bateman, C.R.E.W., Cincinnati Zoo) is involved in on-going research.

In both NARO and ASCO, additional research is needed to improve endocrine monitoring of estrogen metabolites to further address these questions about ovarian cyclicity and ovulatory mechanisms.

Table 11: *Ex-situ* population breeding parameters of *Lutra canadensis* in North American zoological facilities 1980's and 1990's (Reed-Smith & Polechla 2002, Bateman et al. 2009)

Lutra canadensis	
Estrus cycle	Monoestrus; can occur Nov-Jun based on latitude Post-partum elevations in estradiol levels occur 2 – 38 (ave. 19 ± 8.06) days after parturition (Bateman et al. 2009)
Estrus length	42-46 days unless mating occurs; "…the estrus phase of their cycles [N=11] was observed just once per year with an average duration of 15.33±1.98 days (range: 6–54 days). The average duration of estrus elevation coincident with observed breeding (n=4) was 22.00±1.22 days (range: 19–24 days)." (Bateman et al. 2009) Receptivity peaks roughly 6 days apart have been reported but not reflected in Bateman study.
Ovulation	Unclear if they are induced ovulators (Bateman et al. 2009), but suspected; may also ovulate spontaneously (Bateman et al. 2009)
Copulation frequency	Several times a day
Copulation duration	20-45 minutes, varied. One >60 min. reported
Copulation position	Dorsal/ventral most common, also ventro/ventral.
Copulation location	Most frequently in the water, also seen on land
Copulation initiated by	Both. Female advertises, cooperates only when she is ready. She may initiate with invitations to play chase.
Age at 1st breeding	Sexually mature by 2 yrs. Several 2 yr. old males & a 1.5 yr. old female have bred successfully.
Breeding behavior	Female may rub, mark or vocalize to advertise; male/female may initiate w/ play, chase, splashing, genital sniffing, or "butterfly stroke".
Gestation	Total gestation: 332-370 days, documented for 12 litters. 285-380 (Liers 1951); 302-351 (ave. 333.3 ±15.7) days (Bateman et al. 2009) Actual gestation: 68 – 73 (ave. 71.67±1.48) days (Bateman et al. 2009)
Pseudopregnancy	Pseudopregnancies seen with and without breeding and does not always result when breedings are unsuccessful. The period of elevated progesterone ranges from 68 to 72 days as in true pregnancies. (Bateman et al. 2009)
Pair management	Most facilities separate the male from the female, for his safety. A few leave the male in exhibit with female and she keeps him away from pups until they can swim well. One facility offered pair selection; females showed preference for certain males.
Group management	Sire can be reintroduced to female and pups after pups are swimming well. Generally done at about 3-6 months.
Signs of parturition	Females may show visible weight gain, teats may show through coat, increased nesting behavior, change in attitude to keeper &/or male. She may go off food as parturition nears.
Pupping boxes	Pupping boxes should be filled with dry bedding (straw or hay). A choice of birthing boxes should be available.
Contraception	MGA implants and PZP treatment in females. A few males have been neutered.

L. maculicollis**:** Schollhamer (1987) reported that females came into estrus for the first time at about two years of age, but females typically do not conceive until they are three years old. Cycles vary between individuals, but average about 45 days, and estrus generally lasts 5-7 days. There is a postpartum estrus 2-3 weeks after parturition if pups are pulled or die soon after birth.

Males may attempt breeding at 1-2 years of age, but typically are not successful until they are 2-3 years old (Schollhamer 1987). Mating occurs in the water (Schollhamer 1987; R.Willison, personal communication) and involves the male neck biting the female and clasping her with his fore and hind limbs. Copulatory bouts may last up to 45 minutes, generally occur repeatedly over the course of several days (R.Willison, personal communication), and at any time during the day or night. There are no vocalizations associated with copulation in this species in zoos (R.Willison, personal communication) or in the wild (Reed-Smith in prep.). The rate of conception is increased if the male breeds the female for the entire 5-7 day cycle of a typical female; the conception rate is poor if the male only breeds the female for 2-3 days (Schollhamer 1987).

Gestation is roughly 60-63 days (R.Willison, personal communication). *Ex-situ* population births in North America have occurred in January/February and April (R.Willison, personal communication). Births in the wild were recorded during September, based on one year of observation in Tanzania (Proctor 1963); ongoing research in Tanzania substantiates this observation with breeding observed twice in June (2006 and 2007), which would result in late August to mid-September births. However, work by Bateman et al. (2009) and observations of half-grown animals throughout the year in Tanzania indicate that births may occur anytime with a peak during August/September. Further research is required.

***P. brasiliensis*:** Estrus generally occurs every three months, typically lasting 5-7 days with a range of 1-11 days (Autuori & Deutsch 1977; Trebbau 1978; Hagenbeck & Wünnemann 1992; Wünnemann 1995b; Marcato de Oliveira 1995; Corredor & Muñoz 2004; Sykes-Gatz 2005, 1999-2006). The pair will begin exhibiting an increase in rough play and chasing behaviors a few days prior to breeding. These behaviors continue throughout the estrus period (S.Sykes-Gatz, personal observation). Copulation generally takes place in the water, but also may occur on land. The copulatory act is typically repeated several times a day over the course of 5-7 days and may last from 30 seconds to 30 minutes or more (Hagenbeck & Wünnemann 1992; V.Gatz, personal observation; S.Sykes-Gatz, personal observation).

There can be an estrus 5-7 days postpartum/post-loss of a litter that lasts for 3-5 days (Hagenbeck & Wünnemann 1992; Wünnemann 1995b). Delayed implantation occurs in zoos (Flügger 1997; Corredor & Muñoz 2004; Sykes-Gatz 2005, 1999-2006; V.Gatz, personal observation). Generally, this species produces one litter annually in the wild. In zoos, false or pseudo pregnancies are not uncommon in this species (Sykes-Gatz 2005). Gestation ranges from 64-71 days, and in one case a gestation of 77 days occurred in a 9-year-old, although this was unusually long for this female (Autuori & Deutsch 1977; Trebbau 1978; Hagenbeck & Wünnemann 1992; Wünnemann 1995a,b; Corredor & Muñoz 2004; Sykes-Gatz 2005; V.Gatz, personal communication). Often the female's mammary glands become enlarged 30 days prior to parturition and the vulva may become swollen about 14 days prior. In zoos and aquariums, inter-birth intervals of 63 days, 74 days, and 94-103 days have been recorded in cases of pairs that lost litters at birth (Hagenbeck & Wünnemann 1992; Wünnemann 1995b).

Sexual maturity is reached at roughly 2 years of age. *Ex-situ* population records show that at 2 years and 5 months, females can come into their first estrus, mate, and bear a litter at the age of 2 years 7 months. Males can mate at 2.5 years of age, with their first litter born when they are 2 years and 8 months. Due to limited records, it is not known if this is the earliest age at which giant otters can become sexually mature and bear litters. There is some indication that at least some giant otter females, from the age of 10-11 years, may experience a slowing or end to their reproductive capabilities. Females of this age may alternatively experience health problems or other difficulties during gestation and parturition. Whether this is due to their advancing age or a high number of previous litters is unknown. An 18 years and 9 month old male is the oldest successful sire on record (Sykes-Gatz 2005 & 1999-2006, V.Gatz, personal communication).

The International Giant Otter Studbook Husbandry and Management Information and Guidelines (Sykes-Gatz 2005) should be consulted for greater detail on management of this species, particularly their requirement for isolation and pupping den specifications.

Separation of Sexes/Conspecifics: If it is necessary to separate animals for reasons associated with reproduction (e.g., to promote or prevent it), a holding area connected to the exhibit is recommended; ideally this should include a pool with clean water available at all times, proper lighting, a sleeping or den box, enough floor space for grooming and drying areas, and at least one nest box that is heavily bedded to allow excess moisture to be removed from the animals' coats (Lombardi et al. 1998; Reed-Smith 2001). Holding pens should have non-climbable sides. If chain-link barriers are present, the sides should be covered with lexan or similar material to prevent animals from climbing too high and falling. See species-specific recommendations below.

A. cinereus: Females become very aggressive prior to parturition and remain so post-parturition. It is not necessary to separate them from the male or older siblings. It is necessary to provide multiple nest boxes. The sire plays a very active role in rearing the pups and should not be removed prior to their birth. Male behaviors include nest building, carrying pups, and bringing food to the pups during weaning. Access to pools and water sources should be strictly monitored to prevent newborns from drowning.

A. capensis: Pregnant females should be offered nest box choices and separated from the male to give birth (R.Meyerson, unpublished data). To date, records indicate two institutions have successfully bred this species in North America. One facility had one male with two females; both females bred and produced offspring. The other facility housed a pair. The animals were housed together 24 hours a day; females were separated to give birth at both facilities (Reed-Smith & Polechla 2002; R.Meyerson, personal communication). The male can be reintroduced to the female and pups when they are swimming well.

L. canadensis: There seems to be a mate preference for breeding, with some females showing a definite preference for particular male and lack of interest in others when they have a choice. Some successful zoos separate pairs for several months prior to the breeding season, introducing them every few days once the female's estrus begins. Others offer multiple mate selections to the females, and others have been successful keeping single pairs together year around. An extensive *ex-situ* population study (N=13.14 animals) attempted to determine breeding associated behaviors in zoos and aquariums across the species over two years. The study's behavioral results show an increase in pair association, mutual grooming, and extended copulation in those pairs that reproduced successfully (N=3), but was inconclusive otherwise (J.Reed-Smith, data in preparation).

In the wild, males do not participate in pup rearing (Melquist & Hornocker 1983; Rock et al. 1994). In zoos and aquariums, parturient females should be given privacy (particularly for primiparous females) and nest box choices supplied with plenty of dry bedding (all females). Males have been successfully left in the exhibit with parturient females in large exhibits that provide numerous visual barriers and allow the male to stay out of the female's sight. In all other cases, the male and female should be separated prior to the birth to prevent injury to the male or neglect of the pups by the female. In multi-female groups, other females may also need to be separated from the parturient female. When separated, the male or non parturient female should not be required to pass the parturient female's den to enter the exhibit; if this cannot be done, the other animals should be removed entirely from the exhibit or the female sequestered away until she deems it time for the pups to meet the male (see below). Males can be reintroduced to the female and pups once they are swimming proficiently, as early as 60-75 days or more typically by 80 to 90 days (Reed-Smith 2001).

Actual gestation is calculated at roughly 68-73 days (Bateman et al. 2009.); pair separation should occur either when the female becomes aggressive towards the male or roughly 10-14 days prior to anticipated parturition date. Due to this species' delayed implantation and total gestation time of >10 months, it is often difficult to anticipate delivery date, particularly for primiparous females. In these cases, staff should base their management decisions on the female's behavior. If she becomes aggressive to the male or other exhibit mates, begins to show excessive nesting behavior, or spends increasing amounts of time in her nest box, the pair should be separated. It is important to remember that the female should be monitored for health issues during this time, as these behaviors also can be signs of illness.

Management change should be scheduled so that they do not interfere with the birth and rearing of the pups. Any modifications to the exhibit should be finished several months prior to possible pupping season. Denning/nest box choices should be introduced at least one month prior to possible pupping season. Changes in management routines, e.g., closing the female in holding at night, closing her in holding alone, etc. should be introduced to the female at least a month prior to the possible parturition period to allow her time to become comfortable with the new routine. If the male will have to be removed from the exhibit entirely, this should be done several weeks prior to possible parturition to allow the female a period of adjustment.

Generally, the best way to handle pair separation is setting up the female in off-exhibit holding (providing there is adequate space). Once the pups are old enough to begin swimming lessons (some females begin this instruction as early as 30 days, more typically at 40-50 days), the family group and the male can be alternated on exhibit. When the pups are swimming well, after about three months, the male can be introduced to the family group. This should be handled, as with any introduction, via olfactory, visual, and then physical introduction to the female alone first, and under controlled circumstances as far as possible (see Introduction/Reintroduction).

L. maculicollis: Females should be separated from the male at about gestation day 55 (gestation ranges from 60-63+ days calculating from the day of last observed breeding) (Schollhamer 1987), or when she shows signs of aggression towards the male (R.Willison, personal communication). Males should be separated from a pregnant/nursing female unless the exhibit is large enough for him to stay out of her line-of-sight. When separated, he should not be required to pass the female's den to enter the exhibit; if this cannot be done, he should be removed entirely from the exhibit or the female sequestered away until she deems it time for the pups to meet the male. Females with pups are more of a danger to the male than typical males are to the pups. Males can be reintroduced to the female and pups once they are swimming proficiently and eating on their own, typically when the pups are roughly four months of age (R.Willison, personal communication).

Females should always be given a choice of denning sites with bedding provided for them to use if it is wanted. Schollhamer (1987) states that at Institutin A female spotted-necked otters did not use bedding of any kind. Kruuk (1995) references the presence of soft, leafy substrate or pebbles in most dens he or other researchers located. Brookfield Zoo used a nest box made from molded plastic and fiberglass measuring 68.6cm long x 51cm wide x 38.1cm high (27" x 20" x 15") with holes drilled in the bottom for drainage. The box was placed 4cm (1.5in) off the floor, and was accessed via a drop-guillotine door 25.4cm high x 20.3cm wide (10" x 8") (Schollhamer 1987).

P. brasiliensis: This species lives in family groups with pairs and older offspring jointly raising new pups. Therefore, a pair should never be separated during pregnancy or pup rearing. Typically, animals should not be separated from the family group unless health problems, change in social status, or family friction develops. Removal of any member of a group during pup-rearing, or close to parturition, will likely cause litter loss. Animals separated for extended periods should be put through a standard introduction (see Chapter 4, section 4.3), including visual, acoustic, and olfactory contact at first, and then physical contact (Sykes-Gatz 2005). Even a few days of separation have been known to be long enough to cause difficulty, such as serious fighting, when reintroduction was attempted (K.Lengel, personal communication).

Secondary accommodations should be provided for giant otters to allow for the temporary separation of family members if needed. These secondary enclosures should provide husbandry conditions similar to primary enclosures.

Nursery Groups: Typically, nursery groups of neonates are not seen in river otter species. The following species-specific information is available:

A. cinereus: All otters of a family group take an active role in caring for the young. It is not uncommon for the sire and older offspring to be involved in all behavioral activities of the mother and her newborns.

A. capensis: Nursery groups are not typical for this species.

L. canadensis: Nursery groups are not typical for this species. Helper otters have been reported from the wild. In these cases, a female with partially grown pups is accompanied by another adult female (Rock et al. 1994; R.Landis, personal communication). There are reports of two adults with young animals, but the relational composition of these groups is unknown (Reed-Smith 2001).

L. maculicollis: Nursery groups are not reported for this species, but further research is needed. Adolescent groups (roughly one year or older) have been reported (Reed-Smith in prep.).

P. brasiliensis: As with the Asian small-clawed otter, generational groups are typical in the wild; true nursery groups are not reported for either species.

Separation of Mother and Offspring: The timing of mother-offspring separations can have long-term effects on the development of otter pups and on the reproductive success of adults. The following species-specific information is available:

A. cinereus: Adolescents are not forced from the group. In zoos and aquariums, it is necessary to remove older offspring, as the group size can become quite large in a year, leading to aggression resulting from over-crowding. Typically, the age at which older pups should be removed varies with the size of the exhibit and compatibility of the group.

A. capensis: The timing of emigration is unknown in the wild, but emigration of sub-adults at some point is presumed.

L. canadensis: Pups can be removed from the dam when weaned, if absolutely necessary. It is preferable that they be left with the family group until they are at least 8-9 months of age (Reed-Smith 2001) or six months old at a minimum. In the wild, pups will generally leave the female when they are 9 months to over one year of age (Melquist & Hornocker 1983; Melquist & Dronkert 1987).

L. maculicollis: No specific information on emigration is available, but pups should be left in the family group for at least 6-9 months and be removed before reaching sexual maturity. Roving groups of what appear to be young animals, possibly dispersing, and pups remaining with their mothers (or at least utilizing the same core area simultaneously for one year) has been reported in the wild (Reed-Smith in prep).

P. brasiliensis: Pups from previous litters up to the age of about two years generally stay with the family group (Schenck & Staib 1994); after this time, they emigrate to set up a new family group. In the wild, Duplaix (2002) reports that sub-adults may leave the family group after 2 years, before birth of the next litter, or be pushed out by the adults with a fight. Staib (2002) found that animals dispersed at the age of 2-3 years, and separations were gradual, without aggressive behavior. In zoos and aquariums, offspring should be left with the parents for at least the first 6-12 months of life, but preferably they should be left together until they reach sexual maturity at approximately 2 years of age (Sykes-Gatz 2005, 1999-2006; Corredor & Muñoz 2004; G.Corredor, personal communication; V.Gatz, personal communication). This provides a more natural social structure, and allows older siblings to gain experience helping to rear pups, which is highly beneficial towards developing their future parenting skills. Caution should be taken, as in one case three *ex-situ* population-born otters between the ages of 6.5-8.5 months were suspected of competing for milk with their younger siblings. This behavior persisted over a two-month period, causing the death of a younger sibling and the necessary removal of the emaciated survivors for hand- rearing (Flügger 1997). In two other cases, sub-adults were suspected to have caused litter loss because of their over-zealous play with, and attention to, their younger siblings (Flügger 1997; G.Corredor, personal communication). However, experience has shown that removal of any member of a giant otter group during pup-rearing or close to parturition will likely cause litter loss due to the excessive stress caused to the parents by this human disturbance and unnatural social structure change (Flügger 1997; Sykes-Gatz 2005, 1999-2006). If it is necessary to remove a group member from a breeding pair, it should be done when the mother is not pregnant or at the latest in the early stages of pregnancy. Pups younger than 6 months of age should not be removed.

Reproductive Hormone Tracking: Research utilizing techniques to identify reproductive state in these species is ongoing. At this time, it appears that ELISA protocols for testing hormonal secretions in fecal samples is successful in determining pregnancy in Asian small-clawed and North American river otters (Bateman et al. 2005, 2009). The reproductive physiology advisor for the AZA Otter SSP should be contacted for more information.

Pseudopregnancy has been reported for most otter species and is an area that requires further research. For information on the status of current research into this and other reproductive physiology, behavioral, and health issues, contact the current AZA Otter SSP Chair for the most recent information.

Facilities for Reproduction: All expectant females should be provided with nest box choices that are located away from pools, and these should be well stocked with dry bedding. The size of these dens should allow ample room for bedding, pups, and for the female to turn around (*A. capensis*, *L. canadensis*, and *L. maculicollis*). Highly social species (*A. cinereus* and *P. brasiliensis*) should be provided with a nest box or pupping den that allows enough room for the entire group. See species-specific information below and Sykes-Gatz for dimensions and recommendations on pupping dens and nest boxes.

A. capensis: Nest boxes 8-10cm (3-4in) wider and taller than those used for *L. canadensis* (see below) are suitable for this species. Nest box choices and plenty of bedding should be provided 2-3 weeks before expected parturition date to allow the female to become comfortable with them. At this stage, females show a weight gain in the axillary region (R.Meyerson, personal communication). Some females prefer to pup without the bedding, and will remove it from their nest box; in these cases, allow the female her choice.

L. canadensis: Due to delayed implantation (also known as embryonic diapause), it is difficult to determine when a female is near parturition; therefore, close attention should be paid to her behavior changes, appetite, and physical appearance. These may include, but are not limited to: aggression towards exhibit mates or keeper staff, refusal to leave holding or her den, increased or decreased appetite, obvious teat development, slow movements, more frequent floating in the pool, and lethargy.

Parturition boxes should be at least 50.8cm long x 45.72cm wide x 38.1cm high (20" x 18" x 15"), be large enough for an adult animal to move around in, and large enough to accommodate the pups. Slightly smaller boxes with entrance foyers have been used successfully. This box type allows the female to be secluded in a location near the pups but not actually with them. Females should be offered denning choices for parturition and den choices of different sizes to allow for the growth of the pups.

Dimensions for a sample nest box are as follows: total width = 68.58cm (27in), chamber width = 48.26cm (19in), chamber depth = 45.72cm (18in), entrance foyer = 20.32cm (8in), entrance diameter = 16.5cm (6.5in), and 26.67-38cm (10.5-16in) high, with a 16.5cm (6.5in) height at the end of the ramp, and a 36.83cm (14in) height at the entrance to the ramp. The top is hinged on one side for easy lifting and cleaning. The ramp floor is made of wire mesh and the chamber floor should have drainage holes.

L. maculicollis: Spotted-necked females should be separated from exhibit mates prior to parturition at roughly day 55 of a 60-63 day pregnancy (Schollhamer 1987), or when the female begins to show a tendency to keep the male or other group members away from her denning area (R.Willison, personal communication). Generally, females give birth to one pup, sometimes twins (Schollhamer 1987; R.Willison, personal communication).

Secured sleeping dens (R.Willison, personal communication) or nest boxes that are 27" long x 20" wide x 15" high (68.6cm x 51cm x 38cm) have been successful (Schollhamer 1987). Typically, females do not use any nesting material, however, bedding should be offered to all females.

Females should be given rubber tubs in which to swim just prior to parturition (1-3 days), and for the first two months or so of the pup’s life. The female will begin to bring the pup(s) out of the denning box when it is roughly 3-4 weeks old; at this point, she should start teaching pups to swim by placing them in the water tub for a few minutes at a time. It is important that the water level in tubs be kept high (which allows pups to hang on the lip), or that tubs/pools have a slopping ingress and egress so pups can get out of the water. While females are typically very vigilant, pups have suffered from hypothermia from being left too long in the water (Schollhamer 1987).

P. brasiliensis: Giant otters (especially mothers) are susceptible to any human disturbance, especially within the surroundings of the natal den, and to discomfort created by enclosure variables (see in Sykes-Gatz 2005; 2.1 & 6.7; Sykes-Gatz & Gatz 2007). Several steps have been recommended to increase the comfort of the reproductive pair and older siblings, and improve pup-rearing success (Sykes-Gatz 2005):

- Build a positive keeper-animal relationship and allow only familiar staff to work with the otters after parturition
- The provision of food and clean water should be accomplished with minimal disturbance to the otters during pup rearing.
- Cleaning should be minimal and not disruptive to the otters.
- Reduce stress as far as possible, including loud sounds, unfamiliar people, and the introduction of anything new to the exhibit.
- Prohibit visitor and zoo staff (other than immediate caretakers) access to the enclosure area.
- Provide multiple nest box choices located in separate locations to allow parental choice according to their comfort level.
- Provide appropriate substrate and exhibit conditions to include the recommended land/water ratio, substrate depth, digging opportunities, and dry substrate conditions.
- Isolation of the natal den and limitation of all human activity in the vicinity at and prior to the birth is very important.
- Monitoring of the natal den and early pup rearing should be done from a hidden location or carried out via audio and video monitoring equipment with infrared capability.

Exhibits should be provided with multiple den sites; these can include both natural (e.g., dug by the otters) and man-made dens. Dens are often 48-102ft^2 (4.5-9.5m^2) in size. Ideally, pupping boxes just large enough to hold the adults, older siblings, and pups should be placed within the dens to allow the parents choices and maximum privacy. Dens should be provided in locations where the animals are removed from all disturbances (Sykes-Gatz 2005). Nest box temperatures should stay above 20°C (68°F). Den area temperatures (where nest boxes were located) were increased to 22-23°C (71.6-73.4°F) during pup-rearing at one institution (Flügger 1997). *P. brasiliensis* in *ex-situ* populations have been observed to have a low heat tolerance (Carter & Rosas 1997; S.Sykes-Gatz & V.Gatz, personal observation), and pups can be very susceptible to overheating or becoming too cold. Very young pups especially do not thermo regulate well (Read & Meier 1996). Pups <5 months of age should not be exposed to air temperatures below 15°C (59°F), and pups >5 months of age should not be exposed to air temperatures below 10°C (50°F). Parents should be prevented from taking pups outside if temperatures fall below these parameters. Precise recommendations for enclosure and den design are provided by Sykes-Gatz (2005). This publication is available on the Otter Specialist Group website (www.otterspecialistgroup.org) and going to the Otters in Captivity Task Force under Library.

In at least the first days after parturition, dams have been seen to be a little protective of the pups when the sire tries to become involved with them. This is not abnormal behavior. Soon afterwards, the sire will become equally involved (and his involvement will be accepted by his mate) in the care of the pups. Under normal situations, it may appear that both parents and older siblings sometimes treat their pups a little roughly. This kind of behavior may be carried out whether otters are in the nestbox, on the land, or in the water. This is especially evident when parents or older siblings are teaching pups to swim. This seems to be normal behavior for giant otters. However, the situation should be closely monitored to ensure that parents and older siblings are not actually too rough with their pups, as this is abnormal. Starting at 2-3 weeks of age, parents will push pups under the water then let them go to resurface on their own or with help. This may be repeated several times to teach them to submerge (Autuori & Deutsch 1977). Parents and older siblings have been known to teach *ex-situ* population-born pups to submerge starting at 2-6 weeks of age by holding the pup with their front feet and rolling over sideways 360° a few times; this has been called ‘Eskimo rolling’ (Sykes-Gatz 1999-2006). Pups also may be gently pushed or pulled into the water to encourage swimming.

The following is a list of parental and older sibling behaviors observed *ex-situ* that often resulted in pup death and may be a result of sub-optimal environmental or rearing conditions:

- Pups handled, carried, or moved to pools or new nest boxes very frequently. Generally, pups < 2 weeks of age should not be taken into pools, if this occurs it is rare. Pups 2 weeks old or older should not be taken into pools more than 1-2 times per day. Older pups may tolerate more frequent moving, generally no more than 3 times a day. In general, pups should not be moved to new nest boxes on a daily basis or at most more than once or twice a day. Frequent movement of pups should be closely monitored without disturbance to the parents.
- Too frequent entering of the nest box by the parents, e.g., 1-3 times per hour is normal, more can be abnormal.
- Excessively forceful pushing or throwing of the pups into pools or elsewhere may be indicative of a problem and should be monitored, again without disturbance to the parents. In general, excessively forceful, rapid, or uncoordinated interactions with pups are abnormal.
- Inappropriate mothering behavior by the dam. This may include: neglecting the pups, not lying still or lying incorrectly preventing pups from nursing, not staying long enough to allow for sufficient nursing by the pups, not visiting the pups frequently enough to allow for sufficient nursing, and pulling pups off their teats. These behaviors may indicate an inexperienced or stressed mother, or problem with lactation, such as insufficient milk production. This failure to produce sufficient milk amounts has been known to occur for varying periods of time as a reaction to stress.
- Biting, hitting, or laying on the pups; attempted drowning of, or eating pups

Further information on these abnormal occurrences can be obtained from Wünnemann (1995a,b), Flügger (1997, Autuori & Deutsch (1977), Corredor & Muñoz (2004), Sykes-Gatz (2005) and Sykes-Gatz and Gatz (2007).

7.2 Artificial Insemination

The practical use of artificial insemination (AI) with animals was developed during the early 1900s to replicate desirable livestock characteristics to more progeny. Over the last decade or so, AZA-accredited zoos and aquariums have begun using AI processes more often with many of the animals residing in their care. AZA Studbooks are designed to help manage otter populations by providing detailed genetic and demographic analyses to promote genetic diversity with breeding pair decisions within and between our institutions. While these decisions are based upon sound biological reasoning, the efforts needed to ensure that transports and introductions are done properly to facilitate breeding between the animals are often quite complex, exhaustive, and expensive. Also, conception is not guaranteed.

At this time AI is not used in any otter species but semen collection techniques and preservation are being researched by Bateman et al. (2005, 2009).

7.3 Pregnancy and Parturition

It is extremely important to understand the physiological and behavioral changes that occur throughout an otter's pregnancy. This information is contained in Section 7.1.

7.4 Birthing Facilities

As parturition approaches, animal care staff should ensure that the mother is comfortable in the area where the birth will take place, and that this area is "baby-proofed." This information is contained in Section 7.1.

7.5 Assisted Rearing

Although mothers may successfully give birth, there are times when they are not able to properly care for their offspring, both in the wild and in *ex-situ* populations. Fortunately, animal care staffs in AZA-accredited institutions are able to assist with the rearing of these offspring if necessary.

Hand-rearing may be necessary for a variety of reasons: rejection by the parents, ill health of the mother, or weakness of the offspring. Careful consideration should be given as hand-rearing requires a great deal of time and commitment (Muir 2003). Before the decision to hand-rear is made, the potential for undesirable behavioral problems in a hand-reared adult should be carefully weighed (e.g., excessive aggression towards humans (rare in most otters), inappropriate species-specific behavior, etc.) and plans made to minimize deleterious effects on the development of natural behaviors as far as possible. This may require extensive time commitment on the part of staff, plans for fostering, relocation of the young to another facility, exposure to species-specific sounds, etc. At this time, the AZA Otter SSP is recommending hand-rearing of all otter species, if necessary.

Pups that have been abandoned by their mother should be removed as soon as possible to prevent infanticide. See Chapter 6, section 6.5 for a 'Neonatal Examination and Monitoring Protocol'. Offspring that are not receiving milk will be restless, possibly calling continuously, may be hypothermic, and scattered around the enclosure. Another indicator of trouble would be the female moving around the exhibit continuously while carrying the young; this could mean she is not comfortable with the denning provided, or there is a problem with her or the pups (Muir 2003). If it is necessary to remove offspring because of an exceptionally large litter, it is best to remove two of the largest pups. The temptation is often to take the smallest, but they stand the best chance if raised by their mother. Hand-rearing of singletons is more likely to lead to severe imprinting on humans than if they have a conspecific to play with (Muir 2003). The AZA Otter SSP recommends that singleton pups being hand-reared be placed together, if at all possible. To date, fostering has been attempted once with otter pups and was successful. A pup was taken from a female with no milk and sent to another facility where their female was already nursing pups. In these cases, the AZA Otter SSP management team should be consulted first. Other institutions have been successful at supplement feeding pups left with their mother. Young otters removed for hand-rearing should not routinely be reintroduced to the parents with an expectation of acceptance. Introductions of hand-reared animals should follow procedures specified in the Introduction/Reintroduction section.

Physical Care Protocol: Incubators provide the best source of warmth. Heat lamps are too intense and can be dehydrating. In an emergency, hot water bottles wrapped in a towel may be placed in a box with the pups nestled next to it, or they can be warmed slowly by placing them next to your body (Muir 2003). Pups may feel more secure if wrapped in layers of towels; this also aids in keeping them warm (Muir 2003). Pups should be dried after feeding/bathing to prevent hypothermia until they are proficient at self-grooming. The normal body temperature for pups is unknown, but the animal should feel warm to the touch.

Altricial young are unable to self-regulate their body temperature during the early postnatal period and require an external source of warmth. If an incubator is not used, it may be necessary to place a heating pad, set on low, under the housing container until the pups are able to thermo regulate. Meier (1986) and Wallach & Boever (1983) recommend 29.4-32.2°C (85-90°F) and 50-60% humidity as the desired incubator setting for neonate mustelids. The temperature should be gradually reduced to room temperature, 21.2-23.9°C (70-75°F), over the course of about three weeks (unless the neonate becomes ill). Litters of pups are less likely to need additional ambient heat since huddling together may provide an adequate amount of warmth. External temperatures should be closely monitored to prevent hyperthermia. Rapid and/or open-mouth breathing, restlessness, and hair loss are indication of an external environment that is too warm.

Pups should be stimulated to urinate and defecate at least 4-5 times each day for several weeks, generally before feeding. However, some animals may respond better to post-feeding stimulation. The genitals and anal area are rubbed gently with a finger, towel, or damp cotton to stimulate the baby to urinate and have a bowel movement. If pups do not urinate and/or defecate after two successive feedings, the formula should be reviewed and their health status evaluated immediately.

Specific environmental parameters, formula information, etc. for hand-rearing *L. canadensis* and *P. brasiliensis* pups can be found in the North American River Otter Husbandry Notebook, 2nd Edition (Reed-Smith 2001) and International Giant Otter Studbook Husbandry and Management Information and Guidelines (Sykes-Gatz 2005), respectively; these are available on the Otter Specialist Group web site (www.otterspecialistgroup.org). The hand-rearing of giant otters (*P. brasiliensis*) is somewhat different than that of other otter species, because their development is slower. Detailed information on the types of records needed, signs of illness, etc. are available in the Giant Otter Husbandry Manual (Sykes-Gatz 2005).

Feeding Amount and Frequency: Initially, the animal should receive only an electrolyte solution for the first 2-3 feedings, depending on how compromised it is. This is to rehydrate the animal and clear the stomach of the maternal milk. The artificial formula should be started at a diluted concentration, generally at a 1:4 ratio (mixed formula: water) for another 2-3 feedings. It generally takes about 72 hours to get the animal on full-strength formula by gradually offering higher concentrations. Depending on the species, 4-5 feedings of each concentration level (1:2, 1:1, 2:1, full-strength) are required to allow for adaptation and to minimize the onset of digestive problems, particularly diarrhea. During the initial phase (24-36 hours), weight loss is to be expected, but the animal should quickly begin to maintain weight and then start gaining as the formula concentration increases. It is important that the pups are not given full strength formula too soon (in less than 48 hours after pulling for hand-rearing) because the likelihood of diarrhea is extremely high. Diarrhea is of particular concern with neonates less than one week of age, because they have very little or no immunity to infections.

Pups should have a normal body temperature and be properly hydrated before starting them on formula. Young mammals require a specific amount of calories per day for optimum development and growth. A nutritionally dense milk formula will allow for fewer feedings than more dilute formulas that are low in fat or protein. A method for calculating the volume of food to be offered per meal as well as total daily amount is presented below.

The Basal Metabolic Rate (BMR) or Basal Energy Requirement (BER) is the amount of energy (kcal) an animal needs for basic metabolic function at rest in a thermo-neutral zone. This represents the amount of calories it needs to stay alive, without having to use energy to maintain normal body temperatures (Grant 2004). Mustelids have a higher metabolic rate per body weight than many other placental mammals. For that reason, Iversen's equation of 84.6 x body weight (in kg) $^{0.78}$ (Iversen 1972) is used rather than Kleiber's equation of 70 x body weight (in kg) $^{0.75}$ (Kleiber 1947) typically used for other species. Therefore, for a 200g river otter, the BER would be: $84.6 \times 0.2^{0.78} = \sim 24$ kcal/day.

Once the BER is established, the Maintenance Energy Requirement (MER) can be calculated. This measurement determines the amount of calories the animal needs to function in a normal capacity at its life stage. For adults in a maintenance life stage, the BER is multiplied by 2. For pups that have a higher metabolism and are developing and growing, the BER is multiplied by 3 or 4 (Evans 1985), depending on the species and other factors.

The stomach capacity for most placental mammals is 5-7% of the total body weight (Meehan 1994). Convert the body weight into grams to find the stomach volume in ml (cc). To calculate the stomach capacity in ounces, convert body weight into grams (30g ~ 1 oz). It is important that units are the same for body weight and stomach volume. The stomach capacity is the amount of formula an infant can comfortably consume at one feeding. Offering much more than this value may lead to overfilling, stomach distension, and bloat. It also prevents complete emptying of the stomach before the next feeding, and promotes the overgrowth of potentially pathogenic bacteria, diarrhea, and enteritis (Evans 1985).

The following calculations will determine the total volume and kcal to feed/day, as well as the amount of formula for each feeding and the total number of feedings daily.

- Calculate Maintenance Energy Requirement: 84.6 x body wt (kg) $^{0.78}$ x 3.
- Determine stomach capacity (amount that can be fed at each meal): Body weight (in grams) x 0.05.
- Divide Maintenance Energy Requirement (number of calories required per day) by the number of kcal/ml in the formula to determine the volume to be consumed per day (this can be converted into ounces by dividing it by 30).
- Divide ml of formula per day by volume to be consumed at each meal (stomach capacity). This gives the number of meals to offer per day.
- Divide 24 hours by the number of feedings/day to find the time interval between feedings.
- See Table 12.

Table 12: Calculations for formula volume and feeding frequency for neonate with an approximate birth weight of 135g (MER = Maintenance Energy Requirement)

Step 1: calculate MER	84.6 x 0.135kg $^{0.78}$ x 3	~53 kcal/day
Step 2: determine stomach capacity	135g x 0.05 (stomach capacity of 5% body weight)	~7g (ml) per feeding
Step 3: calculate daily volume fed	53 kcal/day (MER) / 1.78 kcal/ml (formula contents)	~30ml/day
Step 4: number of feedings	30ml/day (total volume fed) / 7ml/feeding (stomach capacity)	4.2 feedings/day (=5)
Step 5: feeding schedule	24 hrs/5 feedings	Every 5 hours

New calculations should be performed every few days so formula volume can be adjusted to accommodate growth. The general target average daily gain for infants is 5-8% increase of body wt/day while on formula feeding and 8-10% body wt increase/day on weaning diet (Grant 2005). Since neonates being hand-reared (less than one week of age) are typically severely compromised, they should be given smaller, more frequent feedings than calculated until roughly 2-4 weeks of age.

As a general rule, animals should have an overnight break between feedings that are no longer than twice the time period between daytime feedings (equivalent to missing one feeding). For example, if they are being fed every three hours during the day, they can go six hours at night without food. When they are eating every four hours, they can go eight hours at night. It is not advisable to go more than eight hours between feedings with species that typically nurse throughout the day when mother-raised. Intervals between feeding also will depend on how healthy or strong the infants are. Very weak neonates will probably need feedings every few hours even through the night; typically this is necessary for only a few days to a week. The AZA Otter SSP recommends that neonates be fed every two hours around the clock initially. Depending on how the animal is doing, these feedings may be stretched to every three hours after the first few weeks.

Otter pups should only be fed if the pup is hungry and suckling vigorously. Weak infants may be hypothermic, dehydrated and/or hypoglycemic. Do not offer anything by mouth until the body temperature is within the normal range for its age (i.e., warm, not hot, to the touch). Electrolytes can be offered orally if the pup is suckling, or subcutaneously if it is too weak; 2.5-5% dextrose can also be given to raise the pup's glucose level. More research is required to determine body temperature norms for young of all the otter species. This information should be collected by all facilities hand-rearing otter pups and submitted to the AZA Otter SSP. Young animals will be hungry at some feedings, less at others, but this is quite normal (Muir 2003). However, refusal of two feedings is a sign of trouble in young otters. Pups will not die from being slightly underfed, but overfeeding may result in gastrointestinal disease, which is potentially fatal.

If any animal's formula is changed abruptly, it is likely to cause diarrhea, which can dehydrate the pup quickly. Any formula changes should be made slowly, by combining the formulas and gradually changing the ratio from more of the first to more of the second. If an animal develops diarrhea or becomes constipated with no change having been made in the formula, consult the veterinarian. In general, adjusting the formula ratios should be attempted before medicating the animal. For diarrhea, increase the ratio of water to all the other ingredients. Be sure the water has been boiled or sterilized well, and the bottle is clean. Subcutaneous fluids (e.g., lactated ringers) may be needed if the infant dehydrates significantly.

Feeding Techniques: To bottle feed, hold the pup in the correct nursing position; sternally recumbent (abdomen down, not on its back), with the head up. Place the hand holding the bottle in such a way that it provides a surface for the pup to push against with its front feet. If milk comes through their nose, the nipple hole may be too large or the pup may be trying to eat too quickly. Make sure there is consistency with who is feeding the pups. Note any changes in feeding immediately. Decreased appetite, chewing on the nipple instead of sucking, or gulping food down too quickly can be signs of a problem (Blum 2004).

It is important to keep in mind that neonates are obligate nose breathers and incapable of breathing through their mouths and nursing at the same time. For this reason, respiratory infections can be life threatening because they may interfere with breathing and make nursing difficult or impossible (Meier 1985). Aspirated formula is frequently a contributing factor to neonatal respiratory infections; to avoid this, be sure to select the appropriate nipple. The nipple's hole needs to suit the neonate's sucking reflex. Also, if a nipple is too stiff, the pup may tire and refuse to nurse.

If an animal aspirates fluids the recommended protocol is to hold the infant with head and chest lower than the hind end. A rubber bulb syringe should be used to suck out as much fluid from the nostrils and the back of the throat as possible. If aspiration is suspected, or if fluid is heard in the lungs, contact the veterinarian immediately; do not administer drugs without the veterinarian's involvement. Monitor body temperature closely for the occurrence of a fever and a decline in the animal's appetite and general attitude. Depending on the condition and age of the animal, diagnostic procedures may include radiographs, CBC, and chemistry. It is possible to start a course of antibiotics while results from the bloodwork are pending, and the attending veterinarian can prescribe an appropriate antibiotic course.

Pups will need to be stimulated to urinate and defecate for the first six weeks of life, either immediately before or after feeding. Parent-reared giant otters of *ex-situ* populations are reported to have required stimulation by their parents to urinate or defecate for up to 10 weeks of age. In at least one case, a hand-reared individual needed to be stimulated to urinate/defecate until it was 2.5-3 months old (Sykes-Gatz 1999-2006). In other cases (Corredor & Muñoz 2004), pups were reported using latrines on their own at 9 weeks of age. Some pups also may require "burping" to prevent gas build-up in the abdomen.

P. brasiliensis: One of the most reliable methods of determining if the young are nursing successfully is monitoring for what Sykes-Gatz (2005) calls the "nursing hum", which pups make when they are suckling. This hum is a somewhat higher pitched and faster vocalization than the contact hum described by Duplaix (1980), which has a twittering quality to it. The nursing hum is performed when a pup is nursing from the mother or a bottle. Sykes-Gatz (2005) also reports this call, when given by a caregiver, can encourage pups to feed. From birth, the pups also display "tail wagging" when they nurse, wagging their tails rather quickly and repeatedly from side-to-side. Some individuals may require "burping" to prevent gas build-up in the abdomen. For more detailed information refer to Sykes-Gatz (2005).

A. cinereus: Pups may be slow to learn how to suckle from a bottle, in one case taking eight days before suckling without aspirating (Webb 2008). Care should be taken to ensure that the nipple's hole is not too large and that pups are fed slowly at the beginning. For additional information see Webb 2008 (also available online at (www.otterspecialistgroup.org/Library/TaskForces/OCT.html).

Hand-rearing Formulas: It is important that the artificial milk formula matches the maternal milk in protein, fat, and carbohydrate composition as closely as possible. Table 13 provides information on the nutritional content of otter milk, and Table 14 provides information on the nutritional composition of selected substitute milk formulas/replacers.

Table 13: Otter (*Lutra spp.)* Milk Nutrition Composition on As Fed (AFB) and Dry Matter Basis (DMB) (Ben Shaul 1962; Jenness & Sloan 1970)

Species	Solids %	Kcal (ml)	Fat %	Protein %	Carb. %
Otter	38.0	2.6 (AFB)	24.0 (AFB) 63.2(DMB)	11.0 (AFB) 28.9(DMB)	0.1 (AFB) 0.3 (DMB)

Esbilac® (or Milk-Matrix® 33/40) is preferred as the base for milk formulas offered to otters and provides good pup growth. The addition of Multi-Milk® (or Milk-Matrix® 30/55) increases the total fat and protein content without adding substantially to the carbohydrate content of the formula. The maternal milk composition of otter milk only has a trace amount of milk sugars, so this component of the substitute formula should be kept as low as possible to prevent gastric upset and diarrhea. See Table 14 on the following next page.

Table 14: Nutritional analysis of commercial animal milk replacers

Product	Solids %	Fat %	Protein %	Carbohydrates %	Ash %	Energy (KCAL/ML)
Esbilac						
Undiluted powder	95.00	40.00	33.00	15.80	6.00	6.20
Diluted 1:3*	15.00	6.00	4.95	2.38	0.90	0.93
Diluted 1:1.5*	30.00	12.00	9.90	4.76	1.80	1.86
Liquid product	15.00	6.00	4.95	2.38	0.90	0.93
KMR						
Undiluted powder	95.00	25.00	42.00	26.00	7.00	5.77
Diluted 1:3*	18.00	4.50	7.56	4.68	1.26	1.04
Diluted 1:1.5*	36.00	9.00	15.12	9.36	2.52	2.07
Liquid product	18.00	4.50	7.56	4.68	1.26	1.04
Multi-Milk						
Undiluted powder	97.50	53.00	34.50	0	6.63	6.85
Diluted 1:1*	22.70	12.00	7.83	0	1.51	1.55
Diluted1.5:1*	36.00	19.59	12.75	0	2.54	2.47
Evaporated Milk						
Undiluted product	22.00	7.00	7.90	9.70	0.70	1.49
Multi-Milk:KMR+						
1:1*	22.81	8.93	8.71	3.20	1.55	1.45
3:1*	22.90	10.97	8.63	1.54	1.59	1.57
4:1*	22.90	10.90	8.27	1.17	1.50	1.51
1:3*	22.70	7.28	9.10	4.39	2.30	1.37
1:4*	22.60	6.95	9.16	4.68	1.57	1.36
Multi-Milk:KMR++						
1:1*	34.22	13.40	13.07	4.80	2.33	2.18
3:1*	34.55	16.46	13.03	2.31	2.39	2.36
4:1*	34.55	16.35	12.41	1.76	2.25	2.28
1:3*	34.05	10.92	13.65	6.59	3.45	2.06
1:4*	33.90	10.43	13.74	7.02	2.36	2.04
Multi-Milk:Esbilac+						
1:1*	22.81	10.63	7.70	1.78	1.44	1.49
3:1*	22.93	11.63	8.00	0.89	1.52	1.56
4:1*	22.90	11.60	7.86	0.71	1.49	1.55
1:3*	22.70	9.81	8.75	2.67	2.13	1.51
1:4*	22.60	9.65	7.54	2.84	1.39	1.43
Multi-Milk:Esbilac++						
1:1*	34.22	15.95	11.55	2.67	2.16	2.24
3:1*	34.40	17.45	12.00	1.34	2.28	2.33
4:1*	34.35	17.40	11.79	1.07	2.24	2.33
1:3*	34.05	14.72	13.13	4.01	3.20	2.28
1:4*	33.90	14.48	11.31	4.26	2.09	2.15

* Ratio of powder to water; + Ratio of powder-to-powder, diluted 1 part powder to 1 part water; ++ Ratio of powder-to-powder, diluted 1.5 parts powder to 1 part water (Evans 1985)

The addition of an anti-gas build-up product to the formula should be considered (milk sugars can cause the build-up of gas). Lact-aid® is an enzyme that has been used successfully with many species. Add two drops of Lact-aid® to 100ml of mixed formula. The formula then should be refrigerated for 24 hours prior to feeding for the enzyme to perform correctly (Grant 2005). *Lactobacillus* spp., in Bene-bac® or Probios®, is a group of beneficial gut bacteria that also break down milk sugars in the digestive tract. Follow label instructions for these products.

Table 15: Substitute milk formulas for otters. Values taken from product composition documents available from PetAg™ (K.Grant, personal communication)

Formula	% Solids	% Fat	% Protein	% Carb	Kcal/ml
<u>Formula #1</u> 1 part Esbilac® or Milk Matrix® 33/40 1 part Multi-Milk® or Milk Matrix® 30/55 2 parts water	30.9	15.6	10.5	2.7	1.78
<u>Formula #2</u> 1 part Multi-Milk® or Milk Matrix 30/55® 1 part water	31.3	17.8	10.4	1.1	1.91

<u>L. Canadensis</u>: At this time, the preferred formula is canned Esbilac® due to palatability and good pup growth. Milk Matrix® based formulas also are nutritionally suitable but some facilities have had pups refuse this formula (Blum 2004) while others have had good success.

Weaning: Some of the following recommendations do not apply to *P. brasiliensis* (e.g. offering food in a bowl). The weaning process should be started when the pup shows interest in solid food, generally at about eight weeks of age. If the pup is not gaining enough weight on formula alone, solid food can be added at six weeks of age (this may need to be pureed or chopped). To begin, formula can be mixed with AD diet (canned cat food or similar), baby food, mashed up fish, rice cereal, or ground meat. New food can be added to the bottle; feed this mixture with a syringe, baby bottle, or offer it in a bowl. Do not provide milk formula in a bowl to giant otters, as they tend to inhale liquids into the nose until they become proficient at eating solid foods (McTurk & Spelman 2005). Only add one new food component to their diet every couple of days until they are eating solids well. It is best to be creative, flexible, and not to rush the weaning process. In the case of problems, try different approaches, try them multiple times, and try foods in new ways like bottles, syringes, suction bulbs, bowls, etc. Do not cut back on bottle-feeding to make the pup "hungry". Offer new food at the beginning of the feeding and finish with the bottle (Blum 2004). Situations to watch for during the weaning process include (Blum 2004): weight loss, diarrhea and sucking behavior. If sucking on tails, feet, genitals, etc. is observed between feedings, an additional bottle-feeding should be offered for a few days. R. Green of the Vincent Wildlife Trust recommends putting orange oil on the genitals to discourage sucking; this worked well with *Lutra lutra* and is not harmful to the otter (G.Yoxon, personal communication).

Swimming, Terrestrial Activities, and Behavioral Stimulation: Otter pups are not born knowing how to swim and may even be scared of the water. They will usually start to take interest in the water at 4-8 weeks of age. The pups should be started off in shallow pools and watched carefully; once comfortable, they can gradually be introduced to deeper water. Pups should be dried off completely and warmed after their swim.

Enrichment is crucial to the development of the pups; toddler safe toys, grooming materials, dens, climbing structures, live food, etc. have all been used successfully. The more items they are introduced to otters at an early age, the more they will interact with as they age. All toys need to be safe and approved by the veterinary staff. The suitability of toys should be regularly re-evaluated, as some may no longer be safe as the otter grows. Due to the tendency of all otters to take things into the water, the use of cardboard or other paper-type items, especially for young animals, is not recommended. Cases of these items becoming water logged and congealing in an animal's mouth or over their nose have been reported.

Pup Development: The following information provides a summary of pup development. More specific information can be found in the Otter Husbandry Manuals (Lombardi et al. 1998; Reed-Smith 2001; Sykes-Gatz 2005). See Appendix I for pup weight charts.

<u>A. cinereus</u>:

- Eyes begin opening at between 17 and 28 days, fully open by day 45
- Teeth begin erupting about day 20 and canines erupting ~ day 91 (Webb 2008)
- Thermo regulating well on their own about day 38 (Webb 2008)
- Moving on their own between day 39 and day 50

- Urinating and defecating on their own (hand reared animals) by day 59 (Webb 2008)
- Generally born with mostly grayish fur, darkens by 6-7weeks
- Solid food 7-8 weeks; weaned 82-120 days
- Hand reared animals eating solid well by day 92 and weaned on day 130 at a weight of 2336 grams (Webb 2008)

A. capensis: At this time there is no information available on pup development. More research is required.

L. maculicollis: Pups are born with white on their lips. After a few days, patches of white/orangish colored hair develop on their chest or groin area. These patches change to an orange color, before changing back to cream or white as the pups reach full growth or maturity. The age at which these color changes occur appears to be highly variable and is currently being documented (D.Benza, personal communication; R.Willison, personal communication).

- First spots seen ~6 days, whitish but turned orange in a few days. More orange spots developed by day 42
- Eyes open at 34-46 days
- First crawling at about 20 days, crawling well 42 days
- First teeth erupting at 23-29 days, all teeth in ~78 days
- Walking well at ~ 37 days, running 59 days
- Leaving den on own at about 57 days
- Playing in water bowl ~ 61 days
- First going in to water on their own at about 57-91 days; variation comes from water tub versus pool exploration
- First pool swimming lessons ~ 86 days (timing may be due to when family is allowed into the exhibit)
- First eat solids at about 60-73 days

L. canadensis: Consult the North American River Otter Husbandry Notebook 1st & 2nd editions for more detailed information (Reed-Smith 1994, 2001).

- Birth weight: 120-135g
- Born blind with dark brown fur
- External ears are flat against the head, and claws and toe webbing are well formed.
- Deciduous upper and lower canines erupt at about 12 days
- Eyes fully open at 28-35 days
- Walking at about 35-42 days, first swimming lesson generally at 28-56 days
- Beginning to play ~25-42 days
- Leaving nest box on their own ~49 days
- Pelt change 28-56 days, born with all dark fur
- First solid food taken at 42-56 days
- Localized latrine use ~49 days
- Pups should be weaned by 3-4 months of age

P. brasiliensis: Because this species requires complete isolation and privacy (particularly primiparous pairs), detailed information on pup development is taken from video and audio recordings. Sykes-Gatz (2005) provides more detail on pup rearing and development. McTurk & Spelman (2005) also provide information on hand-rearing and rehabilitation of orphaned giant otters. An outline of giant otter pup development is provided below (Wünnemann 1990, 1995a,b; McTurk & Spelman 2005; Sykes-Gatz 2005, 1999-2006; V.Gatz, personal communication; N.Duplaix, personal communication):

- Weight at birth – 150-265g
- Birth pelt is grayish in color and darkens by 6-7 weeks of age
- Eyes begin opening at ~28 days and are fully open by ~45 days
- Pups should be moving on their own by 39-50 days
- First leave the nest box on their own at 63-67 days
- First swimming lessons at 20-60 days, or as early as 11 days
- Pups can be reliably sexed at 10 weeks
- Pups swim on their own for the first time at 63-67 days

- Pups will begin playing with solid food at roughly 56 days, but generally do not consume any until about 70-90 days.
- Pups will begin weaning at roughly 4 months of age, but can nurse insignificant amounts (this provides little nutritional value) at 6.5 to 8 months of age.
- Fish should first be offered pups at 2.5-4 months of age
- 100% of their required caloric intake should be offered in formula/mother's milk form until roughly 2.5 months of age
- Pups should be weaned from formula between 6.5-10 months of age
- Pups should be weaned on a fish based diet; rice cereal has been used successfully as a dietary addition for hand-reared pups. Formula should not be offered in a bowl, as giant otters tend to inhale liquids into the nose until they are proficient at eating solid foods.
- Pups are approximately ¾ the size of adults at 10 months of age, although this will vary

7.6 Contraception

Many animals cared for in AZA-accredited institutions breed so successfully that contraception techniques are implemented to ensure that the population remains at a healthy size. In addition to reversible contraception, reproduction can be prevented by separating the sexes or by permanent sterilization. In general, reversible contraception is preferable because it allows natural social groups to be maintained while managing the genetic health of the population. Permanent sterilization may be considered for individuals that are genetically well-represented or for whom reproduction would pose health risks. The contraceptive methods most suitable for otters are outlined below. More details on products, application, and ordering information can be found on the Intstitution E webpage: www.stlzoo.org/contraception.

The progestin-based melengestrol acetate (MGA) implant, previously the most widely used contraceptive in zoos, has been associated with uterine and mammary pathology in felids and suspected in other carnivore species (Munson 2006). Other progestins (e.g., Depo-Provera®, Ovaban®) are likely to have the same deleterious effects. C.Osmann (personal communication) specifically recommends against using progestins in *P. brasiliensis* for the reasons mentioned above and because these side effects may compromise future breedings. For carnivores, one institution now recommends GnRH agonists, e.g., Suprelorin® (deslorelin) implants or Lupron Depot® (leuprolide acetate), as safer alternatives. Although GnRH agonists appear safe and effective, dosages and duration of efficacy have not been systematically evaluated for all species. GnRH agonists can be used in either females or males, and side effects are generally those associated with gonadectomy, especially weight gain, which should be managed through diet. Suprelorin® was developed for domestic dogs and has been used successfully in African clawless otters, North American river otters, Asian small clawed otters and sea otters.

Gonadotropin Releasing Hormone (GnRH) Agonists: GnRH agonists (e.g., Suprelorin® implants or Lupron Depot®) achieve contraception by reversibly suppressing the reproductive endocrine system and preventing production of pituitary (FSH and LH) and gonadal hormones (estradiol and progesterone in females and testosterone in males). The observed effects are similar to those following either ovariectomy in females or castration in males, but are reversible. GnRH agonists first stimulate the reproductive system, which can result in estrus and ovulation in females or temporary enhancement of testosterone and semen production in males. Then, down-regulation follows the initial stimulation. The stimulatory phase can be prevented in females by daily Ovaban administration for one week before and one week after implant placement (Wright et al. 2001).

GnRH agonists should not be used during pregnancy, since they may cause spontaneous abortion or prevent mammary development necessary for lactation. They may prevent initiation of lactation by inhibiting progesterone secretion, but effects on established lactation are less likely. New data from domestic cats have shown no effect on subsequent reproduction when treatment began before puberty; no research in prepubertal otters has been conducted.

A drawback of these products is that time of reversal cannot be controlled. Neither the implant (Suprelorin®) nor the depot vehicle (Lupron®) can be removed to shorten the duration of efficacy to time reversals. The most widely used formulations are designed to be effective for either 6 or 12 months, but those are for the most part minimum durations, which can be longer in some individuals.

Although GnRH agonists can also be an effective contraceptive in males, they are more commonly used in females. This is because monitoring efficacy by suppression of estrous behavior or cyclic gonadal steroids in feces is usually easier than ensuring continued absence of sperm in males, since most institutions cannot perform regular semen collections. Suprelorin® has been tested primarily in domestic dogs, whereas Lupron Depot® has been used primarily in humans, but should be as effective as Suprelorin® since the GnRH molecule is identical in all mammalian species.

If used in males, disappearance of sperm from the ejaculate following down-regulation of testosterone may take an additional 6 weeks, as with vasectomy. It should be easier to suppress the onset of spermatogenesis in seasonally breeding species, but that process begins at least 2 months before the first typical appearance of sperm. Thus, treatment should be initiated at least 2 months before the anticipated onset of breeding.

Progestins: If progestins (e.g., Melengestrol acetate (MGA) implants, Depo-Provera® injections, Ovaban® pills) have to be used, they should be administered for no more than 2 years and then discontinued to allow for a pregnancy. Discontinuing progestin contraception and allowing non-pregnant cycles does not substitute for a pregnancy. Use of progestins for more than a total of 4 years is not recommended. MGA implants last at least 2 years, and clearance of the hormone from the system occurs rapidly after implant removal. Progestins are considered safe to use during lactation.

Vaccines: The porcine zona pellucida (PZP) vaccine has not been tested in otters, but may cause permanent sterility in many carnivore species after only one or two treatments. This approach is not recommended.

Ovariectomy or Ovariohysterectomy: Removal of ovaries is a safe and effective method to prevent reproduction for animals that are eligible for permanent sterilization. In general, ovariectomy is sufficient in young females, whereas removal of the uterus as well as ovaries is preferable in older females, due to the increased likelihood of uterine pathology with age.

Vasectomy: Vasectomy of males will not prevent potential adverse effects to females that can result from prolonged, cyclic exposure to the endogenous progesterone associated with the pseudo-pregnancy that follows ovulation. This approach is not recommended for otters.

Chapter 8. Behavior Management

8.1 Animal Training

Classical and operant conditioning techniques have been used to train animals for over a century. Classical conditioning is a form of associative learning demonstrated by Ivan Pavlov. Classical conditioning involves the presentation of a neutral stimulus that will be conditioned (CS) along with an unconditioned stimulus that evokes an innate, often reflexive, response (US). If the CS and the US are repeatedly paired, eventually the two stimuli become associated and the otter will begin to produce a conditioned behavioral response to the CS.

Operant conditioning uses the consequences of a behavior to modify the occurrence and form of that behavior. Reinforcement and punishment are the core tools of operant conditioning. Positive reinforcement occurs when a behavior is followed by a favorable stimulus to increase the frequency of that behavior. Negative reinforcement occurs when a behavior is followed by the removal of an aversive stimulus to also increase the frequency of that behavior. Positive punishment occurs when a behavior is followed by an aversive stimulus to decrease the frequency of that behavior. Negative punishment occurs when a behavior is followed by the removal of a favorable stimulus also to decrease the frequency of that behavior. AZA-accredited institutions are expected to utilize reinforcing conditioning techniques to facilitate husbandry procedures and behavioral research investigations.

Otters are excellent candidates for behavioral training programs focusing on routine and non-routine husbandry tasks, such as shifting, weighing, entering squeeze cages or crates, stationing for close visual inspections or injections, etc. Standard positive reinforcement behavioral training techniques are used successfully on river otters at numerous facilities. As far as possible, all animals should routinely shift into a holding area and readily separate into specific holding areas on cue. Animals should be trained to come to the keeper when called for daily health checks, and remain calm and not aggressive during these checks.

Keepers should avoid use of aversive stimuli in the daily management of otters. Profound aversive stimuli such as squirting with hoses, loud noises, harsh words, and long-term withholding of food are inappropriate unless serious injury of keeper or animal is imminent (e.g., serious fight). In general, otters respond to profound aversive stimuli with fear and/or aggression. It is best to maintain positive and pleasant keeper/animal interactions. Assessing the animal's motivation (e.g., why should it "want" to come in? Why does it "want" to stay outside? What is the animal's motivation, and how does it relate to the animal's behavior in the wild?) is a useful exercise when training problems occur. Patience and planning are keys to success (Wooster 1998). See Table 16 for a list of commonly trained otter behaviors, as well as relevant cues and criteria. Successful training programs include those that involve establishing training goals set by the entire staff. These goals include a list of behaviors that facilitate desired husbandry procedures. Goals are accomplished by developing training plans that define training steps, cues, and criteria for the desired behaviors. Progress of training plans should be monitored and evaluated. Once desired behaviors are achieved, they should be maintained with practice on a regular daily basis. See Appendix J for additional training information.

Otters can be trained through positive reinforcement for almost all behaviors required for husbandry procedures, whether it is routine or a not-so common event. Non-routine husbandry behaviors can include procedures such as hand injection, ultrasound, nipple manipulation/milk collection (for larger species in particular), and tactile body exams. A *P. brasiliensis* female was successfully trained to allow manual milk pumping and ultrasounds to detect pregnancy and uterine condition (Gatz 2002). Giant otters also have been trained to allow tactile body exams, the taking of body temperature, weight, heart and respiration rates, as well as to participate in other husbandry procedures (Sykes-Gatz 2005).

Otters respond quickly to voice commands via operant conditioning. Training can be done on or off exhibit. Otters respond to a protected contact and free contact situation. In general, the otter species should be trained in a protected contact situation (i.e., keeper and animal should be separated by a mesh barrier). Exhibits should be designed with mesh at a particular area specifically for training. There are some species (*A. cinereus*) or cases (*L. canadensis* particularly males) where an institution feels that protected contact training may not be called for, but these decisions should be carefully evaluated on an ongoing basis. If institutional philosophy permits, otters can be a part of an educational talk or keeper talk in a free contact area within their exhibit.

It is recommended that all facilities have holding areas in order to shift animals into/out of their primary enclosure. Husbandry training may occur anywhere the individual animal seems to feel comfortable, and where the keeper can safely access them. Managers and caretakers should decide if food rewards can be hand fed or if a meat stick should be used to deliver the food.

The following table (Table 16) provides some examples of husbandry training cues and criteria for behaviors trained with otters at various AZA institutions. Mckay (2009) describes some basic approaches to training otters.

Table 16: Sample behaviors & training cues for otters (provided by: *Institution F; **Institution G; ***Institution H; ^Institution I; ^^Institution J; & +Institution K). Behaviors not identified are trained at all reporting institutions.

Behavior	Verbal cue	Visual cue	Criteria for reinforcement
Down *	"down"	Hand flat in front of abdomen- moved in a downward motion	Animal lays down quietly
Up *	"up"	Index finger moved in upward motion to place you want them to target to	Animal moves to position of index finger
Up ^	"up"	Left index points into the air	Animal stands up
Stand **	"up"	Use left hand and give the thumbs-up sign	Otter keeps both back feet on the ground while standing up against the cage. Front feet should be hanging onto target pole place against the bars.
Kennel *	"in"	Index finger used to point into the kennel	Animal goes in kennel and allows door to be closed
Entering a crate **	"box"	Hand begins in fist in front of chest. As command is said, swing arm out and up in direction of the box and open hand into a high five.	Animal will enter crate and lie down at the far right end. Animal will wait in position until bridged.
Squeeze/Crate ***/^^	"crate"	Target into squeeze cage or point to crate	Animal enters and allows the door to be closed
Crate +		Hand placed on chain link near back of crate	Animal enters and stands in the crate, tail completely in
Scale *	"scale"	Index finger used to point to scale	Animal gets on scale & waits
Target *	"here"	Closed fist presented to front of mesh	Nose placed at position of fist
Target **	"target"	Hold up target pole	Animal grabs with both hands without biting – ASC otters
Target ***/ ^	"target"	Show target pole	Nose placed on target and holds until bridged
Target +		Show 15' broom handle on fence	Put nose to target
Stay **	"stay"	Right hand palm down and out. While in this position, push slightly toward animal while saying verbal cue.	Animal stands/sits still while trainer moves away and returns
Stay/remote stay ^	"stay"	Hold hand up, palm towards the animal Hold fist up	Animal stays calmly
Hold ^^	"hold"	Hand cue	Animal stays in place
Lying parallel to	'lie"	Palm flat out and facing down.	Animal lays down parallel to and

Behavior	Verbal cue	Visual cue	Criteria for reinforcement
cage front		Sweep arm in direction animal should face.	touching cage front. Remains calm and quiet until bridged.
Shift	“over”	Arm begins up and parallel to chest, index finger pointed up. (Use arm that is in the direction you want the animal to shift. Move arm and corresponding foot in a sweeping motion indicating the direction you want the animal to go).	The animal goes to the are indicated, comes to front of cage, stands quietly with eyes on trainer
Come in *	3 whistles- flat tone	None	Animal moves in to location of person whistling
Recall +	Clicker		Animal moves off exhibit to catch area
Station **/^^	None	Trainer stands in specified location with hands at their sides, beginning of training set	Animal comes to the front of the cage, stands quietly with their eyes on the trainer
Station ^	------	Point using two fingers of either hand to station desired	Animal moves to the spot and stays calmly
Follow	“come”	Say come and walk in direction you want the animal to go	The animal follows and stops directly in front of the trainer
Foot present	“toes”	Begin with right arm up parallel to body, index and middle fingers pointed up. Extend arm straight down (palm side down) continuing to point both fingers.	Animal should place both feet under the bottom of cage while lying down in front of trainer. It should be lying still and focused on the trainer.
Paw	“paw” “right” “left”	Visual signal for stand; point or target foot wanted.	Cue each foot to right or left, can use target or catch less dominant foot when opportunity rises; most have dominant foot they learn easily.
Ultrasound **	“up”	Cue as for up, trainer body can be low	Same as stand, animal should wait while being touched on abdomen with pole or wand.
Ultrasound ***	“touch”	Show wand	Otter stands on back legs and touches target with nose while abdomen/kidneys ultrasounded through cage mesh.
Paint ***	“paint”	Show painting apparatus	Animal grabs paint brush and puts paint on canvas.
Nipple presentation ***	“nipple”	Target up while standing on hind legs. Slowly reach with fingers extended, toward otter	Animal presents chest or abdomen against cage mesh for manipulation
Ventral present +		Target placed high on fence	Animal climbs fence until all feet off the ground and ventrum placed on fence
Jumping into the pool **	“water”	Use right hand with food in it. Start with hand in a fist in front of chest. With a sweeping motion, move fist up to cage. Arm should be parallel. Open hand palm up and out. Tap	The animal should jump into the water to retrieve food.

Behavior	Verbal cue	Visual cue	Criteria for reinforcement
		cage with palm to push the food into the pool	
Water ^	"water"	Right hand motions towards the water	Animal goes in the water
Circle ^	"circle"	Make a circle with right hand	Animal turns in a circle
Steady ^	"steady"	Verbal cue only	Used to keep the animal calm during tactile body examination

8.2 Environmental Enrichment

Environmental enrichment, also called behavioral enrichment, refers to the practice of providing a variety of stimuli to the animal's environment, or changing the environment itself to increase physical activity, stimulate cognition, and promote natural behaviors. Stimuli, including natural and artificial objects, scents, and sounds, are presented in a safe way for the otters to interact with. Some suggestions include providing food in a variety of ways (i.e., frozen in ice or in a manner that requires an animal to solve simple puzzles to obtain it), using the presence or scent/sounds of other animals of the same or different species, and incorporating an animal training (husbandry or behavioral research) regime in the daily schedule.

It is recommended that an enrichment program be based on current information in biology, and should include the following elements: goal-setting, planning and approval process, implementation, documentation/record-keeping, evaluation, and subsequent program refinement. Environmental enrichment programs should ensure that all environmental enrichment devices (EEDs) are safe and are presented on a variable schedule to prevent habituation AZA-accredited institutions must have a formal written enrichment program that promotes behavioral opportunities (1.6.1).

Enrichment programs should be integrated with veterinary care, nutrition, and otter training programs to maximize the effectiveness and quality of animal care provided. AZA-accredited institutions must have specific staff members assigned to oversee, implement, train, and coordinate interdepartmental enrichment programs (1.6.2).

AZA Accreditation Standard

(1.6.1) The institution must have a formal written enrichment program that promotes species-appropriate behavioral opportunities.

AZA Accreditation Standard

(1.6.2) The institution must have a specific staff member(s) or committee assigned for enrichment program oversight, implementation, training, and interdepartmental coordination of enrichment efforts.

Development of enrichment ideas should be goal-oriented, proactive, based upon the animal's natural history, individual history and exhibit constraints, and should be integrated into all aspects of their *ex-situ* population management. Providing the appropriate enclosure designs (e.g., land/water ratios, pool/land designs), substrates, and furnishings for each otter species are essential components of any enrichment program. Enrichment should encourage otters to behave as they would in the wild, as closely as possible. Successful enrichment techniques include variation of exhibit schedule or exhibit mates (where appropriate only), re-arranging of exhibit furniture/features, complete change of furniture (some of the old should always be retained to maintain the animal's scent and an element of the familiar), scents, sounds, toys (natural and artificial), herbs, spices, different substrates for digging/rolling, food items, and novel presentation of food items. It is important that enrichment items are not merely thrown in an exhibit and allowed to stay for extended periods – an enrichment program is only successful and useful if actively managed and constantly reviewed to ensure it encourages natural behaviors. The AAZK Enrichment committee provides the following general guidelines about enrichment:

"The goal of enrichment should be to maximize the benefit while minimizing unacceptable risks. All enrichment should be evaluated on three levels: 1) whether the enrichment item itself poses an unacceptable risk to the animals; 2) what benefit the animals will derive from the enrichment; and 3) whether the manner of enrichment delivery is apt to lead to problems.

A written plan of action that eliminates the most dangerous risk factors while maintaining the benefits of a challenging and complex environment can help animal managers develop a safe and successful enrichment program. Keepers should evaluate new and creative enrichment ideas with their managers and staff from other departments (curatorial, janitorial, maintenance, veterinary, nutritional, etc.) to decrease the frequency of abnormal and stereotypic behaviors or low activity levels, and to fine-tune enrichment ideas. For enrichment to be safely provided, it is strongly recommended that each institution establish enrichment procedures, protocols, and a chain of command that keepers can follow" (AAZK Enrichment Committee).

The AAZK Enrichment Committee also provides an excellent cautionary list for the various types of enrichment provided (accessed through www.aazk.org). This list includes key questions that should be answered for all enrichment items or programs to assess potential hazards. For example:

- Can the animals get caught in it or become trapped by it?
- Can it be used as a weapon?
- Can an animal be cut or otherwise injured by it?
- Can it fall on an animal?
- Can the animal ingest the object or piece of it? Is any part of it toxic, including paint or epoxy?
- Can it be choked on or cause asphyxiation or strangulation?
- Can it become lodged in the digestive system and cause gut impaction or linear obstruction?
- In a multi-species exhibit or other social grouping, could a larger or smaller animal become stuck or injured by the object or get hung up on it?
- Can it destroy an exhibit?
- If fecal material is used for enrichment, has it been determined to be free from harmful parasites?
- Is food enrichment included as part of the animals' regular diet in a manner that will reduce the risk of obesity?
- When introducing animals to conspecifics or in a multi-species exhibit, are there sufficient areas for them to escape undesirable interactions?
- Can the manner of enrichment presentation (i.e., one item or items placed in a small area) promote aggression or harmful competition?
- Has browse been determined to be non-toxic?
- Do animals show signs of allergies to new items (food, browse, substrates)?
- Does the enrichment cause abnormally high stress levels?
- Does the enrichment cause stimulation at a high level for extended periods of time that do not allow the animal natural down time in the species' normal repertoire (e.g., constant activity for public enjoyment when the animal would normally be inactive in its native habitat)?

AAZK Enrichment Committee, Enrichment Caution List

Dietary Enrichment

- Food enrichment, if uncontrolled, can lead to obesity, tooth decay and deviation from the normal diet can cause nutritional problems. Keepers can consult with the nutritionist or commissary staff to determine the best method of introducing novel food items.
- New food items introduced without analysis may cause colic, rumenitis or metabolic acidosis in ungulates.
- Food items can spoil and cause animal illness if left in the exhibit for extended periods of time. Enrichment food items should be removed within a reasonable amount of time to prevent spoilage.
- Animals can have adverse reactions to toxic plants and chemicals. Keepers should be able to correctly discern between toxic and browse plants, ensure that browse is free of fertilizers and herbicides and wash plants to remove free ranging bird and animal feces and debris.
- Foraging or social feedings may give rise to aggression and possible injuries within the animal population.
- Competition for enrichment items may lead to social displacement of subordinate animals. These concerns can be minimized by providing enough enrichment to occupy all of the animals within the population.
- Carcass feedings for omnivores and carnivores may be hazardous if the source of the carcass is not determined and appropriate precautions taken. Diseased animals, chemically euthanized animals or those with an unknown cause of death are not appropriate for an enrichment program. Freezing the carcasses of animals that are determined to be safe to feed to exhibit animals can help minimize the risk of parasitism and disease. Providing enough carcasses in group feedings can minimize competition and aggression within an exhibit.
- Carefully introducing a group of animals to the idea of social feedings can be done by moving carcass pieces closer together at each feeding until the animals are sharing one carcass. This can allow social carnivores to exhibit normal dominance posturing while minimizing the possibility of aggression. During live feedings, prey animals may fight back. Care should be taken to ensure such prey can only inflict superficial wounds on zoo animals.
- Cage furniture may interrupt flight paths or entangle horns and hooves if poorly placed. Careful planning can prevent this.
- If unsecured, some items may fall on an animal or be used as a weapon and cause injuries.
- If position is not thoughtfully considered, limbs and apparatus may provide avenues for escape or may block access into exhibit safety zones, leaving subordinate animals feeling trapped and vulnerable.
- Animals that crib or chew wood should be provided with non-toxic limbs and untreated wood furniture.
- Water features should be tailored to the inhabitants to prevent drowning and ensure that animals such as box turtles can right themselves if they flip over on their backs.
- Animals can be injured in filtration systems if water intake areas are not protected.
- Substrates should provide adequate traction and not cause an intestinal impaction if ingested.
- Caution should be exercised when ropes, cables, or chains are used to hang or secure articles to prevent animals from becoming entangled. Generally, the shortest length possible is recommended. Chain can be covered with a sheath such as PVC pipe; swivels can be used to connect the chain to the enrichment item to minimize kinking.

Olfactory Enrichment

- Scents from different animals or species can lead to aggression if there is an assertion of dominant animals or subordinate animals attempting to use enrichment to advance their status in the hierarchy.
- Animal feces used for olfactory enrichment should be determined to be parasite free through fecal testing and as with other animal by-products such as feathers, sheds, wool and hair, come from only healthy animals. Many of these items can be autoclaved for sterilization.
- Perfumes can be overwhelming to some animals (and keepers) and are therefore best used in open, ventilated areas.
- Some spices may be too strong or toxic to some animals.

Auditory Enrichment
- When provided with audio enrichment, animals may be less threatened by deflected sounds rather than those directed at the animals.
- Some animals may have adverse reactions to recordings of predator calls and should be closely observed when this type of enrichment is provided.
- Providing the animals with an option for escape or the means to mobilize for confrontation when predator calls are played can lessen the stress of this type of enrichment and allow the animals to investigate the sounds and their environment over a period of time.

Manipulable Enrichment
- Individual parts or enrichment devices may be swallowed resulting in choking or asphyxiation.
- If ingested, indigestible enrichment items may cause a gut impaction or linear obstruction.
- Broken items may have sharp edges that can cut an animal. Only items that are appropriate for the species should be provided. For example, some devices will hold up to the play of a fox but not a wolf
- When building or designing enrichment items from wood, it may be wise to use dovetail cuts and glue rather than screws and nails. Rounded corners and sanded edges can prevent the animals from getting splinters.
- Many paints and other chemicals are toxic if eaten. When providing enrichment involving paint or other chemicals, only non-toxic items should be used.
- If used, destructible items such as cardboard boxes and paper bags should be free of staples, tape, wax, strings or plastic liners. In general the Otter SSP advises against using these items.

Factors that should be considered when determining how often behavioral or environmental enrichment is offered include the species and individual(s) involved, as well as the physical characteristics of the exhibit. Large, complex exhibits with appropriate enclosure designs, substrates, and furnishings may offer ample opportunities for animals to exercise natural behaviors with infrequent enrichment (once daily). Other exhibits or individuals may require more frequent enrichment (multiple times per day). Husbandry staff should monitor all individuals in an exhibit and structure an enrichment schedule for the needs of those animals, providing them with opportunities several times a day to interact positively with their environment. Enrichment should never be offered on a regular schedule, instead times, items, and delivery methods should be rotated so there is always an element of novelty associated with each item or activity. It is important to note that the provision of well-designed, complex environments is the foundation of a successful enrichment program. This is particularly true for some of the more sensitive otter species such as *P. brasiliensis*, but applies to all of the otter species due to their inquisitive nature and high-activity level.

More Information: Appendix K provides a list of enrichment initiatives used at several institutions housing mustelids/otters. All enrichment items should be approved by the appropriate management staff, including the veterinarian, curator, horticulturist, and/or nutritionist. Appendix L provides a list of resources for enrichment and training. Institutions working with *P. brasiliensis* should consult the International Giant Otter Studbook Husbandry and Management Information and Guidelines (Sykes-Gatz 2005) for further information on the importance of exhibit design for this species and additional enrichment information.

8.3 Staff and Animal Interactions

Animal training and environmental enrichment protocols and techniques should be based on interactions that promote safety for all involved. Otters are easily trained and work well for positive reinforcement. Trained management and veterinary care behaviors include: weighing, crating, foot inspection, tooth inspection, injections, abdominal presentation and palpation, and tail inspections. It should be kept in mind that otters are capable of inflicting severe bites, particularly sexually mature females, and have been known to turn on their trainers. In general, otters should be trained in a protected contact situation (i.e., keeper and animal should be separated by a mesh barrier). There are some species (*A. cinereus*) or cases (*L. canadensis* particularly males) where an institution may feel that protected contact training may not be called for, but these decisions should be carefully evaluated on an ongoing basis.

Keeper safety should be kept in mind when designing otter exhibits. Animals should be shifted off exhibit for cleaning, maintenance, etc. This is particularly important for *P. brasiliensis*, as they can be dangerous. Animal safety should be considered and exhibits constructed, as far as possible, that prohibit the public from throwing potentially harmful items or food into the animals' space.

8.4 Staff Skills and Training

Staff members should be trained in all areas of animal behavior management. Funding should be provided for AZA continuing education courses, related meetings, conference participation, and other professional opportunities. A reference library appropriate to the size and complexity of the institution should be available to all staff and volunteers to provide them with accurate information on the behavioral needs of the otters with which they work.

The following skills are recommended for all animal caretakers involved in the management of otters of *ex-situ* populations:

- Keepers and managers should have an in-depth understanding of the species' natural history and the individual's history.
- Keepers and managers should have an in-depth understanding of the individual's behaviors, an understanding of the function of those behaviors, and the ability to describe those behaviors orally and in writing.
- Keepers should be able to recognize signs of illness and injury in the otter species they are working with and to communicate those signs orally or in writing to managers and veterinarians.
- Keepers should be able to accurately assess the appropriate level of cleanliness and safety of the animal's exhibit, holding area, and food-prep area.
- Keepers should have the skills to safely capture or restrain the otter species in question.
- Keepers should have some understanding of the species' natural diet and foraging style.
- Keepers and managers should have an understanding of enrichment concepts and have a commitment to consistently enhance the environments of the species in their care.
- Keepers should understand the concepts of animal learning and training, be able to use a variety of techniques (e.g., habituation, operant conditioning) to train the animals under their care, and to create a training plan (identifying training steps, cues, and criteria). See www.animaltraining.org for additional information.
- Managers should understand the concepts of animal learning and training, be able to coach keepers in all aspects of training, review their training plans, look for consistency among keepers in their training techniques, and help their teams prioritize training, enrichment, and other husbandry goals.
- Keepers and managers should have an understanding of the enclosure conditions and husbandry practices needed to maintain the otters' physical and behavioral health, as well as to promote a successful pup-rearing environment.

Chapter 9. Program Animals

9.1 Program Animal Policy

AZA Policies: AZA recognizes many public education and, ultimately, conservation benefits from program animal presentations. AZA's Conservation Education Committee's Program Animal Position Statement (Appendix D) summarizes the value of program animal presentations.

For the purpose of this policy, a program animal is described as an animal presented either within or outside of its normal exhibit or holding area that is intended to have regular proximity to or physical contact with trainers, handlers, the public, or will be part of an ongoing conservation education/outreach program.

Program animal presentations bring a host of responsibilities, including the welfare of the animals involved, the safety of the animal handler and public, and accountability for the take-home, educational messages received by the audience. Therefore, AZA requires all accredited institutions that give program animal presentations to develop an institutional program animal policy that clearly identifies and justifies those species and individuals approved as program animals and details their long-term management plan and educational program objectives.

AZA's accreditation standards require that the conditions and treatment of animals in education programs must meet standards set for the remainder of the animal collection, including species-appropriate shelter, exercise, sound and environmental enrichment, access to veterinary care, nutrition, and other related standards (1.5.4). In addition, providing program otters with options to choose among a variety of conditions within their environment is essential to ensuring effective care, welfare, and management. Some of these requirements can be met outside of the primary exhibit enclosure while the otter is involved in a program or is being transported. For example, housing may be reduced in size compared to a primary enclosure as long as the otter's physical and psychological needs are being met during the program; upon return to the facility the otter should be returned to its housing as described above.

> **AZA Accreditation Standard**
>
> **(1.5.4)** A written policy on the use of live animals in programs should be on file. Animals in education programs must be maintained and cared for by trained staff, and housing conditions must meet standards set for the remainder of the animal collection, including species-appropriate shelter, exercise, social and environmental enrichment, access to veterinary care, nutrition, etc. Since some of these requirements can be met outside of the primary enclosure, for example, enclosures may be reduced in size provided that the animal's physical and psychological needs are being met.

Otter SSP Program Animal Recommendations: *A. cinereus* and occasionally *L. canadensis* are used in training shows however, otters are not recommended as animal handling or off-site education animals.

Conservation Messages: Otters are excellent conservation and wildlife ambassadors; they are appealing to the public, active, and represent well the issues faced by many of the small carnivores.

<u>African spotted-necked otter: </u>"Spotted-Necked Otters are very aquatic and require permanent water sources with high fish densities. They prefer larger rivers, lakes, and swamps with open areas of water. They appear to only make use of fresh water habitats. Because they mainly hunt by sight, they need clear, unpolluted water where there are numerous small fish or fish, crabs and frogs. Long reeds, grass, and bushes are essential to provide cover; holes or other shelters are also needed. The most suitable habitat is the large fish-rich African lakes and the deep, clear areas of the Botswana Okavango.

"The distribution is large, but with some local declines. It occurs in all countries south of Sahara, from Senegal to Ethiopia and south to the Cape provinces where there is suitable habitat.

"The main threats throughout the range are habitat destruction by land drainage or pollution in response to increasing human population density and direct persecution as competitors for fish. There is some hunting for bushmeat and ceremonial practices. In some lakes, introduced large fish such as Nile Perch out-compete the small fish, which comprise the otters' historic food base, reducing prey availability.

Although international and national level legal protection is in place, enforcement is needed. There is a need for increasing local awareness of the species (IUCN Otter Specialist Group)."

The population is considered to be decreasing, and there are some indications that pressure from traditional medical uses, bushmeat consumption, and persecution as competitors for fish on some populations (Lake Victoria) may be increasing (J.Reed-Smith, personal knowledge).

Asian small-clawed otter: "Small-clawed otters prefer shallow water with a good food supply and moderate to low bank side vegetation. They demonstrate a high climatic and trophic adaptability, occurring from tropical coastal wetlands up to mountain streams. They make use of freshwater and peat swamp forests, rice fields, lakes, streams, reservoirs, canals, drainage ditches, rice paddies, mangroves, tidal pools, and along the coastline. In mountainous areas, they frequent swift-flowing forest streams with rocks and boulders. Their preferred food is crustaceans and mollusks. Across much of their range they are sympatric with Eurasian Otters (*Lutra lutra*), Smooth-Coated Otters (*Lutrogale perspicillata*) and Hairy-Nosed Otters (*Lutra sumatrana*), and there is clear evidence of niche separation between the species.

Although the species' range appears large, in the last decade actual distribution has shrunk, especially in the west, compared to historical records. They are currently found from the Himalayan foothills of Himachal Pradesh eastward throughout south Asia, extending up to Philippines and down through Indonesia. A small isolated subpopulation has been reported from southern Indian hill ranges of Coorg (Karnataka), Ashambu, Nilgiri and Palni hills (Tamil Nadu), and some places in Kerala. They were formerly found in Sri Lanka, but their current status there is unknown. The only areas in which these animals are known to be common today are Peninsular Malaysia, especially in Kedah, and in the western forests and southern marshes of Thailand.

The main threats throughout Asia are habitat destruction because of deforestation (loss of the smaller hill streams), agriculture (especially tea and coffee plantations in India, draining of peat swamp forests, and destruction of coastal mangroves for aquaculture), and settlement. Water courses are being polluted with pesticides from plantations and other intensive agriculture and heavy metals, affecting the gill-feeders on which this species depends. This interferes directly with otter physiology. Prey biomass is also being reduced by overexploitation, and the vast aquaculture industry regards otters as pests and persecutes them directly.

Although international and national level legal protection is in place, local legislation is needed. The impact of protection measures on livelihoods needs to be assessed and answered. Habitat protection and interpopulation corridors need to be established. Research on all aspects of this species biology and ecology is needed (IUCN Otter Specialist Group)." The population is considered to be decreasing.

Giant otter: "The wild population is estimated to have a total population of 1,000 to 5,000 individuals. In the past, giant otters were frequently hunted for their fur. This trade in giant otter pelts is one of the primary reasons giant otters are endangered in the wild. Because this species is active during the day, very vocal, and not afraid to approach humans, they were easy to hunt. Much of the population became decimated until efforts were finally made to protect them in the 1970's.

Although they still face other serious threats, habitat destruction and degradation, poaching, and unmanaged tourism are the primary threats faced by giant otter today. The areas in South America where the giant otter lives are rapidly being destroyed and degraded by logging, mining, exploitation of fossil fuels and hydroelectric power (dams), river and land pollution, and over-fishing. Some giant otter cubs are still being taken from the wild illegally to be kept as pets and they usually die in the hands of inexperienced caretakers. Tourists can disturb giant otters when they are rearing cubs. This can have a negative effect on how successfully parents rear their litters (IUCN Otter Specialist Group)." The population is considered to be decreasing.

North American river otter: This species is also referred to as the Nearctic otter. However, whatever you call it, the river otter represents a North American conservation success story. From a historic high when their range extended throughout most of North America, river otter populations fell until:

> "During the late 1800's and early 1900's, the synergistic effect of wetland destruction, pollution, and overexploitation for furs was devastating to North American river otter populations. Additional otter losses were due to road kills, accidental drowning in fishing nets and 'incidental take during beaver trapping'." (Foster-Turley et al. 1990)

By the 1970's, Nilsson & Vaughn (1978) estimated that the river otter was found in only 33% of its former range. They listed the causes of this as intensive trapping, pollution, destruction of habitat by clearing land, draining marshes, and channelizing streams. However, since 1976 over 4,000 otters have been reintroduced in 21 states and provinces throughout their former North American range (including Canada).

As a result of conservation and reintroduction measures the river otter has reoccupied much of its former range; this species it is still considered locally vulnerable, endangered, or extinct in some states and provinces. Many states and provinces where the populations were never threatened or have recovered allow a sustainable harvest of this species for fur. While "current harvest strategies do not pose a threat to maintaining otter populations, harvest may limit expansion of otter populations in some areas. Oil spills present a localized threat to otter populations, especially in coastal areas. Water pollution and other degradation of aquatic and wetland habitats may limit distribution of otters and pose long-term threats if enforcement of water quality standards are not maintained and enforced. Acid drainage from coal mines is a persistent water quality problem in some areas that eliminates otter prey and thereby inhibits recolonization or expansion of otter populations. Recently, there has been discussion of the long-term genetic consequences of reintroduction projects on remnant otter populations (Serfass et al. 1998). The threat of disease to wild otter populations is poorly understood and has received little study (Serfass et al. 1995). Similarly, many perceived threats to otters such as pollution and habitat alterations have not been rigorously evaluated. Additional research is needed to clearly delineate the impact that various forms of water pollution, agricultural and other development along riparian habitats, industrial and housing development in coastal areas, cumulative impacts related to loss or alterations of wetlands, large flood control structures, and interactions that these and other factors have on otter populations. Threats to otter populations in North America vary among regions and are influenced by type, distribution, and density of aquatic habitats and characteristics of human activities (IUCN Otter Specialist Group)."

Recently, concern has been growing that river otters are being demonized as voracious eaters of sport-fish species; this position has been used to justify elimination of river otters from some watersheds to placate special interest groups. Some research has been conducted on the percentage of sport fish species taken by river otter that indicates the true impact is not as great as claimed by many (Hamilton, 1999 & unpublished 2004); See Appendix L. The impact of river otter on sport-fish populations and the growing characterization of the river otter as the primary cause of localized falling fish populations is something that should be researched and monitored.

9.2 Institutional Program Animal Plans

AZA's policy on the presentation of animals is as follows: AZA is dedicated to excellence in animal care and welfare, conservation, education, research, and the presentation of animals in ways that inspire respect for wildlife and nature. AZA's position is that animals should always be presented in adherence to the following core principles:

- Animal and human health, safety, and welfare are never compromised.
- Education and a meaningful conservation message are integral components of the presentation.
- The individual animals involved are consistently maintained in a manner that meets their social, physical, behavioral, and nutritional needs.

AZA-accredited institutions which have designated program animals are required to develop their own Institutional Program Animal Policy that articulates and evaluates the program benefits (see Appendix E for recommendations). Program animals should be consistently maintained in a manner that meets their social, physical, behavioral, and nutritional needs. Education and conservation messaging must be an integral component of any program animal demonstration (1.5.3).

Animal care and education staff should be trained in otter-specific handling protocols, conservation and education messaging techniques, and public interaction procedures. These staff members should be competent in recognizing stress or discomfort behaviors exhibited by the program otters and should be able to address any safety issues that arise.

Program animals that are taken off zoo or aquarium grounds for any purpose have the potential to be exposed to infectious agents that could spread to the rest of the institution's healthy population. AZA-accredited institutions must have adequate protocols in place to avoid this (1.5.5).

AZA Accreditation Standard

(1.5.3) If animal demonstrations are a part of the institution's programs, an education and conservation message must be an integral component.

AZA Accreditation Standard

(1.5.5) For animals used in offsite programs and for educational purposes, the institution must have adequate protocols in place to protect the rest of the collection from exposure to infectious agents.

Careful consideration must be given to the design and size of all program animal enclosures, including exhibit, off-exhibit holding, hospital, quarantine, and isolation areas, such that the physical, social, behavioral, and psychological needs of the species are met and species-appropriate behaviors are facilitated (10.3.3; 1.5.2).

Animal transportation must be conducted in a manner that is lawful, safe, well-planned and coordinated, and minimizes risk to the animal(s), employees, and general public (1.5.11).

9.3 Program Evaluation

AZA-accredited institutions which have Institutional Program Animal Plan are required to evaluate the efficacy of the plan routinely (see Appendix E for recommendations). Education and conservation messaging content retention, animal health and well-being, guest responses, policy effectiveness, and accountability and ramifications of policy violations should be assessed and revised as needed.

AZA Accreditation Standard

(10.3.3) All animal enclosures (exhibits, holding areas, hospital, and quarantine/isolation) must be of a size and complexity sufficient to provide for the animal's physical, social, and psychological well-being; and exhibit enclosures must include provisions for the behavioral enrichment of the animals.

AZA Accreditation Standard

(1.5.2) Animals should be displayed, whenever possible, in exhibits replicating their wild habitat and in numbers sufficient to meet their social and behavioral needs. Display of single specimens should be avoided unless biologically correct for the species involved.

AZA Accreditation Standard

(1.5.11) Animal transportation must be conducted in a manner that is safe, well-planned and coordinated, and minimizes risk to the animal(s), employees, and general public. All applicable local, state, and federal laws must be adhered to.

Chapter 10. Research

10.1 Known Methodologies

AZA believes that contemporary animal management, husbandry, veterinary care and conservation practices should be based in science, and that a commitment to scientific research, both basic and applied, is a trademark of the modern zoological park and aquarium. AZA-accredited institutions have the invaluable opportunity, and are expected, to conduct or facilitate research both in *in-situ* and *ex-situ* settings to advance scientific knowledge of the animals in our care and enhance the conservation of wild populations. For otters, this knowledge might be achieved by AZA Small Carnivore Taxon Advisory Group (TAG) or AZA Otter Species Survival Plan® (SSP) Program sponsored research, conducting original research projects, affiliating with local universities, and/or employing staff with scientific credentials (5.3).

> **AZA Accreditation Standard**
>
> **(5.3)** Institutions should maximize the generation of scientific knowledge gained from the animal collection. This might be achieved by participating in AZA TAG/SSP sponsored research when applicable, conducting original research projects, affiliating with local universities, and/or employing staff with scientific credentials.

Research investigations, whether observational, behavioral, physiological, or genetically based, should have a clear scientific purpose with the reasonable expectation that they will increase our understanding of the otter species being investigated and may provide results which benefit the health or welfare of otters in wild populations. Many AZA-accredited institutions incorporate superior positive reinforcement training programs into their routine schedules to facilitate sensory, cognitive, and physiological research investigations and these types of programs are strongly encouraged by the AZA.

AZA-accredited institutions are required to have a clearly written research policy that identifies the types of research being conducted, methods used, staff involved, evaluations of the projects, the animals included, and guidelines for the reporting or publication of any findings (5.2). Institutions must designate e a qualified individual designated to oversee and direct its research program (5.1). If institutions are not able to conduct in-house research investigations, they are strongly encouraged to provide financial, personnel, logistical, and other support for priority research and conservation initiatives identified by Taxon Advisory Groups or Species Survival Plans®.

> **AZA Accreditation Standard**
>
> **(5.2)** Institutions must have a written policy that outlines the type of research that it conducts, methods, staff involvement, evaluations, animals to be involved, and guidelines for publication of findings.

> **AZA Accreditation Standard**
>
> **(5.1)** Research activities must be under the direction of a person qualified to make informed decisions regarding research.

The AZA Otter SSP, which falls under the Small Carnivore TAG, is the AZA entity tasked with recommending species for management by member institutions. The IUCN/SSC Otter Specialist Group (OSG) serves as an international focus of information sharing for these species. Information on current and past field work can be found at the OSG web site, www.otterspecialistgroup.org. Several universities (e.g. Frostburg State University, University of Wyoming) have professors specializing in otter research. However, situations change and students interested in pursuing work with otters always should research current specialists working in the area and institutions with which they are affiliated. The AZA Otter SSP is compiling a list of institutions involved in or supporting otter research and conservation work. This information will become available in future versions of this document. For input on ongoing research or areas requiring further investigation please refer to the OSG web site or contact the AZA Otter SSP Chair.

Effective Research Methodologies: All sound research approaches should be viable for use on otters, as long as they are not too invasive, require extensive surgery, or cause pain or discomfort. The following methodologies are routinely used by researchers looking at otters:

- Behavioral observation
- Latrine surveys
- Fecal hormone analysis represents an invaluable tool for assessing the reproductive status of individuals and populations in a completely noninvasive manner.
- Fecal DNA analysis

10.2 Future Research Needs

This Otter Care Manual is a dynamic document that will need to be updated as new information is acquired. Knowledge gaps have been identified throughout the Manual and a general list is included in this section to promote future research investigations. Knowledge gained from areas will maximize AZA-accredited institutions' capacity for excellence in otter care and welfare as well as enhance conservation initiatives for all species.

Specific Areas of *Ex-situ* Population Research Needed by Manual Heading:

<u>Chapter 1: Ambient Environment</u>

1.1 Water temperature: More detailed research is required into optimal water temperature levels for the tropical otter species.

1.2 Light: The AZA Small Carnivore TAG is unaware of any hard data on the impact of light intensity on otter health or reproduction; this should be investigated in the future. Similarly, there are no available data on possible deleterious effects of less than full spectrum light on a long-term basis. Health data collected by institutions housing otters in environments with different light intensities and spectrums would be a useful foundation for this research.

1.3 Air: Pupping dens may well need higher rates of air exchange in order to maintain air quality and/or low humidity. It should be noted, however, that no work has been done specifically targeting air change rates for otter exhibits or dens.

1.3 Water - Coliform: There are no standards for coliform yet established for fresh-water otter pools. At this time, it is suggested that coliform levels be maintained at or lower than levels established for rescued pinnipeds by NOAA.

1.3 Water - pH Levels: Further research is needed into the impact, if any, of pH on otters. While research is desirable it is not recommended on these wildlife species and instead caution should be exercised when using chlorine in otter pools.

1.4 Sound: While there is no evidence that low level background noise is disruptive to otters, loud noises can be frightening to them, and high-pitched, long-term noise should be avoided. Otters' hearing is considered to be good but nothing is known definitively about their hearing acuity or frequency ranges heard. Both of these are areas needing further research.

<u>Chapter 3: Transport</u>

3.2 Transport temperature: Identifying appropriate transport temperatures for *L. maculicollis* requires further research. Current recommendations are based on *A. cinereus*, but it is not known whether this is suitable.

<u>Chapter 4: Social Environment</u>

4.1 Single-sexed groups: Information on the success of single-sexed groups for *A. capensis* is unknown at this time; further research is required. Information on the maintenance of family groups of *L. maculicollis* over the long term has not been well documented; this should be monitored to assist in future recommendations for this species. Information from institutions attempting this type of social grouping should be shared with the AZA Small Carnivore TAG.

<u>Chapter 5: Nutrition</u>

5.1 Seasonal changes in nutritional needs: Some institutions report seasonal changes in appetite of some otters, but not in the majority of animals. Further research in this area, and in changing seasonal nutritional requirements for otters, is required.

5.1 Nutritional related diseases: Further research on nutritional requirements of otters and nutrition related diseases.

5.2 Sample diets: The one best diet for any of the otters of *ex-situ* populations has not been found and requires further research. Identifying health issues associated with the provision of different diets should continue, and data shared with the AZA Small Carnivore TAG.

5.3 Nutrition evaluation: The AZA Otter SSP is currently beginning work on a body-condition matrix that can be used to help assess proper weight and condition for otters. At this time there are no known tools for performing clinical nutritional evaluations of otters; this would be a useful area for future research.

Chapter 6: Veterinary Care

6.7 Management of diseases and disorders: Little information on common diseases and disorders for *A. capensis* and *L. maculicollis* is available, and more research is required for these species. Institutions housing these species should record all health issues seen in these species so that a database can be created on health issues and concerns.

Chapter 7: Reproduction

7.1 Reproductive physiology and behavior: Some facilities have reported a small amount of estrus-associated bleeding from the vulva in *L. canadensis*, while others have not seen this; additionally, previous studies attempting to identify behavioral changes associated with estrus were unsuccessful. Information on female estrus behavior would be a helpful area of research. These are areas that require further research and can be achieved through simple observational research during estrus for animals in this species.

7.1 Reproductive physiology and behavior: In both NARO and ASCO, additional research is needed to improve endocrine monitoring of estrogen metabolites to further address these questions about ovarian cyclicity and ovulatory mechanisms.

7.1 Reproductive physiology and behavior: Research utilizing techniques to identify reproductive state in these species is ongoing. At this time, it appears that ELISA protocols for testing hormonal secretions in fecal samples are successful in determining pregnancy in Asian small-clawed and North American river otters (H.Bateman, unpublished data). The reproductive physiology advisor for the AZA Otter SSP, should be contacted for more information.

7.1 Reproductive physiology and behavior: Further study is required to clarify if there is a genetic component to the seasonal regulation of estrus in females and testosterone production in NARO males.

7.2 Pregnancy and parturition: Pseudopregnancy has been reported for most otter species and is an area that requires further research

7.3 Pup development: At this time there is no information available on pup development *of A. capensis;* additional data on the development of *L. maculicollis* pups is also required. Institutions housing these species (*A. capensis* is a phase-out but still maintained by a few member institutions) should set-up an observational research study for this species when reproduction is attempted.

7.4 Nursery groups: Nursery groups are not reported for *L. maculicollis* in the wild, but further field research would help to determine if this aspect of parental care is applicable or appropriate for *ex-situ* population management of this species.

7.5 Hand-rearing: More research is required to determine body temperature norms for young of all otter species. This information should be collected by all facilities hand-rearing otter pups and submitted to the AZA Otter SSP and Small Carnivore TAG.

7.5 Contraception: Research on the effects that GnRH agonists have on future reproductive abilities when provided to prepubertal otters is also needed, as current research is based on studies using domestic cats.

Chapter 8: Behavioral Management

8.1 Training: Training animals to station may be beneficial when attempting introductions, but this has not been tried with any of the otters.

Other Areas of Research:

Giant otters: The following priorities were established for the giant otter during the 2004 meeting of the IUCN/SSC Otter Specialist Group:

1. To continue the assessment of predator-prey relationships, including conflicts with subsistence and commercial fishermen.
2. To evaluate the positive and negative impact of tourism on different habitats and implement management guidelines in order to maximize the benefits.
3. To encourage the development of a long-term research and conservation project in the Llanos of Venezuela or Colombia.
4. To undertake collaborations between field scientists, zoos, and genetic labs to evaluate the potential use of genetic analysis tools in giant otter research.

Field Research: Additional research is needed to clearly delineate the impact that various forms of water pollution, agricultural and other development along riparian habitats, industrial and housing development in coastal areas, cumulative impacts related to loss or alterations of wetlands, large flood control structures, and interactions that these and other factors have on otter populations (N.A. river otter)

The impact of river otter on sport-fish populations and the growing characterization of the river otter as the primary cause of localized falling fish populations is something that should be researched and monitored.

General Areas of Research Needed:

- Nutritional needs to include nutrition related diseases
- Reproductive research – reproductive physiology research is ongoing (Helen Bateman, C.R.E.W.) and should continue for all otter species.
- Veterinary issues – to include efficacy of vaccines, titers, and common causes of death in *ex-situ* populations. All otters should be necropsied and results sent to the veterinary advisor.
- Status in the wild – institutions should assist the AZA Otter SSP in raising awareness and funds to adequately assess the status of wild otter populations of all species.
- *Ex-situ* population behavior studies – to assess extent and type of stereotypes observed in the *ex-situ* population, to assess breeding/parturition behavior; to assess optimum group size and composition for otters of *ex-situ* populations.

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It is important to note that the recommendations contained in this manual are fluid and may need to be modified to offer best practice care to particular individual animals. The most important thing to remember is to know your animals and pay attention to their individual needs. Questions regarding any of the information contained in this manual may be directed to Jan Reed-Smith (lontracat@live.com) or Dusty Lombardi (dusty.lombardi@columbuszoo.org).

For additional information on otters, otter conservation, otter research, and otter care we recommend the IUCN/SSC Otter Specialist Group web site at: www.otterspecialistgroup.org or the International Otter Survival Fund at: www.otter.org.

References

AAFCO. 1994. Association of American Feed Control Officials, Inc.

APHIS (Animal and Plant Health Inspection Service). 2007. Record of Acquisition, Disposition or Transport of Animals (Other than Dogs and Cats). www.aphis.usda.gov/animal_welfare/downloads/forms/aph7020.pdf

Anon. 1981. Outdoor Air Requirements for Ventilation (ASHRAE Standard 62-1981) 3.1 Commercial Facilities (Offices, Stores, Shops, Hotels, Sports Facilities, etc.). American Society of Heating, Refrigeration, Air Conditioning Engineers, 74-85.

Autuori MP, Deutsch LA. 1977. Contribution to the knowledge of the giant Brazilian otter, *Pteronura brasiliensis* (Gmelin 1788), Carnivora, Mustelidae. Zoological Garten 47: 1-8.

AWR (Animal Welfare Regulations). 2005. Animal Welfare Act, 7 U.S.C. Animal Welfare Regulations, 9 CFR Chapter 1, Subchapter A, Parts 1-4.

Ayars GH, Altman LC, Frazier CE, Chi EY. 1989. The toxicity of constituents of cedar and pine woods to pulmonary epithelium. Journal of Allergy Clinical Immunology 83: 610-618.

AZA (Association of Zoos and Aquariums). 2008. AZA Accreditation Standards and Related Policies. Association of Zoos and Aquariums, Silver Spring, MD.

Baitchman EJ, Kollias GV. 2000. Clinical anatomy of the North American river otter (*Lontra canadensis*). Journal of Zoo and Wildlife Medicine 31:473–483.

Bateman HLJ, Bond B, Campbell M, Barrie M, Riggs G, Snyder B, Swanson WF. 2005. Characterization of seasonal seminal traits in the North American river otter (*Lontra canadensis*). Proceedings of the Society for the Study of Reproduction, 124 (abst. 192).

Bateman HLJ, Bond B, Campbell M, Barrie M, Riggs G, Snyder B, Swanson, WF. 2009. Characterization of basal seminal traits and reproductive endocrine profiles in North American river otters and Asian small-clawed otters. Zoo Biology 28:107-126

Beckel AL. 1982. Behavior of free-ranging and captive river otters in northcentral Wisconsin. Ph.D. dissertation Univ. Minn., Madison, 198pp.

Ben-David M, Williams TM, Ormseth OA. 2000. Effects of oiling on exercise physiology and diving behavior in river otters: a captive study. Canadian Journal of Zoology 78: 1380-1390.

Ben-David M, Duffy LK, Bowyer RT. 2001a. Biomarker responses in river otters experimentally exposed to oil contamination. Journal of Wildlife Diseases 37(3): 489-508.

Ben-David M, Duffy LK, Blundell GM, Bowyer RT. 2001b. Natural exposure of coastal river otters to mercury: relation to age, diet, and survival. Environmental Toxicology & Chemistry 20(9): 1986-1992.

Ben Shaul, DM. 1962. Notes on hand-rearing various species of mammals. International Zoo Yearbook Vol. IV: 300-342.

Benza D, Cantabene S, Foland N, Sears R, Reed-Smith J. 2009. Summary of Husbandry Guidelines for African Spotted-necked Otters in Captivity. IUCN/SSC Otter Specialist Group, Otters in Captivity Task Force. 13pgs. lontracat@live.com; www.otterspecialistgroup.org/Library/TaskForces/OCT.html

Bernard JB, Allen MA. 1997. Feeding captive piscivorous animals: nutritional aspects of fish as food. Nutrition Advisory Group Handbook. Fact Sheet 005.

Bertschinger HJ, Asa CS, Calle PP, Long JA, Bauman K, DeMatteo K, Jochle W, Trigg TE, Human A. 2001. Control of reproduction and sex related behaviour in exotic wild carnivores with the GnRH analogue deslorelin: preliminary observations. Journal of Reproduction and Fertility, Supplement 57: 275-283.

Best RC. 1985. Otters and manatees conservation. Conclusions of the 1 Reunion de Trabajo de Expertos en Mamiferos Acuaticos de America del Sur. Buenos Aires, Argentina. 25-29 Jun, 1984. 64-75.

Bitgood S, Patterson D, Benefield A. 1986. Understanding your visitors: ten factors that influence visitor behavior. Annual Proceedings of the American Association of Zoological Parks and Aquariums, 726-743.

Bitgood S, Patterson D, Benefield A. 1988. Exhibit design and visitor behavior. Environment and Behavior 20(4): 474-491.

Blum P. 2004. Florida Aquarium, Hand-rearing *L. canadensis* information. AZA Otter SSP Otter Keeper Workshop, Frostburg, Maryland CD. Columbus Zoo and Aquarium, Columbus, Ohio.

Blundell GM, Kern JW, Bowyer RT, Duffy LK. 1999. Capturing river otters: a comparison of Hancock and leg-hold traps. Wildlife Society Bulletin 27(1): 184-192.

Blundell G, Ben-David M, Bowyer RT. 2002a. Sociality in river otters: cooperative foraging or reproductive strategies. Behavioral Ecology 13(1): 134-141.

Blundell GM, Ben-David M, Groves P, Bowyer RT, Geffen E. 2002b. Characteristics of sex-biased dispersal and gene flow in coastal river otters: Implications for natural recolonization of extirpated populations. Molecular Ecology 11(3): 289-303.

Blundell GM, Ben-David M, Groves P, Bowyer RT, Geffen E. 2004. Kinship and sociality in coastal river otters: are they related? Behavioral Ecology 15(5): 705-714.

Bowyer RT, Blundell GM, Ben-David M, Jewett SC, Dean TA, Duffy LK. 2003. Effects of the Exxon Valdez oil spill on river otters: injury and recovery of a sentinel species. Wildlife Monographs 153: 1-53.

Burkhart CA, Robinson JL. 1978. High rat pup mortality attributed to the use of cedar wood shavings as bedding. Lab Animal 12: 221-222.

Calle PP, Stetter MD, Raphael BL, Cook RA, McClave C, Basinger JA, Walters H, Walsh K. 1997. Use of depot leuprolide acetate to control undesirable male associated behaviors in the California sea lion (*Zalophus californianus*) and California sea otter (*Enhydra lutris*). In Gregory A. Lewbart (Ed.), Proceedings of the International Association of Aquatic Animal Medicine, 28: 6-7.

Calle PP, McClave C, Basinger JA, Walters H, Raphael BL, Cook RA. 1998. Use of depot leuprolide and cyproterone to control aggression in an all male California sea otter (*Enhydra lutris nereis*) colony. In C.K. Baer (Ed.), Proceedings AAZV and AAWV Joint Conference, 375-377.

Carter SK, Rosas FCW. 1997. Biology and conservation of the giant otter *Pteronura brasiliensis*. Mammal Review 27: 1-26.

Chanin P. 1985. The Natural History of Otters. Facts on File Publications, New York, New York. pg 179.

Churchman D. 1985. How and what do recreational visitors learn at zoos? Annual Proceedings of the American Association of Zoological Parks and Aquariums, 160-167.

Conway W. 1995. Wild and zoo animal interactive management and habitat conservation. Biodiversity and Conservation 4: 573-594.

Corredor G, Muñoz TN. 2004. Reproductive behavior and biology of the giant river otter (*Pteronura brasiliensis*) at the Cali Zoo. Unpublished report.

Crait JR, Blundell GM, Ott KE, Herreman JK, Ben-David M. 2006. Late seasonal breeding of river otters in Yellowstone National Park. The American Midland Naturalist 156(1) July 2006:189-192.

Crissey SD. 1998. Handling Fish Fed to Fish-eating Animals: A Manual of Standard Operating Procedures. U.S. Department of Agriculture. Agriculture Research Service, National Agricultural Library.

Davis H, Aulerich R, Bursian S, Sikarskie J, Stuht J. 1992. Feed consumption and food transit time in northern river otters (*Lutra canadensis*). Journal of Zoo and Wildlife Medicine. 23: 241-244.

Davison VM, McMahon L, Skinner TL, Horton CM, Parks BJ. 1993. Animals as actors: take 2. Annual Proceedings of the American Association of Zoological Parks and Aquariums, 150-155.

Dierenfeld ES, Alcorn HL, Jacobsen KL. 2002. Nutrient composition of whole vertebrate prey (excluding fish) fed in zoos. National Agricultural Library Z7994.Z65, 20 pp. www.nal.usda.gov/awic/zoo/WholePreyFinal02May29.pdf

Dunstone N. 1998. Adaptations to the semi-aquatic habit and habitat. In: Behaviour and Ecology of Riparian Mammals. Dunstone N, Gorman ML (Eds.). Cambridge Univ. Press, Cambridge, England, 1-16.

Duplaix N. 1980. Observations on the ecology and behavior of the giant river otter, *Pteronura brasiliensis* in Surinam. Revue Ecologique (Terre Vie) 34: 495-620.

Duplaix N. 2002. Giant Otter Final Report WWF – Guianas Rapid River Bio-assessments and Giant Otter Conservation Project, FG-40 FY2002.

Duplaix-Hall N. 1972. Notes on maintaining river otter in captivity. International Zoo Yearbook 12: 178-181.

Duplaix-Hall N. 1975. “River Otters in Captivity” In: Breeding Endangered Species in Captivity. R.D. Martin, Ed. Academic Press, London.

Earle KE, Smith PM. 1993. A balanced diet for dogs and cats. In: The Waltham Book of Companion Animal Nutrition. Burger IH (Ed.). Pergamon Press, New York, NY.

Engelhardt FR, Geraci JR. 1978. Effects of experimental vitamin E deprivation in the Harp seal (*Phoca groenlandica*). Canadian Journal of Zoology 56: 2186-2193.

Estes J. 1989. Adaptions for aquatic living by carnivores. In: Carnivore Behavior, Ecology, and Evolution, Gittleman J (Ed.). Comstock Publ. Assoc., Ithaca, NY, 242-282.

Evans RH. 1985. Rearing orphaned wild mammals. In: Proceedings of the Fourth and Fifth Dr. Scholl Conferences on the Nutrition of Captive Wild Animals. (Dec. 7 & 8, 1984; Dec. 13 & 14, 1985 Lincoln Park Zoological Gardens, Chicago, IL.) Meehan T DVM, Allen ME Ph.D. (Eds.), 122-151.

Fernandez-Moran, J. 2003. Mustelidae. In: Zoo and Wild Animal Medicine 5th Edition. Fowler M, Miller R (Eds.). Saunders, St. Louis, Missouri, 501-515.

Flugger M. 1997. Haltung, zucht und erkrankungen von riesenottern (*Pternonura brasiliensis*) in Carl Hagenbeck's Tierpark. Verh. Ber. Erkg. Zootiere 38: 125-130.

Foster-Turley P. 1990. Otters in captivity. In: Otters: an action plan for their conservation. Foster-Turley P, Macdonald S, Mason C (Eds.). International Union for Conservation of Nature (IUCN), Gland Switzerland.

Foster-Turley P, Macdonald S, Mason C. (Eds.). 1990. Otters: an action plan for their conservation. International Conservation of Nature (IUCN), Gland, Switzerland.

Gatz V. 2002. Protected contact training of the giant otter (*Pteronura brasiliensis*). In: Proceedings of the VIIth Internationa Otter Colloquium, March 14 – 19, 1998. Trebon, Czech Republic. Dulfer R et al. (Eds.). IUCN/SSC Otter Specialist Group, Aktion Fischotterschutz, Hankensbüttel, Germany, 69-71.

Gorman TA, Erb JD, McMillan BR, Martin DJ. 2006. Space use and sociality of river otters (*Lontra canadensis*) in Minnesota. Journal of Mammalogy 87(4):740-747.

Grant K. 2004. *Erethizon dorsatum*. AZA Porcupine Husbandry Manual. www.glenoakzoo.org/Rodent%20TAG.htm.

Grant K. 2005. Hand-rearing cheetah (*Acinonyx jubatus*) cubs: milk formulas. Animal Keepers' Forum 32(7/8): 294-302

Hagenbeck C, Wünnemann K. 1992. Breeding the giant otter *Pteronura brasiliensi)* at Carl Hagenbeck's Tierpark. International Zoo Yearbook 31: 240-245.

Hancocks D. 1980. Bringing nature into the zoo: inexpensive solutions for zoo environments. International Journal for the Study of Animal Problems 1:170-177.

Hansen H. 2004. The effects of diet, extraction technique and microsatellite primers on genotyping success and error rates of river otter (*Lontra canadensis*) fecal DNA. Thesis. Univ. of Wyoming, Laramie.

Hamilton D. 1999. Missouri Fish and Wildlife (www.mdc.mo.gov/conmag/1999/11/40.htm) MDC Online November 1999, Vol. 60, Issue 11.

Hamilton D. 2004 unpublished. Paper presented at IXth International Otter Colloquium, Frostburg, Maryland, June 2004.

Hamilton WJ, Eadie WR. 1964. Reproduction in the otter, *Lutra canadensis*. Journal of Mammalogy 45(2): 242-252.

Harris CJ. 1968. Otters, a study of the recent Lutrinae. Weidenfeld & Nicolson, London, England, 397.

Hoover JP, Tyler RD. 1986. Renal function and fractional clearances of American river otters (*Lutra canadensis*). Journal of Wildlife Disease 22(4): 547-556

IATA (International Air Transport Association). 2007. IATA Live Animal Requirements, 34th edition. International Air Transport Association, Montreal-Geneva (ISBN978-92-9229-058-0), 345-346.

ITIS. 29 June 2009. Integrated Taxonomic Information System. (www.itis.gov).

IUCN Otter Specialist Group. Web site, 2009. (www.otterspecialistgroup.org).

IUCN/SSC. 1992. Otters. World conservation Union (IUCN), Gland, Switzerland, 32.

Iversen JA. 1972. Basal energy metabolism of mustelids. Journal of Comparative Physiology 81: 341-344.

Jenness R, Sloan RE. 1970. The composition of milks of various species: a review. Dairy Science. Abstracts 32(10): 599-612, Review Article No. 158.

Johnson SA, Berkley KA. 1999. Restoring river otters in Indiana. Wildlife Society Bulletin 27(2): 419-427.

Johnston RJ. 1998. Exogenous factors and visitor behavior: a regression analysis of exhibit viewing time. Environment and Behavior 30(3): 322-347.

Kiku M, Byeong-Kirl B, Chae-Woong L.. 2003. Eurasian otter (*Lutra lutra*), a definitive host for *Dirofilaria immitis*. Journal of Zoo and Wildlife Medicine 34: 200-201.

Kleiber M. 1947. Body size and metabolic rate. Physiological Review 27: 511-541.

Koepfli KP, Wayne RK. 1998. Phylogenetic relationships of otters (Carnivora: Mustelidae) based on mitochondrial cytochrome *b* sequences. Journal of the Zoological Society of London 246: 401-416.

Koepfli KP, Wayne RK. 2003. Type I STS markers are more informative than cytochrome *b* in phylogenetic reconstruction of the Mustelidae (Mammalia: Carnivora). Systematic Biology. 52(5). October 2003. 571-593.

Kruuk H. 1995. Wild otters: predation and populations. Oxford University Press, Oxford England & New York, New York.

Kruuk H, Goudswaard PC. 1990. Effects of changes in fish populations in Lake Victoria on the food of otters (*Lutra maculicollis* Schinz and *Aonyx capensis* Lichtenstein). African Journal of Ecology 28: 322-329.

Kruuk H, Moorehouse A. 1990. Seasonal and spatial differences in food selection by otters *Lutra lutra* in Shetland. Journal of Zoology 221: 621-637.

Legrand-Defretin V, Munday H. 1993. Feeding Dogs and Cats for Life. In: Burger IH. (Ed.), The Waltham Book of Companion Animal Nutrition. Pergamon Press, New York, NY, 57-68.

Lewis J. 1995. Veterinary considerations. In: Husbandry Handbook for Mustelids. ABWAK, 12 Tackley Road, Eastville, Bristol BS56UQ, U.K.

Liers EE. 1951. Notes on the river otter (*Lutra canadensis*). Journal of Mammalogy. 32(1): 1-9.

Lombardi C, Meyerson R, Reed-Smith J, Crissey S, Maslanka M, Petrini K, Coker M, Morris D, Kuenzer D, Willis K, Engfer S, Doherty J. 1998. Asian Small-clawed Otter (*Aonyx cinerea*) Husbandry Manual. Columbus Zoo and Aquarium, Columbus, Ohio. 102 pages.

Lombardi C. 2004. AZA Otter Keeper Workshop, June 3-4, 2004, Frostburg, Maryland.

MacMillen O. 1994. Zoomobile effectiveness: sixth graders learning vertebrate classification. Annual Proceedings of the American Association of Zoological Parks and Aquariums, 181-183.

Marcato de Oliveira S. 1995. Compartamento social e reprodutive de *Pteronura brasiliensis* (Mammalia – Mustelidae) em cativeiro no zoologico da UFMT. Cuiaba – MT. Unpublished Report.

Maslanka MT, Crissey SD. 1998. Nutrition and diet. In: The Asian small-clawed Otter Husbandry Manual. Columbus Zoological Gardens, Columbus, OH.

Mckay K. 2009. Basics of Otter Training. IUCN/SSC Otter Specialist Group, Otters in Captivity Task Force. 12pgs. lontracat@live.com; www.otterspecialistgroup.org/Library/TaskForces/OCT.html

McTurk D, Spelman L. 2005. Hand-rearing and rehabilitation of ophaned wild giant otters, *Pteronura brasiliensis*, on the Rupununi River, Guyana, South America. Zoo Biology 24: 153-167.

Mead R. 1989. Physiology and evolution of delayed implantation in carnivores. In: Carnivore Behaviour, Ecology and Evolution, Gittleman J (Ed.). Comstock Press, Ithaca, NY, 437-464

Meehan T. 1994. Handrearing felids. In: AZA Infant Diet Notebook. American Zoo and Aquarium Assoc. Silver Springs, MD.

Meier JE. 1985. Hand rearing techniques for neonatal exotic animals. In: Proceedings of the Fourth and Fifth Dr. Scholl Conferences on the Nutrition of Captive Wild Animals. (Dec. 7 & 8, 1984; Dec. 13 & 14, 1985 Lincoln Park Zoological Gardens, Chicago, IL.) Meehan T DVM, Allen ME Ph.D. (Eds.), 36-48.

Meier JE. 1986. Neonatology and hand-rearing of carnivores. In: Zoo & Wild Animal Medicine, 2nd ed. Fowler M. (Ed.). W.B. Saunders Co., Philadelphia, PA.

Melquist WE, Hornocker MG. 1983. Ecology of river otters in west central Idaho. The Wildlife Society Monographs: 83.

Melquist WE, Dronkert AE. 1987. River otter. In: Wild Furbearer Management and Conservation in North America. Novak M, Baker JA, Obbard ME, Mallock B (Eds.), 626-641.

Merck Veterinary Manual, 6th Edition. 1986. A Handbook of Diagnosis, Therapy, and Disease prevention and Control for the Veterinarian. Merck & Co., Inc. Rahway, N.J.

Moore D. 1997. AZA Minimum Husbandry Guidelines for Mammals: Mustelids. AZA Mammal Standards Task Force, 1997. 7pg.

Morgan JM, Hodgkinson M. 1999. The motivation and social orientation of visitors attending a contemporary zoological park. Environment and Behavior 31(2): 227-239.

Muir S. 2003. Hand-rearing guidelines for small carnivores. Shaldon Wildlife Trust, UK

Munson L. 2006. Contraception in felids. Theriogenology 66: 126-34.

Munson L, Bauman JE, Asa CS, Jochle W, Trigg TE. 2001. Efficacy of the GnRH analogue deslorelin for suppression of oestrus cycle in cats. Journal of Reproduction and Fertility, Supplement 57: 269-273.

National Research Council (NRC) 1982. Nutrient Requirements of Mink and Fox. National Academy Press, Washington, D.C.

National Research Council (NRC) 1986. Nutrient Requirements of Cats. National Academy Press, Washington, D.C.

National Research Council (NRC) 2006. Nutrient Requirements of Cats and Dogs. National Academy Press, Washington, D.C.

Neiffer DL, Klein EC, Calle PP, Terrell SP, Walker RL, Todd D, Vice CC, Marks SK. 2002. Mortality associated with melarsomine dihydrochloride administration in two North American river otters (*Lontra canadensis*) and a red panda (*Ailurus fulgens fulgens*). Journal of Zoo & Wildlife Medicine 33(3): 242-248.

Nilsson G, Vaughan AS. 1978. A turning point for the river otter. National Parks and Conservation Magazine 52(4):10 – 15.

NOAA, 2006. National Oceanic and Atmospheric Administration. Interim Policies and Best Practices Marine Mammal Response, Stranding, and Release Standards for Rehabilitation Facilities. L.Gage. www.nmfs.noaa.gov/pr/pdfs/health/rehab_facilities.pdf.

Ormseth OA, Ben-David M. 2000. Ingestion of crude oil: effects on digesta retention times and nutrient uptake in captive river otters. Journal of Comparative Physiology B Biochemical Systemic & Environmental Physiology 170(5-6): 419-428.

Osmann C, Wisser J. 2000. Hypothyreose bei Riesenotter-Welpen-eine hereditäre problematic? Teil 1: Eltern- und Handaufzuchten im klinischen Verlauf. Teil 2: Darstellung pathomorphologischer Befunde. 20. Arbeitstagung der Zootierärzte im deutschsprachigen Raum vom 3. bis 5. November 2000 in Salzburg. Tagungsbericht S. 114-129.

Ott Joslin J, Collins D. 1999. Chapter 5: Designing an Ideal Animal Shipment. In: Zoo and Wild Animal Medicine, Current Therapy 4. M. Fowler, DVM & R. Miller, DVM editors, W.B. Saunders Co., Philadelphia and London. pp. 17-26.

Owens M, Alm V, Helten B, Shelly S, Reed-Smith J. 2009. Summary of Husbandry Guidelines for North American (aka Nearctic) River Otters in Captivity. IUCN/SSC Otter Specialist Group, Otters in Captivity Task Force. 25pgs. (lontracat@live.com); (www.otterspecialistgroup.org/Library/TaskForces/OCT).

Petrini K. 1998. Health care section. In: Asian small-clawed Otter Husbandry Manual. Asian small-clawed Otter SSP, Columbus Zoological Park.

Petrini K, Spelman L, Reed-Smith J. 2001. Health care. In: North American River Otter (*Lontra canadensis*) Husbandry Notebook, 2nd edition. John Ball Zoo, Grand Rapids, MI. pp 165-205.

Povey KD. 2002. Close encounters: the benefits of using education program animals. Annual Proceedings of the Association of Zoos and Aquariums, 117-121.

Povey KD, Rios J. 2002. Using interpretive animals to deliver effective messages in zoos. Journal of Interpretation Research 7: 19-28.

Procter J. 1963. A contribution to the natural history of the spotted-necked otter (*Lutra maculicollis*) in Tanganyika. East African Widlife Journal 1: 93-102.

Read BW, Meier JE. 1996. Neonatal care protocols. In: Wild Mammals in Captivity, Principles and Techniques. Kleiman DG, Allen ME, Thompson KV, Lumpkin S (Eds.). University of Chicago Press, Chicago, IL., 41-55.

Reed-Smith J. 1994. Diet. In: The North American River Otter Husbandry Notebook. John Ball Zoological Garden. Grand Rapids, MI.

Reed-Smith J. 2001. The North American River Otter Husbandry Notebook, 2nd Edition. John Ball Zoological Garden, Grand Rapids, MI.

Reed-Smith J, Polechla P. 2002. Husbandry and captive breeding of otters (Lutrinae) in North and Latin America: a synopsis of current progress. In: Proceedings of the VIIth International Otter Colloquium, March 14 – 19, 1998. Trebon, Czech Republic. Dulfer R. et al. (Eds.). IUCN/SSC Otter Specialist Group, Aktion Fischotterschutz, Hankensbüttel, Germany, 276-281.

Reed-Smith J. 2004a. AZA Otter SSP Otter Keeper Workshop, Frostburg, Maryland.

Reed-Smith J. 2004b. Reproduction in North American River Otter – behavioral indicators. Unpublished research.

Reuther C. 1991. Otters in Captivity – A Review with Special Reference to *Lutra lutra*. Proc. V. Inter. Otter Colloquium, Hankensbuttel, 1989. Reuther C, Röchert R (Eds.). Habitat (Aktion Fischotterschutz): 6 269-307. WR 251.

Rock KR, Rock ES, Bowyer RT, Faro JB. 1994. Degree of association and use of a helper by coastal river otters, *Lutra canadensis*, in Prince William Sound, Alaska. Canadian Field Naturalist 108(3): 367-369.

Rosas FCW, Zuanon JAS, Carter SK. 1999. Feeding ecology of the giant otter, *Pteronura brasiliensis.* Biotropica 31(3): 502-506.
Rostain RR. 2000. Social behavior and olfactory communication in the North American river otter, *Lontra canadensis*. MS thesis. San Francisco, California, San Francisco State Univ.
Rostain R, Ben-David M, Groves P, Randall JA.. 2004. Why do river otters scent-mark? An experimental test of several hypotheses. Animal Behaviour 68(4): 703-711.
Rowe-Rowe DT. 1978. Biology of two otter species in South Africa. In: Duplaix N (Ed.), Otters: Proceedings of the First Meeting of the Otter Specialists Group, IUCN, Morges, 130-139.
Rowe-Rowe DT. 1991. Status of otters in Africa. Proceedings V International Otter Colloquium, Hankensbüttel, Germany, 1989. Reuther C, Rochert R (Eds.), Aktion Fischotterschutz, Habitat 6:15-20
Schenck C, Staib E. 1994. Giant otter – a giant under even bigger pressure. München.
Schollhamer M. 1987. Captive management of African spot-necked otters, *Hydrictus maculicollis*, at Brookfield Zoo. Unpublished account.
Serfass TL, Whary MT, Peper RL, Brooks RP, Swimley TJ, Lawrence WR, Rupprecht CE. 1995. Rabies in a river otter intended (*Lutra canadensis*) for reintroduction. Journal of Zoo and Wildlife Medicine 26:311-314.
Serfass TL, Brooks RP, Novak JM, Johns PE, Rhodes Jr. OE. (1998). Genetic variation among populations of river otter in North America: considerations for reintroduction projects. Journal of Mammalogy 79:736-746.
Sherwood KP, Rallis SF, Stone J. 1989. Effects of live animals vs. preserved specimens on student learning. Zoo Biology 8: 99-104.
Silva JL. 1993. Compostion, lipid changes, and sensory evaluation of two sizes of channel catfish during frozen storage. Journal of Applied Aquaculture 2(2): 39-50.
Snyder DE, Hamir AN, Nettles VG, Rupprecht CE. 1989. *Dirofilaria immitis* in a river otter (*Lutra canadensis*) from Louisiana. Journal of Wildlife Diseases 25: 629.
Spelman LH. 1998. North American river otter (*Lutra canadensis*) translocation in North Carolina 1989-1996. In: Proceedings of the 2nd Sci. Mtg. of the European Association of Zoo & Wildlife Veterinarians (EAZWV), May 21 – 24, 1998. Chester, United Kingdom. Zwart P et al (Eds.), 461-465.
Spelman LH. 1999. Otter anesthesia. In: Fowler ME, Miller ER (Eds.). Zoo and Wild Animal Medicine, 4th edition. W.B. Saunders Co., Philedelphia, Pennsylvania, 436-443
Staib E. 2002. Öko-Ethologie von Riesenottern (*Pteronura brasiliensis*) in Peru. Aachen: Shaker 2002. (Translation provided in Sykes-Gatz 2005).
Stevens SS, Serfass TL. 2008. Visitation patterns of nearctic river otters (*Lontra canadensis*) at latrines. Northeastern Naturalist 15(1):1-12.
Sykes-Gatz S. 2005. International Giant Otter Studbook Husbandry and Management Information and Guidelines 2005, Husbandry and Management of the Giant Otter (*Pteronura brasiliensis*), 2nd Edition). Zoo Dortmund, Germany, E-mail: sheilasykes@hotmail.com or sheilasykes@hotmail.com.
Sykes-Gatz S. 2005. International Giant Otter Studbook Husbandry and Management Information and Guidelines (2005). WAZA website: www.waza.org/virtualzoo/factsheet.php?id=112-004-025-001&view=Small%20Carnivores.
Sykes-Gatz S. 1999-2006. Giant otter (*Pteronura brasiliensis*) preproduction, parental and older sibling care of litters & cub development at Zoo Dortmund. Zoo Dortmund, Dortmund, Germany. Unpublished reports.
Sykes-Gatz, S. & Gatz, V. 2007. Giant Otter EEP Annual Report 2007, EAZA Yearbook 2007 (in publication), EAZA Executive Office (European Association of Zoos and Aquariums), Amsterdam.
Tarasoff FJ. 1974. Anatomical adaptations in the river otter, sea otter and Harp seal in reference to thermal regulation. In: Harrison RJ. (Ed.) Functional Anatomy of Marine Mammals, Vol. 2, Academic Press, New York, New York, 111-142.
Testa JW, Holleman DF, Bowyer RT, Faro JB. 1994. Estimating populations of marine river otters in Prince William Sound, Alaska, using radiotracer implants. Journal of Mammalogy 75: 1021-1032.
Toddes B. 2005 & 2006. Analysis of fish fed to giant otter at the Philadelphia Zoo. Unpublished Philadelphia Zoo document, Philadelphia, PA.
Toweill DE, Tabor JE. 1982. River otter: *Lutra canadensis*. In: Wild Mammals of North America: Biology, Management and Economics. Chapman, JA, Feldhamer GA (Eds.). Johns Hopkins University Press.

Trebbau P. 1978. Some observations on the mating behavior of the Brazilian giant otter (*Pteronura brasiliensis*). Der Zoologische Garten 48: 187-188.

Tumanov IL, Sorina EA. 1997. Dynamics of nutritional energetics of female mustelids. In: Small Carnivore Conservation - The Newsletter and Journal of the IUCN/SSC Mustelid, Viverrid, and Procyonid Specialist Group. No. 17 (Oct), 10-14. Edegem, Belgium.

Twibell RG, Brown PB. 2000. Dietary choline requirement of juvenile yellow perch (*Perca flavescens*). Journal of Nutrition 130: 95-99.

Vesell ES. 1967. Induction of drug-metabolizing enzymes in liver microsomes of mice and rats by softwood bedding. Science 157: 1057-1058.

Wallach J, Boever WJ. 1983. Diseases of Exotic Animals: Medical and Surgical Management. W.B. Saunders Co., Philadelphia.

Webb T. 2008. Hand-rearing 1.1 Asian small-clawed otter (*Amblonyx cinereus*). Animal Keepers Forum, January 2008, 17-20.

Weisel JW, Nagaswami C, Peterson RO. 2005. River otter hair structure facilitates interlocking to impede penetration of water and allow trapping of air. Canadian Journal of Zoology, May 2005: 83:649-655.

Wilson DE, Reeder DM (Eds.). 2005. Mammal Species of the World: A Taxonomic and Geographic Reference, 3rd ed. Johns Hopkins Univ. Press. 2,142pp. (www.bucknell.edu/MSW3/)

Wilson C, Tropea K, Calle P. Asian small-clawed otter husbandry protocol. New York Zoological Park. Unpublished.

Wolf RL, Tymitz BL. 1981. Studying visitor perceptions of zoo environments: a naturalistic view. In: Olney PJS (Ed.), International Zoo Yearbook. Dorchester: The Zoological Society of London, 49-53.

Wooster D. 1998. Role of keeper in management of small cats. In: Husbandry Manual for Small Felids. Mellen J, Wildt D (Eds). AZA & Disney's Animal Kingdom.

Wright PJ, Verstegen JP, Onclin K, Jochle W, Armour AF, Martin GB, Trigg TE. 2001. Suppression of the oestrous responses of bitches to the GnRH analogue deslorelin by progestin. Journal of Reproduction and Fertility, Supplement 57: 263-268.

Wünnemann K. 1990. Veterinärmedzinische Aspekte bei der Aufzucht junger Riesenotter in Carl Hagenbecks Tierpark. 10. Arbeitstagung der Zootierärzte im deutschsprachigen Raum vom 2-4 November 1990 in Basel, Tagungsbericht S. 70-73.

Wünnemann K. 1995a. Giant otter husbandry. In: Husbandry Handbook for Mustelids. Partridge, J (Ed.). Association of British Wild Animal Keepers. Bristol, UK., 181-184.

Wünnemann K. 1995b. Breeding giant otters (*P. brasiliensis*) in Germany. In: Habitat Arbeitsberichte der Aktion Fischotterschutz e.V.. Proceedings VI. International Otter Colloquium Pietermaritzburg 1993. Reuther C, Rowe-Rowe D (Eds.). GN-Gruppe Naturschutz GmbH: Hankensbuttel.

Yerke R, Burns A. 1991. Measuring the impact of animal shows on visitor attitudes. Annual Proceedings of the American Association of Zoological Parks and Aquariums, 532-534.

Yerke R, Burns A. 1993. Evaluation of the educational effectiveness of an animal show outreach program for schools. Annual Proceedings of the American Association of Zoological Parks and Aquariums, 366-368.

Zeller F. 1960. Notes on the giant otter (*Pteronura brasiliensis*) at Cologne Zoo. International Zoo Yearbook. 2: 81.

Personal communications:

Ben-David, Merav: Univ. of Wyoming. IUCN/SSC Otter Specialist Group, bendavid@uwyo.edu.

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Yoxon, Grace: International Otter Survival Fund. Scotland. Iosf2@aol.com.

Appendix A: Accreditation Standards by Chapter

The following specific standards of care relevant to Lutrinae are taken from the AZA Accreditation Standards and Related Policies (AZA 2009) and are referenced fully within the chapters of this animal care manual:

Chapter 1

(1.5.7) The animal collection must be protected from weather detrimental to their health.

(10.2.1) Critical life-support systems for the animal collection, including but not limited to plumbing, heating, cooling, aeration, and filtration, must be equipped with a warning mechanism, and emergency backup systems must be available. All mechanical equipment should be under a preventative maintenance program as evidenced through a record-keeping system. Special equipment should be maintained under a maintenance agreement, or a training record should show that staff members are trained for specified maintenance of special equipment.

(1.5.9) The institution must have a regular program of monitoring water quality for collections of fish, pinnipeds, cetaceans, and other aquatic animals. A written record must be maintained to document long-term water quality results and chemical additions.

Chapter 2

(1.5.2) Animals should be displayed, whenever possible, in exhibits replicating their wild habitat and in numbers sufficient to meet their social and behavioral needs. Display of single specimens should be avoided unless biologically correct for the species involved.

(10.3.3) All animal enclosures (exhibits, holding areas, hospital, and quarantine/isolation) must be of a size and complexity sufficient to provide for the animal's physical, social, and psychological well-being; and exhibit enclosures must include provisions for the behavioral enrichment of the animals.

(11.3.1) All animal exhibits and holding areas must be secured to prevent unintentional animal egress.

(11.3.6) Guardrails/barriers must be constructed in all areas where the visiting public could have contact with other than handleable animals.

(11.2.3) All emergency procedures must be written and provided to staff and, where appropriate, to volunteers. Appropriate emergency procedures must be readily available for reference in the event of an actual emergency. These procedures should deal with four basic types of emergencies: fire, weather/environment; injury to staff or a visitor; animal escape.

(11.6.2) Security personnel, whether staff of the institution, or a provided and/or contracted service, must be trained to handle all emergencies in full accordance with the policies and procedures of the institution. In some cases, it is recognized that Security personnel may be in charge of the respective emergency (i.e., shooting teams).

(11.2.4) The institution must have a communication system that can be quickly accessed in case of an emergency.

(11.2.5) A written protocol should be developed involving local police or other emergency agencies and include response times to emergencies.

(11.5.3) Institutions maintaining potentially dangerous animals (sharks, whales, tigers, bears, etc.) must have appropriate safety procedures in place to prevent attacks and injuries by these animals. Appropriate response procedures must also be in place to deal with an attack resulting in an injury. These procedures must be practiced routinely per the emergency drill requirements contained in these standards. Whenever injuries result from these incidents, a written account outlining the cause of the incident, how the injury was handled, and a description of any resulting changes to either the safety procedures or the physical facility must be prepared and maintained for five years from the date of the incident.

Chapter 3

(1.5.11) Animal transportation must be conducted in a manner that is safe, well-planned and coordinated, and minimizes risk to the animal(s), employees, and general public. All applicable local, state, and federal laws must be adhered to.

Chapter 5

(2.6.2) A formal nutrition program is recommended to meet the behavioral and nutritional needs of all species and specimens within the collection.

(2.6.3) Animal diets must be of a quality and quantity suitable for each animal's nutritional and psychological needs. Diet formulations and records of analysis of appropriate feed items should be maintained and may be examined by the Visiting Committee. Animal food, especially seafood products, should be purchased from reliable sources that are sustainable and/or well managed.
(2.6.1) Animal food preparations must meet all local, state/provincial, and federal regulations.
(2.6.4) The institution should assign at least one person to oversee appropriate browse material for the collection.

Chapter 6

(2.1.1) A full-time staff veterinarian is recommended. However, the Commission realizes that in some cases such is not practical. In those cases, a consulting/part-time veterinarian must be under contract to make at least twice monthly inspections of the animal collection and respond as soon as possible to any emergencies. The Commission also recognizes that certain collections, because of their size and/or nature, may require different considerations in veterinary care.
(2.1.2) So that indications of disease, injury, or stress may be dealt with promptly, veterinary coverage must be available to the animal collection 24 hours a day, 7 days a week.
(2.2.1) Written, formal procedures must be available to the animal care staff for the use of animal drugs for veterinary purposes and appropriate security of the drugs must be provided.
(1.4.6) A staff member must be designated as being responsible for the institution's animal record-keeping system. That person must be charged with establishing and maintaining the institution's animal records, as well as with keeping all animal care staff members apprised of relevant laws and regulations regarding the institution's animal collection.
(1.4.7) Animal records must be kept current, and data must be logged daily.
(1.4.5) At least one set of the institution's historical animal records must be stored and protected. Those records should include permits, titles, declaration forms, and other pertinent information.
(1.4.4) Animal records, whether in electronic or paper form, including health records, must be duplicated and stored in a separate location.
(1.4.3) Animals must be identifiable, whenever practical, and have corresponding ID numbers. For animals maintained in colonies or other animals not considered readily identifiable, the institution must provide a statement explaining how record keeping is maintained.
(1.4.1) An animal inventory must be compiled at least once a year and include data regarding acquisitions and dispositions in the animal collection.
(1.4.2) All species owned by the institution must be listed on the inventory, including those animals on loan to and from the institution. In both cases, notations should be made on the inventory.
(2.7.1) The institution must have holding facilities or procedures for the quarantine of newly arrived animals and isolation facilities or procedures for the treatment of sick/injured animals.
(2.7.3) Quarantine, hospital, and isolation areas should be in compliance with standards or guidelines adopted by the AZA.
(2.7.2) Written, formal procedures for quarantine must be available and familiar to all staff working with quarantined animals.
(11.1.2) Training and procedures must be in place regarding zoonotic diseases.
(11.1.3) A tuberculin testing and surveillance program must be established for appropriate staff in order to ensure the health of both the employees and the animal collection.
(2.5.1) Deceased animals should be necropsied to determine the cause of death. Disposal after necropsy must be done in accordance with local/federal laws.
(2.4.1) The veterinary care program must emphasize disease prevention.
(1.5.5) For animals used in offsite programs and for educational purposes, the institution must have adequate protocols in place to protect the rest of the collection from exposure to infectious agents.
(2.3.1) Capture equipment must be in good working order and available to authorized, trained personnel at all times.
(2.4.2) Keepers should be trained to recognize abnormal behavior and clinical symptoms of illness and have knowledge of the diets, husbandry (including enrichment items and strategies), and restraint procedures required for the animals under their care. However, keepers should not evaluate illnesses nor prescribe treatment.
(2.3.2) Hospital facilities should have x-ray equipment or have access to x-ray services.

(1.5.8) The institution must develop a clear process for identifying and addressing animal welfare concerns within the institution.

Chapter 8

(1.6.1) The institution must have a formal written enrichment program that promotes species-appropriate behavioral opportunities.

(1.6.2) The institution must have a specific staff member(s) or committee assigned for enrichment program oversight, implementation, training, and interdepartmental coordination of enrichment efforts.

Chapter 9

(5.3) A written policy on the use of live animals in programs should be on file. Animals in education programs must be maintained and cared for by trained staff, and housing conditions must meet standards set for the remainder of the animal collection, including species-appropriate shelter, exercise, social and environmental enrichment, access to veterinary care, nutrition, etc. Since some of these requirements can be met outside of the primary enclosure, for example, enclosures may be reduced in size provided that the animal's physical and psychological needs are being met.

(1.5.3) If animal demonstrations are a part of the institution's programs, an education and conservation message must be an integral component.

Chapter 10

(5.3) Institutions should maximize the generation of scientific knowledge gained from the animal collection. This might be achieved by participating in AZA TAG/SSP sponsored research when applicable, conducting original research projects, affiliating with local universities, and/or employing staff with scientific credentials.

(5.2) Institutions must have a written policy that outlines the type of research that it conducts, methods, staff involvement, evaluations, animals to be involved, and guidelines for publication of findings.

(5.1) Research activities must be under the direction of a person qualified to make informed decisions regarding research.

Appendix B: Acquisition/Disposition Policy

I. Introduction: The Association of Zoos and Aquariums (AZA) was established, among other reasons, to foster continued improvement in the zoological park and aquarium profession. One of its most important roles is to provide a forum for debate and consensus building among its members, the intent of which is to attain high ethical standards, especially those related to animal care and professional conduct. The stringent requirements for AZA accreditation and high standards of professional conduct are unmatched by similar organizations and also far surpass the United States Department of Agriculture's Animal and Plant Health Inspection Service's requirements for licensed animal exhibitors. AZA member facilities must abide by a Code of Professional Ethics - a set of standards that guide all aspects of animal management and welfare. As a matter of priority, AZA institutions should acquire animals from other AZA institutions and dispose of animals to other AZA institutions.

AZA accredited zoological parks and aquariums cannot fulfill their important missions of conservation, education and science without living animals. Responsible management of living animal populations necessitates that some individuals be acquired and that others be removed from the collection at certain times. Acquisition of animals can occur through propagation, trade, donation, loan, purchase, capture, or rescue. Animals used as animal feed are not accessioned into the collection.

Disposition occurs when an animal leaves the collection for any reason. Reasons for disposition vary widely, but include cooperative population management (genetic or demographic management), reintroduction, behavioral incompatibility, sexual maturation, animal health concerns, loan or transfer, or death.

The AZA Acquisition/Disposition Policy (A/D) was created to help (1) guide and support member institutions in their animal acquisition and disposition decisions, and (2) ensure that all additions and removals are compatible with the Association's stated commitment to "save and protect the wonders of the living natural world." More specifically, the AZA A/D Policy is intended to:

- Ensure that the welfare of individual animals and conservation of populations, species and ecosystems are carefully considered during acquisition and disposition activities;
- Maintain a proper standard of conduct for AZA members during acquisition and disposition activities;
- Ensure that animals from AZA member institutions are not transferred to individuals or organizations that lack the appropriate expertise or facilities to care for them.
- Support the goal of AZA’s cooperatively managed populations and associated programs, including Species Survival Plans® (SSPs), Population Management Plans (PMPs), and Taxon Advisory Groups (TAGs).

The AZA Acquisition/Disposition Policy will serve as the default policy for AZA member institutions. Institutions may develop their own A/D Policy in order to address specific local concerns. Any institutional policy must incorporate and not conflict with the AZA acquisition and disposition standards.

Violations of the AZA Acquisition/Disposition Policy will be dealt with in accordance with the AZA Code of Professional Ethics. Violations can result in an institution's or individual's expulsion from membership in the AZA.

II. Group or Colony-based Identification: For some colonial, group-living, or prolific species, such as certain insects, aquatic invertebrates, schooling fish, rodents, and bats, it is often impossible or highly impractical to identify individual specimens. These species are therefore maintained, acquisitioned, and disposed of as a group or colony. Therefore, when this A/D Policy refers to animals or specimens, it is in reference to both individuals and groups/colonies.

III. Germplasm: Acquisition and disposition of germplasm should follow the same guidelines outlined in this document if its intended use is to create live animal(s). Ownership of germplasm and any resulting animals should be clearly defined. Institutions acquiring or dispositioning germplasm or any animal parts or samples should consider not only its current use, but also future possible uses as new technologies become available.

IV(a). General Acquisitions: Animals are to be acquisitioned into an AZA member institution’s collection if the following conditions are met:

1. Acquisitions must meet the requirements of all applicable local, state, federal and international regulations and laws.
2. The Director or Chief Executive Officer of the institution is charged with the final authority and responsibility for the monitoring and implementation of all acquisitions.
3. Acquisitions must be consistent with the mission of the institution, as reflected in its Institutional Collection Plan, by addressing its exhibition/education, conservation, and/or scientific goals.
4. Animals that are acquired for the collection, permanently or temporarily, must be listed on institutional records. All records should follow the Standards for Data Entry and Maintenance of North American Zoo and Aquarium Animal Records Databases®.
5. Animals may be acquired temporarily for reasons such as, holding for governmental agencies, rescue and/or rehabilitation, or special exhibits. Animals should only be accepted if they will not jeopardize the health, care or maintenance of the animals in the permanent collection or the animal being acquired.
6. The institution must have the necessary resources to support and provide for the professional care and management of a species, so that the physical and social needs of both specimen and species are met.
7. Attempts by members to circumvent AZA conservation programs in the acquisition of SSP animals are detrimental to the Association and its conservation programs. Such action may be detrimental to the species involved and is a violation of the Association's Code of Professional Ethics. All AZA members must work through the SSP program in efforts to acquire SSP species and adhere to the AZA Full Participation policy.
8. Animals are only to be acquired from sources that are known to operate legally and conduct their business in a manner that reflects and/or supports the spirit and intent of the AZA Code of Professional Ethics as well as this policy. Any convictions of state, federal, or international wildlife laws should be reviewed, as well as any previous dealings with other AZA accredited institutions.
9. When acquiring specimens managed by a PMP, institutions should consult with the PMP manager.
10. Institutions should consult AZA Wildlife Conservation and Management Committee (WCMC)-approved Regional Collection Plans (RCPs) when making acquisition decisions.

IV(b). Acquisitions from the Wild: The maintenance of wild animal populations for education and wildlife conservation purposes is a unique responsibility of AZA member zoos and aquariums. To accomplish these goals, it may be necessary to acquire wild-caught specimens. Before acquiring animals from the wild, institutions are encouraged to examine sources including other AZA institutions or regional zoological associations.

When acquiring animals from the wild, careful consideration must be taken to evaluate the long-term impacts on the wild population. Any capture of free-ranging animals should be done in accordance with all local, state, federal, and international wildlife laws and regulations and not be detrimental to the long-term viability of the species or the wild or *ex-situ* population(s). In crisis situations, when the survival of a population is at risk, rescue decisions are to be made on a case-by-case basis.

V(a). Disposition Requirements – Living Animals: Successful conservation and animal management efforts rely on the cooperation of many entities, both within and outside of AZA. While preference is given to placing animals within AZA member institutions, it is important to foster a cooperative culture among those who share the primary mission of AZA accredited facilities. The AZA draws a strong distinction between the mission, stated or otherwise, of non-AZA member organizations and the mission of professionally managed zoological parks and aquariums accredited by the AZA.

An accredited AZA member balances public display, recreation, and entertainment with demonstrated efforts in education, conservation, and science. While some non-AZA member organizations may meet minimum daily standards of animal care for wildlife, the AZA recognizes that this, by itself, is insufficient to warrant either AZA membership or participation in AZA's cooperative animal management programs. When an animal is sent to a non-member of AZA, it is imperative that the member be confident that the animal will be cared for properly.

Animals may only be disposed of from an AZA member institution's collection if the following conditions are met:

1. Dispositions must meet the requirements of all applicable local, state, federal and international regulations and laws.
2. The Director or Chief Executive Officer of the institution is charged with the final authority and responsibility for the monitoring and implementation of all dispositions.
3. Any disposition must abide by the Mandatory Standards and General Advisories of the AZA Code of Professional Ethics. Specifically, "a member shall make every effort to assure that all animals in his/her collection and under his/her care are disposed of in a manner which meets the current disposition standards of the Association and do not find their way into the hands of those not qualified to care for them properly."
4. Non-domesticated animals shall not be disposed of at animal auctions. Additionally, animals shall not be disposed of to any organization or individual that may use or sell the animal at an animal auction. In transactions with AZA non-members, the recipient must ensure in writing that neither the animal nor its offspring will be disposed of at a wild animal auction or to an individual or organization that allows the hunting of the animal.
5. Animals shall not be disposed of to organizations or individuals that allow the hunting of these animals or their offspring. This does not apply to individuals or organizations which allow the hunting of only free-ranging game species (indigenous to North America) and established long-introduced species such as, but not limited to, white-tailed deer, quail, rabbit, waterfowl, boar, ring-necked pheasant, chukar, partridge, and trout. AZA distinguishes hunting/fishing for sport from culling for sustainable population management and wildlife conservation purposes.
6. Attempts by members to circumvent AZA conservation programs in the disposition of SSP animals are detrimental to the Association and its conservation programs. Such action may be detrimental to the species involved and is a violation of the Association's Code of Professional Ethics. All AZA members must work through the SSP program in efforts to deacquisition SSP species and adhere to the AZA Full Participation policy.
7. Domesticated animals are to be disposed of in a manner consistent with acceptable farm practices and subject to all relevant laws and regulations.
8. Live specimens may be released within native ranges, subject to all relevant laws and regulations. Releases may be a part of a recovery program and any release must be compatible with the AZA Guidelines for Reintroduction of Animals Born or Held in Captivity, dated June 3, 1992.
9. Detailed disposition records of all living or dead specimens must be maintained. Where applicable, proper animal identification techniques should be utilized.
10. It is the obligation of every loaning institution to monitor, at least annually, the conditions of any loaned specimens and the ability of the recipient to provide proper care. If the conditions and care of animals are in violation of the loan agreement, it is the obligation of the loaning institution to recall the animal. Furthermore, an institution's loaning policy must not be in conflict with this A/D Policy.
11. If live specimens are euthanized, it must be done in accordance with the established policy of the institution and the Report of the American Veterinary Medical Association Panel on Euthanasia (Journal of the American Veterinary Medical Association 218 (5): 669-696, 2001).
12. In dispositions to non-AZA members, the non-AZA member's mission (stated or implied) must not be in conflict with the mission of AZA, or with this A/D Policy.
13. In dispositions to non-AZA member facilities that are open to the public, the non-AZA member must balance public display, recreation, and entertainment with demonstrated efforts in conservation, education, and science.
14. In dispositions to non-AZA members, the AZA members must be convinced that the recipient has the expertise, records management practices, financial stability, facilities, and resources required to properly care for and maintain the animals and their offspring. It is recommended that this documentation be kept in the permanent record of the animals at the AZA member institution.
15. If living animals are sent to a non-AZA member research institution, the institution must be registered under the Animal Welfare Act by the U.S. Department of Agriculture Animal and Plant Health Inspection Service. For international transactions, the receiving facility should be registered by that country's equivalent body with enforcement over animal welfare.
16. No animal disposition should occur if it would create a health or safety risk (to the animal or humans) or have a negative impact on the conservation of the species.

17. Inherently dangerous wild animals or invasive species should not be dispositioned to the pet trade or those unqualified to care for them.
18. Under no circumstances should any primates be dispositioned to a private individual or to the pet trade.
19. Fish and aquatic invertebrate species that meet ANY of the following are inappropriate to be disposed of to private individuals or the pet trade:
    a. species that grow too large to be housed in a 72-inch long, 180 gallon aquarium (the largest tank commonly sold in retail stores)
    b. species that require extraordinary life support equipment to maintain an appropriate *ex-situ* environment (e.g., cold water fish and invertebrates)
    c. species deemed invasive (e.g., snakeheads)
    d. species capable of inflicting a serious bite or venomous sting (e.g., piranha, lion fish, blue-ringed octopus)
    e. species of wildlife conservation concern
20. When dispositioning specimens managed by a PMP, institutions should consult with the PMP manager.
21. Institutions should consult WCMC-approved RCPs when making disposition decisions.

V(b). Disposition Requirements – Dead Specimens: Dead specimens (including animal parts and samples) are only to be disposed of from an AZA member institution's collection if the following conditions are met:

1. Dispositions of dead specimens must meet the requirements of all applicable local, state, federal and international regulations and laws.
2. Maximum utilization is to be made of the remains, which could include use in educational programs or exhibits.
3. Consideration is given to scientific projects that provide data for species management and/or conservation.
4. Records (including ownership information) are to be kept on all dispositions, including animal body parts, when possible.
5. SSP and TAG necropsy protocols are to be accommodated insofar as possible.

VI. Transaction Forms: AZA member institutions will develop transaction forms to record animal acquisitions and dispositions. These forms will require the potential recipient or provider to adhere to the AZA Code of Professional Ethics, the AZA Acquisition/Disposition Policy, and all relevant AZA and member policies, procedures and guidelines. In addition, transaction forms must insist on compliance with the applicable laws and regulations of local, state, federal and international authorities.

Appendix C: Recommended Quarantine Procedures

Quarantine Facility: A separate quarantine facility, with the ability to accommodate mammals, birds, reptiles, amphibians, and fish should exist. If a specific quarantine facility is not present, then newly acquired animals should be isolated from the established collection in such a manner as to prohibit physical contact, to prevent disease transmission, and to avoid aerosol and drainage contamination.

Such separation should be obligatory for primates, small mammals, birds, and reptiles, and attempted wherever possible with larger mammals such as large ungulates and carnivores, marine mammals, and cetaceans. If the receiving institution lacks appropriate facilities for isolation of large primates, pre-shipment quarantine at an AZA or AALAS accredited institution may be applied to the receiving institutions protocol. In such a case, shipment must take place in isolation from other primates. More stringent local, state, or federal regulations take precedence over these recommendations.

Quarantine Length: Quarantine for all species should be under the supervision of a veterinarian and consist of a minimum of 30 days (unless otherwise directed by the staff veterinarian). Mammals: If during the 30-day quarantine period, additional mammals of the same order are introduced into a designated quarantine area, the 30-day period must begin over again. However, the addition of mammals of a different order to those already in quarantine will not have an adverse impact on the originally quarantined mammals. Birds, Reptiles, Amphibians, or Fish: The 30-day quarantine period must be closed for each of the above Classes. Therefore, the addition of any new birds into a bird quarantine area requires that the 30-day quarantine period begin again on the date of the addition of the new birds. The same applies for reptiles, amphibians, or fish.

Quarantine Personnel: A keeper should be designated to care only for quarantined animals or a keeper should attend quarantined animals only after fulfilling responsibilities for resident species. Equipment used to feed and clean animals in quarantine should be used only with these animals. If this is not possible, then equipment must be cleaned with an appropriate disinfectant (as designated by the veterinarian supervising quarantine) before use with post-quarantine animals.

Institutions must take precautions to minimize the risk of exposure of animal care personnel to zoonotic diseases that may be present in newly acquired animals. These precautions should include the use of disinfectant foot baths, wearing of appropriate protective clothing and masks in some cases, and minimizing physical exposure in some species; e.g., primates, by the use of chemical rather than physical restraint. A tuberculin testing/surveillance program must be established for zoo/aquarium employees in order to ensure the health of both the employees and the animal collection.

Quarantine Protocol: During this period, certain prophylactic measures should be instituted. Individual fecal samples or representative samples from large numbers of individuals housed in a limited area (e.g., birds of the same species in an aviary or frogs in a terrarium) should be collected at least twice and examined for gastrointestinal parasites. Treatment should be prescribed by the attending veterinarian. Ideally, release from quarantine should be dependent on obtaining two negative fecal results spaced a minimum of two weeks apart either initially or after parasiticide treatment. In addition, all animals should be evaluated for ectoparasites and treated accordingly.

Vaccinations should be updated as appropriate for each species. If the animal arrives without a vaccination history, it should be treated as an immunologically naive animal and given an appropriate series of vaccinations. Whenever possible, blood should be collected and sera banked. Either a -94°F (-70ºC) frost-free freezer or a -4°F (-20ºC) freezer that is not frost-free should be available to save sera. Such sera could provide an important resource for retrospective disease evaluation.

The quarantine period also represents an opportunity to, where possible, permanently identify all unmarked animals when anesthetized or restrained (e.g., tattoo, ear notch, ear tag, etc.). Also, whenever animals are restrained or immobilized, a complete physical, including a dental examination, should be performed. Complete medical records should be maintained and available for all animals during the quarantine period. Animals that die during quarantine should have a necropsy performed under the supervision of a veterinarian and representative tissues submitted for histopathologic examination.

Quarantine Procedures: The following are recommendations and suggestions for appropriate quarantine procedures for Lutrinae:

Required:
1. Direct and flotation fecals
2. Vaccinate as appropriate

Strongly Recommended:
1. CBC/sera profile
2. Urinalysis
3. Appropriate serology (FIP, FeLV, FIV)
4. Heartworm testing in appropriate species

Appendix D: Program Animal Position Statement

The Conservation Education Committee (CEC) of the Association of Zoos and Aquariums supports the appropriate use of program animals as an important and powerful educational tool that provides a variety of benefits to zoo and aquarium educators seeking to convey cognitive and affective (emotional) messages about conservation and wildlife. Utilizing these animals allows educators to strongly engage audiences. As discussed below, the use of program animals has been demonstrated to result in lengthened learning periods, increased knowledge acquisition and retention, enhanced environmental attitudes, and the creation of positive perceptions concerning zoo and aquarium animals.

Audience Engagement: Zoos and aquariums are ideal venues for developing emotional ties to wildlife and fostering an appreciation for the natural world. However, developing and delivering effective educational messages in the free-choice learning environments of zoos and aquariums is a difficult task. Zoo and aquarium educators are constantly challenged to develop methods for engaging and teaching visitors who often view a trip to the zoo as a social or recreational experience (Morgan & Hodgkinson 1999). The use of program animals can provide the compelling experience necessary to attract and maintain personal connections with visitors of all motivations, thus preparing them for learning and reflection on their own relationships with nature.

Program animals are powerful catalysts for learning for a variety of reasons. They are generally active, easily viewed, and usually presented in close proximity to the public. These factors have proven to contribute to increasing the length of time that people spend watching animals in zoo exhibits (Wolf & Tymitz 1981; Bitgood, Patterson & Benefield, 1986, 1988). In addition, the provocative nature of a handled animal likely plays an important role in captivating a visitor. In two studies (Povey & Rios 2002; Povey 2002), visitors viewed animals three and four times longer while they were being presented in demonstrations outside of their enclosure with an educator than while they were on exhibit. Clearly, the use of program animals in shows or informal presentations is effective in lengthening the potential time period for learning and overall impact.

Program animals also provide the opportunity to personalize the learning experience, tailoring the teaching session to what interests the visitors. Traditional graphics offer little opportunity for this level of personalization of information delivery and are frequently not read by visitors (Churchman 1985; Johnston 1998). For example, Povey (2002) found that only 25% of visitors to an animal exhibit read the accompanying graphic; whereas, 45% of visitors watching the same animal handled in an educational presentation asked at least one question and some asked as many as seven questions. Having an animal accompany the educator allowed the visitors to make specific inquiries about topics in which they were interested.

Knowledge Acquisition: Improving our visitors' knowledge and understanding regarding wildlife and wildlife conservation is a fundamental goal for many zoo educators using program animals. A growing body of evidence supports the validity of using program animals to enhance delivery of these cognitive messages as well.

- MacMillen (1994) found that the use of live animals in a zoomobile outreach program significantly enhanced cognitive learning in a vertebrate classification unit for sixth grade students.
- Sherwood et al. (1989) compared the use of live horseshoe crabs and sea stars to the use of dried specimens in an aquarium education program and demonstrated that students made the greatest cognitive gains when exposed to programs utilizing the live animals.
- Povey and Rios (2002) noted that in response to an open-ended survey question ("Before I saw this animal, I never realized that…"), visitors watching a presentation utilizing a program animal provided 69% cognitive responses (i.e., something they learned) versus 9% made by visitors viewing the same animal in its exhibit (who primarily responded with observations).
- Povey (2002) recorded a marked difference in learning between visitors observing animals on exhibit versus being handled during informal presentations. Visitors to demonstrations utilizing a raven and radiated tortoises were able to answer questions correctly at a rate as much as eleven times higher than visitors to the exhibits.

Enhanced Environmental Attitudes: Program animals have been clearly demonstrated to increase effective learning and attitudinal change.

- Studies by Yerke and Burns (1991) and Davison et al. (1993) evaluated the effect live animal shows had on visitor attitudes. Both found their shows successfully influenced attitudes about conservation and stewardship.
- Yerke and Burns (1993) also evaluated a live bird outreach program presented to Oregon fifth-graders and recorded a significant increase in students' environmental attitudes after the presentations.
- Sherwood et al. (1989) found that students who handled live invertebrates in an education program demonstrated both short and long-term attitudinal changes as compared to those who only had exposure to dried specimens.
- Povey and Rios (2002) examined the role program animals play in helping visitors develop positive feelings about the care and well-being of zoo animals.
- As observed by Wolf and Tymitz (1981), zoo visitors are deeply concerned with the welfare of zoo animals and desire evidence that they receive personalized care.

Conclusion: Creating positive impressions of aquarium and zoo animals, and wildlife in general, is crucial to the fundamental mission of zoological institutions. Although additional research will help us delve further into this area, the existing research supports the conclusion that program animals are an important tool for conveying both cognitive and affective messages regarding animals and the need to conserve wildlife and wild places.

Appendix E: Developing an Institutional Program Animal Policy

Membership in AZA requires that an institution meet the AZA Accreditation Standards collectively developed by our professional colleagues. Standards guide all aspects of an institution's operations; however, the accreditation commission has asserted that ensuring that member institutions demonstrate the highest standards of animal care is a top priority. Another fundamental AZA criterion for membership is that education be affirmed as core to an institution's mission. All accredited public institutions are expected to develop a written education plan and to regularly evaluate program effectiveness.

The inclusion of animals (native, exotic and domestic) in educational presentations, when done correctly, is a powerful tool. CEC's Program Animal Position Statement (Appendix D) describes the research underpinning the appropriate use of program animals as an important and powerful educational tool that provides a variety of benefits to zoo and aquarium educators seeking to convey cognitive and affective messages about conservation and wildlife. Ongoing research, such as AZA's Multi-Institutional Research Project (MIRP) and research conducted by individual AZA institutions will help zoo educators to determine whether the use of program animals conveys intended and conflicting messages and to modify and improve programs accordingly.

When utilizing program animals our responsibility is to meet both our high standards of animal care and our educational goals. Additionally, as animal management professionals, we must critically address both the species' conservation needs and the welfare of the individual animal. Because "wild creatures differ endlessly," in their forms, needs, behavior, limitations and abilities (Conway 1995), AZA, through its Animal Welfare Committee, has recently given the responsibility to develop taxon-specific animal welfare standards to the Taxon Advisory Groups (TAG) and Species Survival Plan® Program (SSP). Experts within each TAG or SSP, along with their education advisors, are charged with assessing all aspects of the taxons' biological and social needs and developing animal care standards that include specifications concerning their use as program animals.

However, even the most exacting standards cannot address the individual choices faced by each AZA institution. Therefore, each institution is required to develop a program animal policy that articulates and evaluates program benefits. The following recommendations are offered to assist each institution in formulating its own Institutional Program Animal Policy.

The Policy Development Process: Within each institution, key stakeholders should be included in the development of that institution's policy, including, but not limited to representatives from:

- The Education Department
- The Animal Husbandry Department
- The Veterinary and Animal Health Department
- The Conservation & Science Department
- Any animal show staff (if in a separate department)
- Departments that frequently request special program animal situations (e.g., special events, development, marketing, zoo or aquarium society, administration)
- Additionally, staff from all levels of the organization should be involved in this development (e.g., curators, keepers, education managers, interpreters, volunteer coordinators).

To develop a comprehensive Program Animal Policy, we recommend that the following components be included:

I. Philosophy: In general, the position of the AZA is that the use of animals in up close and personal settings, including animal contact, can be extremely positive and powerful, as long as:

- The use and setting is appropriate.
- Animal and human welfare is considered at all times.
- The animal is used in a respectful, safe manner and in a manner that does not misrepresent or degrade the animal.
- A meaningful conservation message is an integral component. Read the AZA Board-approved Conservation Messages.
- Suitable species and individual specimens are used.

Institutional program animal policies should include a philosophical statement addressing the above, and should relate the use of program animals to the institution's overall mission statement.

II. Appropriate Settings: The Program Animal Policy should include a listing of all settings both on and off site, where program animal use is permitted. This will clearly vary among institutions. Each institution's policy should include a comprehensive list of settings specific to that institution. Some institutions may have separate policies for each setting; others may address the various settings within the same policy. Examples of settings include:

<u>On-site programming:</u>

Informal and non-registrants:

- On-grounds programming with animals being brought out (demonstrations, lectures, parties, special events, and media)
- Children's zoos and contact yards
- Behind-the-scenes open houses
- Shows
- Touch pools

Formal (registration involved) and controlled settings:

- School group programs
- Summer Camps
- Overnights
- Birthday parties

<u>Offsite and outreach:</u>

- PR events (TV, radio)
- Fundraising events
- Field programs involving the public
- School visits
- Library visits
- Nursing Home visits (therapy)
- Hospital visits
- Senior Centers
- Civic Group events

In some cases, policies will differ from setting to setting (e.g., on-site and off-site use with media). These settings should be addressed separately, and should reflect specific animal health issues, assessment of stress in these situations, limitations, and restrictions.

III. Compliance with Regulations: All AZA institutions housing mammals are regulated by the USDA's Animal Welfare Act. Other federal regulations, such as the Marine Mammal Protection Act, may apply. Additionally, many states, and some cities, have regulations that apply to animal contact situations. Similarly, all accredited institutions are bound by the AZA Code of Professional Ethics. It is expected that the Institution Program Animal Policy address compliance with appropriate regulations and AZA Accreditation Standards.

IV. Collection Planning: All AZA accredited institutions should have a collection planning process in place. Program animals are part of an institution's overall collection and must be included in the overall collection planning process. The AZA Guide to Accreditation contains specific requirements for the institution collection plan. For more information about collection planning in general, please see the Collection Management pages in the Members Only section of the AZA website (www.aza.org). The following recommendations apply to program animals:

1. Listing of approved program animals (to be periodically amended as collection changes). Justification of each species should be based upon criteria such as:
    a. Temperament and suitability for program use
    b. Husbandry requirements
    c. Husbandry expertise
    d. Veterinary issues and concerns
    e. Ease and means of acquisition / disposition
    f. Educational value and intended conservation message
    g. Conservation Status
    h. Compliance with TAG and SSP guidelines and policies

2. General guidelines as to how each species (and, where necessary, for each individual) will be presented to the public, and in what settings
3. The collection planning section should reference the institution's acquisition and disposition policies.

V. Conservation Education Message: As noted in the AZA Accreditation Standards, if animal demonstrations are part of an institution's programs, an educational and conservation message must be an integral component. The Program Animal Policy should address the specific messages related to the use of program animals, as well as the need to be cautious about hidden or conflicting messages (e.g., “petting” an animal while stating verbally that it makes a poor pet). This section may include or reference the AZA Conservation Messages.

Although education value and messages should be part of the general collection planning process, this aspect is so critical to the use of program animals that it deserves additional attention. In addition, it is highly recommended to encourage the use of biofacts in addition to or in place of the live animals. Whenever possible, evaluation of the effectiveness of presenting program animals should be built into education programs.

VI. Human Health and Safety: The safety of our staff and the public is one of the greatest concerns in working with program animals. Although extremely valuable as educational and affective experiences, contact with animals poses certain risks to the handler and the public. Therefore, the human health and safety section of the policy should address:

- Minimization of the possibility of disease transfer from non-human animals to humans, and vice-versa (e.g., hand washing stations, no touch policies, use of hand sanitizer).
- Safety issues related to handlers' personal attire and behavior (e.g., discourage or prohibit use of long earrings, perfume and cologne, not eating or drinking around animals, smoking etc.).

AZA’s Animal Contact Policy provides guidelines in this area; these guidelines were incorporated into accreditation standards in 1998.

VII. Animal Health and Welfare: Animal health and welfare are the highest priority of AZA accredited institutions. As a result, the Institutional Program Animal Policy should make a strong statement on the importance of animal welfare. The policy should address:

- General housing, husbandry, and animal health concerns (e.g. that the housing and husbandry for program animals meets or exceeds general standards and that the needs of the individual animal, such as enrichment and visual cover, are accommodated).
- The empowerment of handlers to make decisions related to animal health and welfare; such as withdrawing animals from a situation if safety or health is in danger of being compromised.
- Requirements for supervision of contact areas and touch tanks by trained staff and volunteers.
- Frequent evaluation of human/animal interactions to assess safety, health, welfare, etc.
- Ensure that the level of health care for the program animals is consistent with that of other animals in the collection.

VIII. Taxon Specific Protocols: The AZA encourages institutions to provide taxonomically specific protocols, either at the genus or species level, or the specimen, or individual, level. Some taxon-specific guidelines may affect the use of program animals. To develop these, institutions refer to the Conservation Programs Database. Taxon-specific protocols should address:

- How to remove the individual animal from and return it to its permanent enclosure.
- How to crate and transport animals.
- Signs of stress, stress factors and discomfort behaviors.
- Situation specific handling protocols (e.g., whether or not animal is allowed to be touched by the public, and how to handle in such situations)
- Guidelines for disinfecting surfaces, transport carriers, enclosures, etc.
- Animal facts and conservation information.
- Limitations and restrictions regarding ambient temperatures and or weather conditions.
- Time limitations (including animal rotation and rest periods, as appropriate, duration of time each animal can participate, and restrictions on travel distances).

- The numbers of trained personnel required to ensure the health and welfare of the animals, handlers and public.
- Taxon-specific guidelines on animal health.

IX. Logistics, and Managing the Program: The Institutional Policy should address a number of logistical issues related to program animals, including:

- Where and how the program animal collection will be housed, including any quarantine and separation for animals used off-site.
- Procedures for requesting animals, including the approval process and decision making process.
- Accurate documentation and availability of records, including procedures for documenting animal usage, animal behavior, and any other concerns that arise.

X. Staff Training: Thorough training for all handling staff (keepers, educators, and volunteers, and docents) is clearly critical. Staff training is such a large issue that many institutions may have separate training protocols and procedures. Specific training protocols can be included in the Institutional Program Animal Policy or reference can be made that a separate training protocol exists. It is recommended that the training section of the policy address:

- Personnel authorized to handle and present animals.
- Handling protocol during quarantine.
- The process for training, qualifying and assessing handlers including who is authorized to train handlers.
- The frequency of required re-training sessions for handlers.
- Personnel authorized to train animals and training protocols.
- The process for addressing substandard performance and noncompliance with established procedures.
- Medical testing and vaccinations required for handlers (e.g., TB testing, tetanus shots, rabies vaccinations, routine fecal cultures, physical exams, etc.).
- Training content (e.g., taxonomically specific protocols, natural history, relevant conservation education messages, presentation techniques, interpretive techniques).
- Protocols to reduce disease transmission (e.g., zoonotic disease transmission, proper hygiene and hand washing requirements, as noted in AZA's Animal Contact Policy).
- Procedures for reporting injuries to the animals, handling personnel or public.
- Visitor management (e.g., ensuring visitors' interact appropriately with animals, do not eat or drink around the animal, etc.).

XI. Review of Institutional Policies: All policies should be reviewed regularly. Accountability and ramifications of policy violations should be addressed as well (e.g., retraining, revocation of handling privileges, etc.). Institutional policies should address how frequently the Program Animal Policy will be reviewed and revised, and how accountability will be maintained.

XII. TAG and SSP Recommendations: Following development of taxon-specific recommendations from each TAG and SSP, the institution policy should include a statement regarding compliance with these recommendations. If the institution chooses not to follow these specific recommendations, a brief statement providing rationale is recommended.

Appendix F: AZA Accreditation Standards for Otters

The following specific standards of care and recommendations for otters are taken from the AZA Accreditation Standards and Related Policies (AZA 2008):

Water Quality: The provision of fresh potable water is a requirement of USDA Animal Welfare Regulations (AWR 2005) as stated: "If potable water is not accessible to the animals at all times, it must be provided as often as necessary for the health and comfort of the animal. Frequency of watering shall consider age, species, condition, size, and type of the animal. All water receptacles shall be kept clean and sanitary" (AWR 2005). Considering the needs of otters, the AZA Small Carnivore TAG state that otters should be given fresh water daily if their pools are not filtered or dumped and filled daily on a daily basis. AZA Accreditation Standards require that institutions abide by relevant federal laws and regulations:

"*The institution must comply with all relevant local, state, and federal wildlife laws and regulations. It is understood that, in some cases, AZA accreditation standards are more stringent than existing laws and regulations. In these cases the AZA standard must be met*" (AZA 2008).

Transport: The standards of care identified in Chapter 3 are based on IATA regulations (IATA 2007). Institutions transporting otters are obliged to abide by these regulations as stated in the following AZA Accreditation Standard:

"*The institution must comply with all relevant local, state, and federal wildlife laws and regulations. It is understood that, in some cases, AZA accreditation standards are more stringent than existing laws and regulations. In these cases the AZA standard must be met*" (AZA 2008).

Quarantine: "*Quarantine, hospital, and isolation areas should be in compliance with standards/guidelines adopted by the AZA*" (AZA 2008)

Quarantine facility: "A separate quarantine facility, with the ability to accommodate mammals, birds, reptiles, amphibians, and fish should exist. If a specific quarantine facility is not present, then newly acquired animals should be isolated from the established collection in such a manner as to prohibit physical contact, to prevent disease transmission, and to avoid aerosol and drainage contamination. Such separation should be obligatory for primates, small mammals, birds, and reptiles … More stringent local, state, or federal regulations take precedence over these recommendations."

Quarantine length: "Quarantine for all species should be under the supervision of a veterinarian and consist of a minimum of 30 days (unless otherwise directed by the staff veterinarian). Mammals: If during the 30-day quarantine period, additional mammals of the same order are introduced into a designated quarantine area, the 30-day period begins over again. However, the addition of mammals of a different order to those already in quarantine will not have an adverse impact on the originally quarantined mammals."

Quarantine personnel: "A keeper should be designated to care only for quarantined animals or a keeper should attend quarantined animals only after fulfilling responsibilities for resident species. Equipment used to feed and clean animals in quarantine should be used only with these animals. If this is not possible, then equipment must be cleaned with an appropriate disinfectant (as designated by the veterinarian supervising quarantine) before use with post-quarantine animals. Institutions must take precautions to minimize the risk of exposure of animal care personnel to zoonotic diseases that may be present in newly acquired animals. These precautions should include the use of disinfectant foot baths, wearing of appropriate protective clothing and masks in some cases, and minimizing physical exposure in some species; e.g., primates, by the use of chemical rather than physical restraint. A tuberculin testing/surveillance program must be established for zoo/aquarium employees in order to ensure the health of both the employees and the animal collection."

Quarantine protocol: "During this period, certain prophylactic measures should be instituted. Individual fecal samples or representative samples from large numbers of individuals housed in a limited area (e.g., birds of the same species in an aviary or frogs in a terrarium) should be collected at least twice and examined for gastrointestinal parasites. Treatment should be prescribed by the attending veterinarian.

Ideally, release from quarantine should be dependent on obtaining two negative fecal results spaced a minimum of two weeks apart either initially or after parasiticide treatment. In addition, all animals should be evaluated for ectoparasites and treated accordingly. Vaccinations should be updated as appropriate for each species. If the animal arrives without a vaccination history, it should be treated as an immunologically naive animal and given an appropriate series of vaccinations. Whenever possible, blood should be collected and sera banked. Either a -70ºC freezer or a -20ºC freezer that is not frost-free should be available to save sera. Such sera could provide an important resource for retrospective disease evaluation. The quarantine period also represents an opportunity to, where possible, permanently identify all unmarked animals when anesthetized or restrained (e.g., tattoo, ear notch, ear tag, etc.). Also, whenever animals are restrained or immobilized, a complete physical, including a dental examination, should be performed. Complete medical records should be maintained and available for all animals during the quarantine period. Animals that die during quarantine should have a necropsy performed under the supervision of a veterinarian and representative tissues submitted for histopathologic examination."

Quarantine procedures: The following are recommendations and suggestions for appropriate quarantine procedures for small carnivores:

Required	Strongly recommended
1) Direct and floatation fecal exam	1) CBC/sera profile
2) Vaccinate as appropriate	2) Urinalysis
	3) Appropriate serology (FIP, FeLV, FIV)
	4) Heartworm testing in appropriate species

Appendix G: Giant Otter Enclosure Design

Adapted excerpts from the “International Giant Otter Studbook Husbandry and Management Information and Guidelines (2005)” for WAZA Website, and with added U.S. units of measurements (Sheila Sykes-Gatz 2006).

The total minimum size enclosure for one giant otter pair should be at least 2,583ft^2 (240m^2) and when indoor enclosures are needed, they should be a minimum of 807ft^2 (75m^2) of the total area. In temperate climates, it is necessary that an outdoor enclosure (with or without heated outdoor water) provides access to a heated indoor enclosure. Both indoor and outdoor enclosures require the same recommended conditions (i.e., land to water ratios, substrates etc.). At least 2 dens (i.e., separable rooms) to contain nest boxes/briefly hold animals should be provided, and at least 2-3 nest boxes (or natural underground dens) are needed.

Providing the recommended land to water area ratios (i.e., enough land area), substrate types and depths to cover all surfaces, and deep digging area sizes, is just as important as the need to provide a swim area in all giant otter enclosures. These are among the most crucial husbandry provisions needed to maintain giant otter physical and behavioral health, and they are also necessary to promote successful pup-rearing and adjustment to new/unusual situations. To meet these needs, nearly the entire enclosure surface area, including dens and nestboxes, must keep sufficiently dry, soft, and sanitary and otters need to be able to effectively dig and groom throughout that entire area. The enclosure must also offer a sufficient proportion of land, deep digging, and water area. The provisions in the two paragraphs below are essential to providing these conditions.

Different enclosure sizes require different land to water area ratios. As enclosure sizes decrease below 2,583ft^2 (240m^2), proportionately increased land area sizes are needed. It is necessary to provide, within each indoor and outdoor giant otter enclosure, at least the minimum percentage land area that the following formula determines. Convert enclosure sizes to m^2 (ft^2 x 0.093 = m^2). For every 1m^2 that the (indoor or outdoor) enclosure size is below 240m^2, multiply that number (without the m^2 symbol) by 0.1, then add the result to the number 60, and this resulting number is the minimum land area percentage (%) that the (indoor or outdoor) enclosure requires. For example, a 1,615ft^2 (150m^2) enclosure requires a minimum of 69% land area, and an 807ft^2 (75m^2) enclosure requires at least 76.5% land area. Enclosures between 2,583-6,458ft^2 (240-600m^2) in size require at least 60% land area.

It is crucial that nearly the entire area of surfaces/substrates that giant otters are directly exposed to are soft, natural, well-draining, not coarse, sufficiently dry and deep, and loose enough so that otters can easily dig into them. It is necessary that every indoor and outdoor enclosure surface, including dens, is nearly entirely covered with soft pebble-free sand or mulch (i.e., tree bark pieces only), at least 4-8" (10-20cm) in depth, or deep soft loose soil with the needed qualities. The substrates used should not have gravel, pebbles, rocks/stones <8" (20cm) in dia./width, wood chips, or abrasive sand mixed throughout them and if areas of these individual or combined substrates already exist, they should be removed or covered over with at least 24" (60cm) of a recommended substrate. Otters should not be directly exposed to these inappropriate substrates or to more than a small area of hard, artificial, tightly packed, continually damp/wet or poor draining surfaces. Many soils are or will become too packed or will not remain dry enough after otters dig, clear vegetation and track water throughout them; these activities should not be prevented. Soil should not be used to cover hard or artificial surfaces. Sand and mulch are ideal to cover over any surface/substrate. Add new mulch/sand on top of the existing layer (e.g., yearly) to maintain minimum depth/cover broken down mulch. Each indoor and outdoor giant otter enclosure also needs at least a 430ft^2 (40m^2) area, where sand or mulch, a minimum of 16-24" (40-60cm) in depth, or soil hillsides, allow for deep digging. The hillsides should be at least 6.5ft (2m) high and have an angle no more or less steep than 40-45^o.

The following conditions, with the recommended land to water ratios and substrates, are needed to keep surfaces sufficiently dry. In every enclosure, the land area bordering the water area should extend at least 16ft (5m) in the direction leading away from the water’s edge. In enclosures below or ca. 807ft^2 (75m^2) in size, the land area should only be bordered by water on one of its sides and in enclosures ca. 2,583ft^2 (240m^2) in size, no more than two sides of the land area should be bordered by water. Also, long water area contour lines should not be used in enclosures ca. or below 2,583ft^2 (240m^2) in size, but varied contour shapes are recommended. Dens and nestboxes should be located at least 10ft (3m) away from the water’s edge.

Pools should have deep areas (at least 3.28ft (100cm) deep), shallow areas (which are frequently used), and plentiful areas of gently sloping edges for safe pup exits. Varied natural furnishings, e.g., logs, tree stumps with roots, cut bamboo, boulders, should be placed on land and in and over pools. Thin logs connected with brackets or large sloping rocks placed just behind and bordering pool edges, fence covered drains/filters, and drain pipe extensions help prevent substrates from entering water areas, cleaning and drainage systems. Furnishings should allow otters, especially pups and parents, easy and safe pool access and exits. Enclosure designs, furnishings, and husbandry methods that offer visual and acoustic privacy from human disturbances (zoo staff and visitors) during pup-rearing and that allow safe gradual introduction of unfamiliar or temporarily separated otters should be provided. Fish should be fed exclusively. A variety of good quality fresh water fish, low in thiaminase and fat, should be offered as the main diet. Saltwater fish can be offered occasionally.

Appendix H: Description of Nutrients

Protein: Protein is the main building blocks of animal structure on a fat-free basis. In addition to being an important constituent of animal cell walls, protein is one of the nutrients responsible for making enzymes, hormones, lipoproteins, and other crucial elements needed for proper bodily functions. Protein also is essential for building and repairing body tissue, as well as protecting the animal from harmful bacteria and viruses. Furthermore, protein aids in the transportation of nutrients throughout the body and facilitates muscle contractions. The requirements for crude protein are effectively requirements for dietary amino acids. The requirements are based on the needs of the animal, the quality of the protein, the source of the protein, and the digestibility of the protein available.

Fat: Dietary fat plays an important role in the manufacture of certain hormones. It also plays a crucial role in a wide variety of chemical bodily functions. Also, fat functions as a concentrated energy source, serves as a carrier for fat-soluble vitamins (Vitamins A, D, E, and K), and provides essential fatty acids. The requirements for fat are effectively requirements for dietary fatty acids.

Vitamin A: Vitamin A is a fat-soluble vitamin essential for maintaining good vision and healthy mucous membranes. It contributes to the differentiation and growth of skin tissue and bone formation (including teeth), as well as bone remodeling in growing animals, and glycoprotein synthesis. Vitamin A can improve skin and hair/fur conditions, help to increase resistance to certain infections, and improve fertility in both genders. In many cases, a vitamin A requirement is effectively a requirement for carotenoids (precursors to vitamin A).

Vitamin C (Ascorbic Acid): Vitamin C is a water-soluble antioxidant, which plays an important role in biochemical oxidation-reduction reactions, as well as in the formation of collagen, an important protein needed for the formation of skin, scar tissue, tendons, ligaments, and blood vessels. Because of this, Vitamin C is crucial to an animal's ability to heal wounds and repair and or maintain cartilage, teeth, and bones. It also may reduce infection by increasing immunity.

Vitamin D: Vitamin D is a fat-soluble vitamin necessary for active calcium absorption, calcium metabolism and resorption from bone. Requirements for vitamin D can be totally or partially met by exposure to sunlight or artificial UV light (vitamin D is biosynthesized in the skin of animals or in some plant cells upon exposure to the appropriate wavelength of UV light; 285-315nm).

Vitamin E: Vitamin E is a fat-soluble antioxidant that helps to maintain the structure of cellular and subcellular membranes by preventing oxidation of unsaturated fatty acids. It also protects tissues from free radicals, which are substances known to harm cells, tissues, and organs. Vitamin E is essential in the formation of red blood cells and aids the body in Vitamin K utilization.

Thiamine (B-1): Thiamine is a water-soluble vitamin, which functions as a necessary coenzyme in carbohydrate metabolism (converting carbohydrates into energy) and is hypothesized to play a role in nerve or neuromuscular impulse transmission. Thiamine also is important in the proper functioning of the heart, muscles, and the nervous system.

Riboflavin (B-2): Riboflavin is a water-soluble vitamin. It functions in two coenzymes: Flavin adenine dinucleotide or "FAD" and flavin mononucleotide. Riboflavin is important for growth and the production of red blood cells. It also helps the body to release energy from carbohydrates. Microbial synthesis of riboflavin occurs in the gastrointestinal tract of some animals, but synthesis appears to be dependent on the type of animal and the source of dietary carbohydrate.

Niacin (Nicotinic Acid): Similar to Riboflavin, niacin is a water-soluble vitamin which functions in two coenzymes: Nicotinamide adenine dinucleotide or "NAD" and nicotinamide adenine dinucleotide phosphate or "NADP". Niacin plays a crucial role in assisting the normal functioning of the digestive, skin, and nerve systems. Like riboflavin, niacin helps the body to convert energy from food. The niacin requirement of many animals theoretically could be satisfied by synthesis of the vitamin from the amino acid tryptophan. However, removal rate of an intermediate in the pathway to create niacin is often so rapid that virtually none is produced.

Pyridoxine (B-6): Pyridoxine also known as B-6 is a water-soluble vitamin, which aids the body in the synthesis of antibodies by the immune system. It also plays a role in the formation of red blood cells and helps to promote healthy nerve functions. Pyridoxine is required to produce the chemical activity necessary for protein digestion.

Choline: Choline is an essential nutrient, which contributes to the function of nerve cells. It is a component (helps to form phosphatidylcholine, the primary phospholipid of cell membranes) of the phospholipid lecithin (found in cells throughout the body) and is critical to normal membrane structure and formation. It also functions as a "methyl donor", but this role can be completely replaced by excess amounts of the amino acid methionine in the diet.

Folacin (Folate, Folic Acid, B-9, Pteroylglutamic Acid): Folacin, or folate, is a water-soluble vitamin, which assists the body in the formation of red blood cells. It also plays a major role in the formation of genetic material (synthesis of DNA, the hereditary and functioning blueprint of all cells) within all living cells. Folacin functions as a coenzyme, which is important at the cellular and subcellular levels in decarboxylation, oxidation-reduction, transamination, deamination, phosphorylation, and isomerization reactions. Working in conjunction with Vitamin C and B-12, Folacin assists in digestion and protein utilization and synthesis. This vitamin may be used to increase appetite and stimulate healthy digestive acids.

Vitamin B-12: Vitamin B-12 is a water-soluble vitamin, which functions as a coenzyme in single carbon and carbohydrate metabolism. In addition to playing a role in metabolism, B-12 assists in the formation of red blood cells and aids in the maintenance of the central nervous system.

Pantothenic Acid: Pantothenic acid is a water-soluble vitamin and part of the B vitamin complex. It is needed to break down and use (metabolize) food. Pantothenic acid also is needed for the synthesis of both hormones and cholesterol.

Calcium: The mineral calcium (in association with phosphorus) is a major component of the body and is largely associated with skeletal formation. It is important in blood clotting, nerve function, acid-base balance, enzyme activation, muscle contraction, and eggshell, tooth, and bone formation and maintenance. It is one of the most important minerals required for growth, maintenance, and reproduction of vertebrates.

Phosphorus: In addition to acting as a major component of the body and being largely associated with skeletal and tooth formation (in conjunction with calcium), phosphorus is involved in almost every aspect of metabolism (energy metabolism, muscle contractions, nerve function, metabolite transport, nucleic acid structure, and carbohydrate, fat, and amino acid metabolism). Phosphorus is needed to produce ATP, which is a molecule the body uses to store energy. Working with the B vitamins, this mineral also assists the kidneys in proper functioning and helps to maintain regularity in heartbeat.

Magnesium: Magnesium is a mineral, which serves several important metabolic functions. It plays a role in the production and transport of energy. It also is important for the contraction and relaxation of muscles. Magnesium is involved in the synthesis of protein, and it assists in the functioning of certain enzymes in the body.

Potassium: Potassium is a mineral that is involved in both electrical and cellular functions in the body. (In the body it is classified as an electrolyte.) It has various roles in metabolism and body functions. Potassium assists in the regulation of the acid-base balance and water balance in blood and the body tissues. It also assists in protein synthesis from amino acids and in carbohydrate metabolism. Potassium is necessary for the building of muscle and for normal body growth, as well as proper functioning of nerve cells, in the brain and throughout the body.

Sodium (salt): Sodium is an element, which the body uses to regulate blood pressure and blood volume. Sodium also is critical for the functioning of muscles and nerves.

Iron: Iron is a trace element and is the main component of hemoglobin (oxygen carrier in the blood), myoglobin in muscles (oxygen carrier with a higher affinity for oxygen than hemoglobin), and many proteins and enzymes within the body. It also functions in immune defenses against infection.

Zinc: Zinc also is a trace element that is second only to iron in terms of concentration within the body. Zinc plays an important role in the proper functioning of the immune system in the body. It is required for the enzyme activities necessary for cell division, cell growth, and wound healing. It plays a role in the acuity of the senses of smell and taste. Zinc also is involved in the metabolism of carbohydrates. Zinc is essential for synthesis of DNA, RNA, and proteins, and it is a component or cofactor of many enzyme systems.

Manganese: Manganese is essential for carbohydrate and lipid metabolism, for synthesis of one of the precursors to cartilage formation, and for proper bone formation. Manganese plays a key role in the growth and maintenance of tissues and cartilage, specifically proper bone development. It particularly aids in development at the ends of bones where new bone formation takes place. This therefore helps to reduce the risk of osteoporosis. Manganese also helps to produce certain hormones, metabolizes fat, and is part of superoxide dismutase (SOD) an antioxidant. Studies on humans have shown that manganese also may lower the frequency of epileptic seizures and enhance immune functioning.

Copper: Copper is an essential trace mineral present in all body tissues. Copper, along with iron, helps in the formation of red blood cells. It also helps in keeping the blood vessels, bones, and nervous and immune systems healthy.

Selenium: Selenium is an essential trace element. It is an integral part of enzymes, which are critical for the control of the numerous chemical reactions involved in brain and body functions. Selenium has a variety of functions. The main one is its role as an antioxidant in the enzyme selenium-glutathione-peroxidase. This enzyme neutralizes hydrogen peroxide, which is produced by some cell processes and would otherwise damage cell membranes. Selenium also seems to stimulate antibody formation in response to vaccines. It also may provide protection from the toxic effects of heavy metals and other substances. Selenium may assist in the synthesis of protein, in growth and development. In humans, selenium has been shown to improve the production of sperm and sperm motility.

Iodine: Iodine is a trace mineral and an essential nutrient. Iodine is essential for the normal metabolism of cells. It is a necessary nutrient for the production of thyroid hormones and normal thyroid function.

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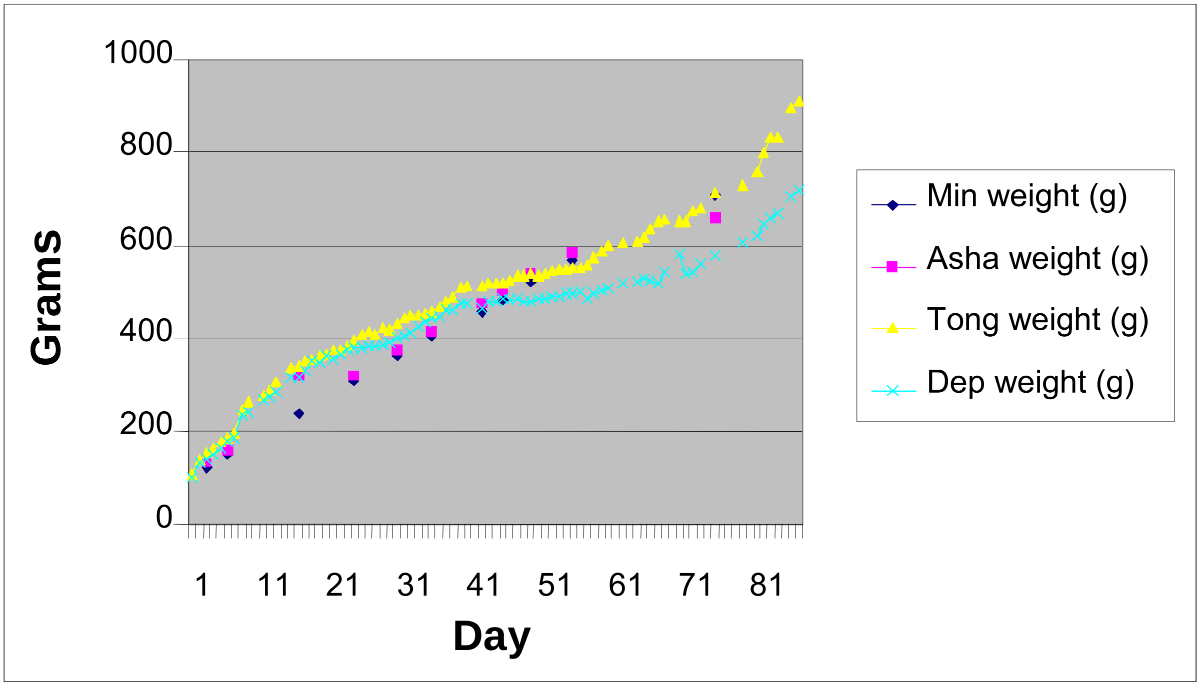
Appendix I: Pup Weights of *Ex-situ* Population Bred Otters

Asian-small clawed otter (*A. cinereus*) pup weights (mother-reared) at Institution F (N = 1.3)



Spotted-necked otter (*L. maculicollis*) pup weights (mother-reared) at Institution M N=1.0

Age in days	Weight (kg)	Age in days	Weight (kg)
28	0.75	98	1.9
35	0.95	107	1.9
42	1.2	113	1.95
49	1.4	120	2.1
56	1.5	127	2.7(after eating)
65	1.6	134	2.5
72	1.65	140	2.6
77	1.8	148	2.5
84	1.75	155	2.45
91	1.9	-	-

North American river otter (*L. canadensis*) pup weights (mother-reared). Data taken from AAZK Zoo Infant Development Notebook 1994, Institution N, Institution O, Institution P.

Males (N = 9)				**Females** (N = 8)			
Age/days	**Weight (g)**	**Age/days**	**Weight (kg)**	**Age/days**	**Weight (g)**	**Age/days**	**Weight (kg)**
1	110-170	**32**	0.992-1.03	**1**	170	**32**	0.971
2	177-184	**33**	0.998-1.09	**2**	177	**33**	-
3	193-220	**34**	1.08-1.11	**3**	198	**34**	1.01
4	204-241	**35**	1.11-1.14	**4**	213	**35**	1.05-1.15
5	241-276	**36**	1.13-1.19	**5**	248	**36**	1.06
6	249-298	**37**	1.16-1.18	**6**	262	**37**	1.09-1.23
7	266-333	**38**	1.20-1.25	**7**	298	**38**	1.13
8	280-354	**39**	1.23-1.28	**8**	333	**39**	1.15-1.30
9	325-376	**40**	1.28-1.34	**9**	347	**40**	1.23
10	353-404	**41**	1.35-1.36	**10**	383	**41**	1.28
11	364-425	**42**	1.32-1.41	**11**	397	**42**	1.25-1.35
12	398-453	**43**	1.35-1.39	**12**	411	**43**	1.28
13	414-475	**44**	1.40-1.43	**13**	439	**44**	1.35
14	496	**45**	1.45-1.57	**14**	454	**45**	1.39
15	531-539	**46**	1.52-1.62	**15**	489	**46**	1.43
16	499-574	**47**	1.43-1.62	**16**	517	**47**	1.34-1.48
17	595	**48**	1.59-1.69	**17**	546	**48**	1.46-1.60
18	617-624	**49**	1.59-1.67	**18**	560	**49**	1.58
19	624-645	**50**	1.69-1.79	**19**	609-685	**50**	1.62
20	666-680	**51**	1.62-1.74	**20**	637	**51**	1.56
21	687	**52**	1.67-1.87	**21**	652	**52**	1.53
22	765-780	**53**	1.74-1.88	**22**	660-730	**53**	1.62
23	780-808	**54**	1.74-1.92	**23**	723	**54**	1.64
24	810-843	**55**	1.71-1.96	**24**	758-850	**55**	1.66-1.81
25	822-858	**56**	1.54-1.68	**25**	720-795	**56**	-
26	829-872	**57**	1.71-2.03	**26**	772	**57**	1.93
27	850-872	**58**	1.87-2.10	**27**	794	**58**	1.76
28	865-910	**59**	1.90-2.06	**28**	815-900	**59**	1.80
29	907-921	**60**	1.52-2.12	**29**	872	**60**	1.86-1.70
30	935-978	**61**	1.97-2.15	**30**	907	**61**	1.84-2.33
31	971-1000	**62**	1.96-2.24	**31**	928-1060	**62**	1.88

Appendix J: List of Commonly Trained Behaviors for Otters

(Adapted from AAZK Animal Training Committee)

Animal Training Committee AAZK, Inc.

Commonly Trained Behaviors for: Mustelids, Procyonids, and Viverrids

Purpose of the list and source of the data:
The following list of behaviors was derived using data from a 2003 survey conducted by the American Association of Zoo Keepers Animal Training Committee (AAZK, ATC). The goal of the survey was to census the existence and depth of training programs for species in AZA facilities. For each species trained, each respondent was asked to list trained behaviors, types of reinforcement, and conditioned reinforcers used. Additional information about facility design, training tools, and general comments was also requested.

Survey results pertaining to the list of behaviors:
219 AZA facilities were surveyed. There were 71 respondents. 31 of these train species within the Mustelid, Procyonid and Viverid taxonomic group. Because many similarities were found within taxonomic groups, commonly trained behaviors were compiled to serve as a reference for animal training programs. Of the 31 respondents that train within this group, the percentage that train each behavior is listed next to the behavior.

Facility Differences and Individual Animals:
Not every behavior will work for every animal. The appropriateness of a behavioral goal for an individual will depend on management policy and building design of the facility, as well as the needs and disposition of the animal.

The ATC hopes that this data will aid in the design of training programs for the Mustelid, Procyonid, and Viverrid taxa. Where appropriate, these commonly trained behaviors can greatly enhance the husbandry of species in this group. For questions or comments about this list or the Trained Behaviors Survey, please contact the AAZK Animal Training Committee at www.aazk.org. See next page for chart.

Animal Training Committee AAZK, Inc.

Commonly Trained Behaviors for: Mustelids

MUSTELIDS (Behavior and % of responding institutions)

Otter (river, small-clawed, & sea)		**Skunk (striped and spotted)**	
Shifting	83%	Shifting	33%
Separations	66%	Separations	17%
Target	79%	Target	67%
Scale	59%	Scale	33%
Squeeze entry/crate	76%	Harness training	17%
Anal/genital present	7%	Crate/Squeeze entry	83%
Back	10%	Nail trim	33%
Belly	21%	Back	17%
Ears	7%	Belly	17%
Otters (continued)		**Skunk (continued)**	
Eyes	14%	Paws/feet	33%
Head presentation	14%	Tactile desensitization	50%
Mouth	21%	Station	17%
Paws/feet	52%		
Sides	10%	**Badger**	
Oral meds	21%	Shifting	100%
Brushing teeth	3%	Target	0
Injection w/ syringe	7%	Scale	0
Stethescope	3%		
In-water behaviors	31%	**Wolverine**	
Vocalization	7%	Belly	50%
Stay (hold)	10%	Paws/feet	50%
Retrivals	17%	Separations	100%
Station	59%	Shifting	100%
Fecal Collection	7%	Station	50%
A to B	10%	Squeeze/crate	50%
Climb	24%	Flashlight desensitization	50%
Flashlight	7%	Scale	50%
X-ray	3%		
Opthalmoscope	3%		
Blood collection	3%		

Appendix K: Enrichment Items Commonly Provided to Otters

The table below lists items used at various North American facilities for behavioral and environmental enrichment of otters.

Natural	Exhibit Furniture	Non-edible manmade	Live Food	Edibles
- Soil, sand, mulch - Grass, wheat grass, sedges, etc. - Trees - Vines "vine hoops" - Aquatic plants - Hay, straw, grass, leaves, wood wools as bedding - Grass piles - Leaf piles - Rocks, all sizes for play and manipulation - Knot holes - Bark sheets - Pine Cones - Mud - Sod - Bank over-hangs - Floating wood - Blocks - Pine needles - Other animal urines - Powdered scents and herbs - Fresh herbs - Extracts, i.e., vanilla, etc. - Grapevine balls - Shells - Turkey feathers - Corn stalks - Blowing bubbles into exhibit - Kudzu vines - Cow hooves	- Climbing areas (available in all exhibits, i.e., cliffs, ledges) - Logs (on land, submerged, floating; hollow and/or solid) - Rocks (not artificial) - Stream - Sticks - Browse (leafy branches on land and/or floating) - Slides - Tunnels - Stream bed - Running water - Holts - Jacuzzi-like jets in pool - Islands in pool - Bridges made from logs, etc. - Stumps - Natural fiber mat - Movable sand box - Logs brought from other exhibits - Log ladder - Non-sprayed evergreen trees - Moving soil pots - Hanging logs with holes for food - Snow piles - Piles of ice cubes	- Boomer balls and other products like the "spoolie", “bobbin” & "ice cube". - Ice blocks, cubes, pops. - Snow & ice - PVC cricket feeder - Buckets - Blankets, burlap, non-fraying rags, towels - Barrels of water - Frisbees - Tubs of water - Carpet over board - Rubber-coated heating pad* - Astro turf - Floating plastic toys - Phone books - Swim through plastic ring - Kids puzzle balls, billiard balls, hard balls - Pieces of PVC pipe and fittings - Kong chews - Metal bowls and pans - Plastic tubs and bottles - Bread tray - Plastic slide, house - Stock tank - Hanging tub* - Warm water hose - Vari-kennel tubs with substrates - PVC tube hung for climbing in	- Fish (smelt, shiners, goldfish, trout, mackerel, tilapia salmon)* - Crayfish - Crickets - Giant mealworms - Earthworms - Freshwater clams - Mussels - Krill - Eels- naturally found - Shrimp - Aquatic insects - naturally found - Mice- naturally found - Frogs – naturally found - Grubs - Chub - Minnows - Bluegill - Clams - Mud minnows	- Ice blocks w/fish, fish-sicles, fish cubes, etc. - krill cubes, clam cubes, etc. - Frozen or thawed sand eels - Fish pieces - Chicken necks - Mice - Whole-fish -frozen or thawed - Whole apples/oranges - Fruit & berries incl. grapes, blueberries, strawberries - Small pumpkins and squash - Omnivore biscuits - Monkey chow - Pigs ears - Frozen blood blocks, cubes, etc. - Hard-boiled eggs - Day-old chicks - Crabs - Melons - Coconuts - Frozen feline balls - Milk bones - Screw pine nuts, unsalted peanuts - Krill patties - Hamster ball w/ treat - Gelatin Jigglers - Corn on the cob - Yogurt with fish - Unsalted ham

* These items should be monitored for safety.

The following list provides more examples of enrichment initiatives offered to otters at the Institution J and Institution L:

Institution J – ASC otter

Non-food items
- Boomer balls & Jolly balls
- Bowling pins
- Brushes
- Bucket lids
- Beer kegs, feed barrels & trash cans
- Feed bags
- Clover clumps
- Milk crates, Plastic wagons & Plastic logs
- Water cooler bottles
- Grass flats/clumps
- Hang paper maché figures
- Hollow coconut shells
- Oscillating fan, wind chimes, & bubble machine (outside of enclosure)
- Large logs, rearrange furniture, etc.
- Leaves, sand, and rock piles
- PVC tubes
- Towels, clothes, blankets
- Cardboard boxes and tubes (caution needed when using paper products that can become wet)
- Laser pointer
- Nature tapes
- Perfume/body sprays & Glad scented sprays
- Traffic cones
- Hummus
- Ice piles
- Rose petals
- Burlap sacs
- Straw piles
- Reindeer antlers
- Varied of feeding devices & times
- Nyla bones
- Spices and extracts
- Mirror

Food items
- Honey smears
- Blood popsicles
- Cooked chicken
- Crickets
- Horse meat
- Meal worms
- Peanut butter
- Pinkies
- Dry cat food
- Milk bones
- Tuna

Institution L – N. A. river otter/ASC otter

Non-food items
- Bobbin with smelt rubbed on it
- Whole coconuts to roll around
- Yellow pages
- Bengay™ ointment inside a boomer ball
- Log switching between animal exhibits
- Regular Alka Seltzer® in PVC tube (very small holes in PVC)
- Corn stalks
- Blocks of recycled plastic with holes drilled in them to dig food items out
- Crickets in PVC tube feeder
- PVC shaker toys
- Milk crates, cardboard box, use with caution
- Pinecone soaked in scents
- Extracts – vanilla, almond, lemon & spices
- Elephant manure
- Deodorant spray
- Reindeer antlers & pronghorn sheaths
- Paper maché
- Pig ears and cow hooves
- Painting
- Mustard or tomato sauce
- Large black kong toy
- Floating PVC tube to swim through

Food items
- Liver
- Anchovy paste
- Hard boiled eggs, apples, pumpkins, carrots, blueberries
- Gelatin jigglers
- Live crawdads, live trout in pool, crickets
- Frozen smelt ice blocks
- Blood popsicles
- Knuckles
- Beef hearts
- Mice and rats

Appendix L: Resources for Enrichment and Training

Enrichment

Coe J. 1992. Plan ahead for behavioral enrichment in environmental kaleidoscope: Research, management, and design. In: Proceedings of the AAZPA Annual Meeting. Wheeling WV:AAZPA. pp. 120-23.

du Bois T. 1992. The Los Angles Zoo environmental enrichment program: We get a lot of help from our friends. In: Proceedings of the AAZPA Annual Meeting. Wheeling. WV:AAZPA. pp. 112-19.

Goss D. 1999. Creating artificial vines for natural zoo displays. AAZK Forum: 26(11):449-51.

Grams K, Ziegler G. 1995. Enrichment companies and products. AAZK Forum: 22(6):210.

Hare V, Worley K. 1995. The shape of enrichment: The first generation. In: Proceedings of the AAZPA Annual Meeting. Wheeling. WV:AAZPA. pp.180-86.

Harris A. 1998. Enrichment: Trials, tribulations and magic moments. In: Proceedings of the AAZPA Annual Meeting. Wheeling. WV:AAZPA. pp. 271-74.

Houts L. 1999. Folsom City Zoo pinatas. AAZK Forum: 26(5): 177-78.

Houts L. 1999. Utilizing young visitors for behavioral enrichment. In: Proceedings of the AAZPA Annual Meeting. Wheeling. WV:AAZPA. pp 72-3.

Houts, L. 2000. Enhancing animal welfare with limited resources. Proceedings of the AZA Annual Meeting. Wheeling. WV: AZA.

Houts L, Greaves J. 2000. Enrichment options- Using Graphics to promote enrichment. AAZK Forum: 27(7): 307-08.

Lacinak CT, Turner TN, Kuczaj SA. 1995. When is environmental enrichment most effective? In: Proceedings of the AAZPA Annual Meeting. Wheeling. WV:AAZPA. pp. 324-27.

Lattis, R. 2000. Animal welfare and the AZA. Proceedings of the AZA Annual Meeting. Wheeling. WV:AZA.

Laule G. 1992. Addressing Psychological well being: Training as enrichment. In: Proceedings of the AAZPA Annual Meeting. Wheeling. WV:AAZPA. pp. 415-22.

Laule G, Desmond T. Positive reinforcement training as an enrichment strategy. In: Shepherdson DJ, Mellen JD, Hutchins M, editors. Second Nature: Environmental enrichment for captive animals. Washington: Smithsonian Institution Press; 1998. pp. 302-313.

Lindburg D. Enrichment of captive animals through provisioning. In: Shepherdson DJ, Mellen JD, Hutchins M, editors. Second Nature: Environmental enrichment for captive animals. Washington: Smithsonian Institution Press; 1998. pp. 262-301.

Markowitz, H. Behavioral enrichment in the zoo. New York: van Nostrand Reinhold Company; 1982.

Martin S. 1996. Training as enrichment. In: Proceedings of the AAZPA Annual Meeting. Wheeling. WV: AAZPA. pp. 139-41.

McPhee ME, Foster JS, Sevenich M, Saunders CD. 1996. Public perceptions of behavioral enrichment. In: Proceedings of the AAZPA Annual Meeting. Wheeling. WV:AAZPA. Pp 248-53.

Mellen J, Shepherdson D, Hutchins M. Epilogue: The future of environmental enrichment. In: Shepherdson, D, Mellen, J, Hutchins, M, editors. Second nature: Environmental enrichment for captive animals. Washington, DC: Smithsonian Institution Press; 1998. p. 329-336.

Mellen J, Sevenich M. 1999. Philosophy of animal enrichment: past, present, and future. In: Proceedings of the PAZAAB Conference. Cape Town: South Africa.

Moore, D. 1997. AZA Minimum Husbandry Guidelines for Mammals: Mustelids. AZA Mammal Standards Task Force, 1997. 7pg.

Moore, D, Reiss, D. 2000. A multi-institutional program addressing welfare, enrichment, husbandry training and behavioral research. Proceedings of the AZA Annual Meeting. Wheeling. WV: AZA.

Myers M. 1999. Feeder designs at the Audubon Park and Zoological Gardens. AAZK Forum: 26(11):369-72.

Seidensticker J, Forthman D. Evolution, ecology, and enrichment: Basic considerations for wild animals in zoos. In: Shepherdson DJ, Mellen JD, Hutchins M, editors. Second Nature: Environmental enrichment for captive animals. Washington: Smithsonian Institution Press; 1998. pp. 15-29.

Sevenich MacPhee, M, Mellen, J. 2000a. Framework for planning, documenting, and evaluating enrichment programs (and the Director's, Curator's, and Keeper's roles in the Process). Proceedings of the AZA Annual Meeting. Wheeling. WV:AZA.

Sevenich MacPhee, M, Mellen, J. 2000b. Training and enrichment models for avian exhibits. Proceedings of the AZA Annual Meeting. Wheeling. WV:AZA.
Sheng S. 1992. The ultimate enriched environment. In: Proceedings of the AAZPA Annual Meeting. Wheeling. WV:AAZPA. pp 124-27.
Shepherdson D. 1992. Environmental enrichment: an overview. In: Proceedings of the AAZPA Annual Meeting. Wheeling. WV:AAZPA. pp 100-3.
Shepherdson D. 1997. The animal's perspective: Developing strategies for successful enrichment. In: Proceedings of the AAZPA Annual Meeting. Wheeling. WV:AAZPA. pp 486-89.
Shepherdson, D. Tracing the path of environmental enrichment in zoos. In: Shepherdson, D, Mellen, J, Hutchins, M, editors. Second nature: Environmental enrichment for captive animals. Washington, DC: Smithsonian Institution Press; 1998. p. 1-14.
Shepherdson, D, Carlstead, K. 2000. When did you last forget to feed your tiger? Proceedings of the AZA Annual Meeting. Wheeling. WV:AZA.
Shepherdson D, Mellen J. 1993. Environmental enrichment: Carrying forth the message. In: Proceedings of the AAZPA Annual Meeting. Wheeling. WV:AAZPA. pp 216-18.
Shields J. 1995. Behavioral enrichment for the rest of the zoo. In: Proceedings of the AAZPA Annual Meeting. Wheeling. WV:AAZPA. pp 456-57.
Smith C. 1993. An inexpensive simulated thundershower for small enclosures. AAZK Forum: 20(9):322-23.
Stern S. 1994. Whose life is really being enriched here anyway? Shape of enrichment 3(3).
Widner K. 1994. Initiation of the zoo-wide enrichment program at the Knoxville Zoo. In: Proceedings of the AAZPA Annual Meeting. Wheeling. WV:AAZPA. pp 51-2.
Williams L. 1996. Ethological considerations for designing behavioral enrichment. Lab Animal. pp 29-33.
Whorley, K. E. & V. J. Hare. 1995. An overview of the 1995 Environmental Enrichment Conference. The Shape of Enrichment. Vol. 4, No. 4, November.

There are also many enrichment resources available on-line and in print, including:

"Enrichment Options" – A regular column featuring brief descriptions of ideas published monthly in the Animal Keepers' Forum. Published by the American Association of Zoo Keepers, Inc. AAZK Administrative Office, Susan Chan, Editor. 3601 S.W. 29th Street, Suite 133 Topeka, KS 66614. Phone: (785) 273-9149, Fax: (785) 273-1980. Email: akfeditor@zk.kscoxmail.com. Website: www.aazk.org
"The Shape of Enrichment" Newsletter – A newsletter devoted entirely to enrichment of captive wild animals. Published by The Shape of Enrichment, Inc., V. Hare & K. Worley, (eds.). 1650 Minden Drive, San Diego, CA 92111. Phone: (619) 270-4273. Fax: (619) 279-4208. E-mail: shape@enrichment.org. Website: www.enrichment.org
The American Association of Zoo Keepers Enrichment Committee: www.aazk.org
Disney Animal Kingdom - www.animalenrichment.org
AAZK Enrichment Notebook 3rd ed. 2004 ISBN1-929672-11-X, www.aazk.org/2004enrichnotebookcd.php
Fort Worth Zoo's Enrichment Online: www.enrichmentonline.org/browse/index.asp

Training

Baker A. 1991. Training as a management tool: Creating the climate and maintaining the momentum. In: AAZPA Annual Conference Proceedings. Wheeling, W.Va.: American Association of Zoological Parks and Aquariums, 563-568.
Blasko D, Doyle C, Laule G, Lehnhardt J. 1996. Training terms list. Principles of Elephant Management School. St. Louis: American Zoo and Aquarium Association, Schools for Zoo and Aquarium Personnel.
Bloomsmith M, Laule G, Thurston R, Alford P. 1992. Using training to moderate chimpanzee aggression. In: AAZPA Regional Conference Proceedings. Wheeling, W.Va.: American Association of Zoological Parks and Aquariums, 719-722.
Coe J. 1992. Animal training and facility design – a collaborative approach. In: AAZPA/CAZPA Regional Conference Proceedings. Wheeling, W.Va.: American Association of Zoological Parks and Aquariums, 411-414.

Coe J. 1992. Animal training and facility design – a collaborative approach. In: AAZPA/CAZPA Regional Conference Proceedings. Wheeling, W.Va.: American Association of Zoological Parks and Aquariums, 411-414.
Dewsbury D. 1978. Comparative animal behavior. New York: McGraw-Hill.
Grandin T. 1995. Thinking in pictures. New York: Vintage book. 222 p.
Hediger H. 1950. Wild animals in captivity. London: Butterworths. 207 p.
Hediger H. 1969. Man and animal in the zoo. London: Routledge and Kegon Paul. 303 p.
Kazdin A. 1994. Behavior modification in applied settings. Pacific Grove CA: Brooks/Cole Publishing Company. 508 p.
Kleiman D, Allen M, Thompson K, Lumpkin S, editors. 1996. Wild mammals in captivity: Principles and techniques.
Laule G. 1992. Addressing psychological well-being: Training as enrichment. In: AAZPA/CAZPA Annual Conference Proceedings. Wheeling, W.Va.: American Association of Zoological Parks and Aquariums, 415-422.
Laule G. 1995. The role of behavioral management in enhancing exhibit design and use. In: AZA Regional Conference Proceedings. Wheeling, W.Va.: American Association of Zoological Parks and Aquariums, 83-88.
Lyons J. 1991. Lyons on horses: John Lyons' proven conditioned-response training program. New York: Doubleday. 228p.
Mellen J, Ellis S. 1996. Animal learning and husbandry training. In: Kleiman D, Allen M, Thompson K, Lumpkin S, editors. Wild mammals in captivity: Principles and techniques. Chicago: The University of Chicago Press, 88-99.
Mellen J, Sevenich MacPhee M. (2001). Philosophy of environmental enrichment: Past, present, and future. Zoo Biology 20: 211-226.
Pryor K. 1984. Don't shoot the dog! Simon & Schuster: New York.
Pryor K. 1995. On behavior. North Bend: Sunshine Books, Inc.
Ramirez K. 1999. Animal training: Successful animal management through positive reinforcement. Chicago: Ken Ramirez and The Shedd Aquarium. 578 p.
Reichard T, Shellabarger W, Laule G. 1992. Training for husbandry and medical purposes. In: AAZPA Annual Conference Proceedings. Wheeling, W.Va.: American Association of Zoological Parks and Aquariums, 396-402.
Reynolds G. 1975. A primer of operant conditioning. Palo Alto: Scott, Foresman and Co.
Sevenich MacPhee M, Mellen J. 2000. Framework for planning, documenting, and evaluating enrichment programs (and the director's, curator's, and keeper's roles in the process). In: AAZPA Annual Conference Proceedings. Wheeling, W.Va.: American Association of Zoological Parks and Aquariums.
Shepherdson, D.J. 1998. Tracing the path of environmental enrichment in zoos. In: Shepherdson DJ, Mellen JD, Hutchins, M, editors. Second nature: Environmental enrichment for captive animals. Washington, DC: Smithsonian Institution Press, 1-12.
Shepherdson, D., Carlstead, K. 2000. When did you last forget to feed your tiger? In: AAZPA Annual Conference Proceedings. Wheeling, W.Va.: American Association of Zoological Parks and Aquariums.
Stevens, B., Groff, A., Joseph, S., Mellen, J., Ogden, J., Plasse, R., 2000. Evolving a new species of animal care professional: Combining AZA expertise with Disney leadership. In: AAZPA Annual Conference Proceedings. Wheeling, W.Va.: American Association of Zoological Parks and Aquariums.
Wilkes G. 1994. A behavior sampler. North Bend: Sunshine Books, Inc.

-S. Maher

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Appendix M: Missouri Fish and Wildlife Otter Stomach Contents

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Otter Stomachs containing identifiable fall prey items

Type	Percent
Crayfish	61
Fish	51
Frogs	17
Muskrats	3
Ducks	1
Empty	4
Ozark otter stomachs containing identifiable fish species	
Species	percent
Bass (sunfish family)	39
Suckers and Carp	31
Minnows	14
Shad	11
Pike (chain pickerel)	6
Trout	3
Catfish	3
Drum	3
Unidentified Fish	19

Age of Game Fish in Ozark Otter Stomachs

Age	Percent
1-3 years	40
4-6 years	40
7-9 years	20

Appendix N: Basic Considerations in the Design and Maintenance of Otter Exhibit Life Support Systems

FOREWORD

This document fulfills two objectives, 1) providing information on maintaining pools in otter exhibits (see Appendices) and 2) providing information to those who are thinking about building a new otter exhibit (or renovating an existing one) regarding some of the basic variables to consider. In our experience many operational problems stem from the design phase, and it can sometimes be useful to go back to the design and construction phase for answers.

Those with an existing exhibit and/ or with a specific issue may skip past the Planning and Design sections straight to the appendices.

APPENDICES

1 Glossary of relevant terms
2 Request for proposal (RFP) [abridged form]
3 Disinfection
4 Skimmer boxes
5 Algae Control

Those with more detailed questions may contact the author at: juan.sabalones@marylandzoo.org

INTRODUCTION

Aquatic systems in zoos include:

1. Aquariums.
2. Aquatic exhibits which feature aquatic animals (otters, polar bears, crocodilians, etc.). Fish may or may not be displayed.
3. Water features i.e. ponds, lakes, waterfalls and streams (natural and man made) which may or may not feature deliberately introduced exhibit animals.
4. Interactive experiences such as water rides and children's "splash zones". Swimming pool regulations may apply.
5. Various combinations of the aforementioned systems.

Otter exhibits generally fall into the second category. Anyone planning to build a new otter exhibit or to renovate an existing one will have a variety of issues to consider. Each institution will have varying circumstances affecting how they approach the project. Nevertheless, there are number of basic steps that can be followed regardless of the situation. While it is beyond the scope of this chapter to go into any great detail, we have included some appendices to expand on some of the more salient issues.

PLANNING AND DESIGN

Typically, a planning and design team will be formed for the initial phase of the project. In addition to management and exhibit designers, the expertise of the following parties is needed to achieve the best results:

- Operations
  The husbandry, maintenance, engineering and exhibits, etc. staff have dealt with the idiosyncrasies of exhibiting particular flora and fauna. At this initial stage, the designers can use their input to help make the system more practical and ergonomic. Once the project gets past a certain point in the construction phase, changes becomes very difficult if not, for all practical purposes, impossible. In addition, because each aquatic system is a custom installation, problem solving is much easier if the operating staff has a good understanding of the design and construction phase of their system.
- Project Manager
  Someone should serve as liaison and coordinator between the staff, the contractors and the part of the institution that approves the financial expenditures. Experience in designing, building and operating similar exhibits can be very useful. Experience with both roles, i.e. in managing contractors and in being a contractor, is also highly desirable. A project manager should strike a balance between what is desired and what is possible within the constraints of the overall master

plan/budget and then bring the balance to all relevant parties and work with them to implement it, on time and on budget.

Once the team does its due diligence, if the decision is to hire outside contractors, it is best to have its plans as much in order as possible prior to engaging one. This saves on expensive consulting time and also helps focus the institution on the task at hand. Regardless of the level of expertise of the contractors hired, an institution should maintain this involvement and focus as much as possible throughout the project. There is a cliché that you get what you pay for, but the reality is more that many projects could have been completed for much less had the planning and design team been more efficient and effective.

CONTRACTORS

If they have the wherewithal, they may choose to handle all aspects of such a project in-house, but most institutions will have to hire contractors to handle major portions of the job. For new exhibit construction or renovation, each institution will have its own procedures for hiring contractors. These can generally be categorized as follows:

A. Hiring a life support designer and a separate installer and managing the operation in- house.
B. Hiring a design firm (many institutions already have relationships with one) who will then subcontract a life support designer and a separate installer. The institution will handle any other related work.
C. Hiring a firm that will handle all of the aspects of building or renovating an entire exhibit (design and construction of the exhibit, its décor, life support system and all associated aspects) in what is often referred to as a “turn key” operation.
D. Some combination of these procedures.

Each institution will have to weigh the positives and negatives of each approach. In terms of what to look for in designers and installers, building or renovating an exhibit in a zoo or aquarium represents a unique challenge for most contractors, and adapting to that environment takes time and money. The ideal contractor will be able to adjust to the particular institution’s environment quickly and successfully:

- Life Support System/Filtration System Designer Traits
    - Balance: A competent designer will be able to come up with a design that balances the needs of the staff with the temporal and budget parameters set by the institution. A bad designer can set you back significantly.
    - Practicality: Some designers have the technical capabilities but lack the practical sense to translate them into a workable, affordable design in a zoo or aquarium setting. Closely scrutinize the consultant’s CV particularly when it comes to previous projects cited. Current operators of those projects should be interviewed. Bear in mind other institutions may be reluctant to be candid about a contractor’s shortcomings.
    - Experience: The best designers have some operating and maintenance experience with the types of systems they are designing and can therefore relate well with the operating staff.
- Installer Traits
    - Expertise: A good installer will be able to take the designer’s plans and build a system that works as designed. This is harder than it sounds.
    - Ergonomics: Besides the normal technical competencies one would expect, it is important that they demonstrate a good understanding of operational ergonomics (see Appendix E) as it relates to such systems. The easier it is for staff to access and operate machinery and controls, the more likely they are to properly operate and maintain them. Ideally you should inspect their most recent installation.
    - Experience: Installing an aquatic system in a zoo or aquarium is often an unorthodox project even for the experienced contractor. Hiring the local swimming pool installer may be cheaper, until the time spent by the designer, project manager and operating staff guiding them through the project and correcting mistakes is added up.

WATER QUALITY MEASUREMENT

“If it cannot be measured, it cannot be managed “

Nolan Karras, Speaker of the House, Utah House of Representatives

The key to properly managing any aquatic system is to have the best handle possible on the water quality. A good water quality lab is staffed by people with the experience to interpret the data. Along with good record keeping, this is an invaluable aid in problem solving. Here is a suggested equipment list:

- Well Equipped (>$50,000)
    - Liquid Spectrophotometer
    - Ion Chromatograph
    - Travel liquid spectrophotometer
    - Data Sonde
    - Dissolved Oxygen Meter
    - Turbidity Meter
    - Flame Spectrophotometer
    - Temperature compensated pH Probe
    - Refractometer
    - Micro scale
    - Small scale
    - Large scale
    - Micro Pipette
    - Autoclave
    - Large Spinner Plate
    - Small Spinner Plate
    - Testing Kits (CO_2, Alkalinity, Hardness)
    - Microbiology Incubator
    - Fume hood
    - Cabinetry / Island
    - Miscellaneous testing materials
    - Miscellaneous safety equipment
- Moderately Equipped ($20,000-$49,999)
    - Liquid spectrophotometer
    - Colorimeter
    - DO meter
    - Temperature compensated pH probe
    - Refractometer
    - Small Scale
    - Large Scale
    - Small Spinner plate
    - Testing kits
    - Fume Hood
    - Cabinet / storage closet
    - Misc. Testing material / Tools
    - Safety Equipment
- Minimally Equipped (<$20,000)
    - Colorimeter or Drip test kits
    - DO meter
    - Temperature compensated pH probe
    - Refractometer
    - Small spinner plate
    - Test kits
    - Safety equipment
    - Testing materials

Some institutions may only have one or two exhibits to deal with and a very limited water quality testing budget. Other institutions may wish to explore a relationship with entities such the local municipal water treatment plant or the local public aquarium especially when it comes to more involved testing such as coliforms.

REQUEST FOR PROPOSALS

After the planning and design team has done their due diligence and developed a plan, they are then ready to issue a request for proposal (RFP).

We have included sections from an actual Request for Proposal (RFP) for the design of a life support system for an Otter Exhibit (see Appendix R). Due to its length, it has been abridged but enough remains to give you an idea of what is entailed. Other institutions have their own basic version of such a document so they will have to modify the pro forma language to suit their requirements. A performance specification for the exhibit has been included. The performance of a life support system is measured by its water quality and water clarity. Those parameters can and should be quantified as they give the client a point of reference with the contractor as to whether the design has met expectations.

This RFP does not address the issue of temperature control because this particular exhibit did not require it. Otters in general seem to be tolerant of a wide range of temperatures depending on the species (see Chapter 1.1). Nevertheless, each institution should determine as early as possible if water temperature is a concern. If there is any doubt, it should be included in the performance specifications as addressing it after the fact is usually very cost prohibitive.

-Juan Sabalones, Instution S

Part 1: GLOSSARY OF RELEVANT TERMS

Aquatic Life Support Systems: There are two types of aquatic life support systems--open and closed. Open systems are only possible in certain locations. The primary advantage is (usually) lower cost for construction and, potentially, ongoing operations. The primary disadvantage is less control over exhibit water quality due to the inability to control the quality of the primary water source.

<u>Open:</u> An open or flow through system takes water from a source outside the exhibit such as a river, lake, stream or even the ocean, and runs it through the exhibit at such a rate as to reduce the need for filtration and then returns the water to that source. Such systems are not possible for all locations and are far from ideal. The question of how much an open system costs verses a closed one, for example, is difficult to answer. It depends upon how open the system is and what the water quality is adjacent to the intake. If the otter exhibit is located where an open system is a viable option (i.e. all the proper parameters can be met) then the primary considerations, especially in regards to salt water, involve a need for some pretreatment (usually some form of sterilization and mechanical filtration). Depending on the regulations in your local jurisdiction, there may be a need to treat the water prior to returning it to the source. Zoos have long practiced a variation on this known as “dump and fill” where the water leaving the exhibit goes straight to a drain.

<u>Closed:</u> A closed system, ideally, recycles the water so that water changes are performed only because of evaporation and should keep all the required water quality parameters at appropriate levels. In reality, even the best-designed and best-operated closed systems can only increase the amount of time between water changes. Nevertheless, in many locations they may be the only option.

Closed systems are generally more expensive to build than an open counterpart. This would include capabilities such as raw make-up (for salt water systems) and storage, proper mechanical, biological and chemical filtration, denitrification, aeration, organics control, and backwash water recovery and especially, a top-notch water quality laboratory.

Water Parameters: The United States Department of Agriculture has certain coliform counts that need to be performed for exhibits that fall under the marine mammal category. Otters do not fall under that category and therefore water tests are not required by any federal regulatory agency. Nevertheless, it is prudent to perform at least weekly water quality tests for bacterial counts and daily tests of chemical additive levels. Records should be maintained and available for inspection and reference if problems arise.

<u>Coliform bacteria</u>*:* Coliform bacterial counts are used to monitor filtration system efficiency and keep track of potentially harmful bacteria. Coliform counts should be done at least every other week and more often if there are multiple animals using the pool (a policy regarding coliform testing should be set by the institution). Often a MPN (Most Probable Number) per 100ml is given as an acceptable limit. However, a more accurate measure is the total or fecal coliform count (NOAA 2006). There are no standards established for fresh-water otter pools at this writing. At this time, it is suggested that coliform levels be maintained at or lower than levels established for rescued pinnipeds by NOAA. These are:

- Total coliform counts should not exceed 500 per ml water, or a MPN of 1000 coliform bacteria per 100ml water.
- Fecal coliform count should not exceed 400 per ml. If animal caretakers are routinely exposed to pool water, an institution may establish a higher standard of 100 per ml, which is the level considered safe for humans; this should be based on institutional policy.

Temperature: More detailed research is required into optimal water levels for the tropical otter species; however, at this time the AZA Otter SSP recommends the following temperature guidelines:

A. cinereus: The water temperature for *A. cinereus* should be maintained between 18.3-29.4°C (65-85°F), preferably at the warmer end of this scale (Petrini 1998).

A. capensis, L. canadensis: The water temperature for *A. capensis* and *L. canadensis* does not appear to be critical.

L. maculicollis: Water temperature in successful *L. maculicollis* exhibits has ranged from 8.9-15.6°C (48-60°F). Temperatures in the 15.6-21.1°C (60-70°F) range may encourage this species to spend more time in the water, however, this is has not been objectively demonstrated at this time.

P. brasiliensis: Further study into optimal pool temperatures and water temperature exposure recommendations for *P. brasiliensis* is required. Sykes-Gatz (2005) recommends this species should not be allowed to swim in unheated water when air temperatures are below 5°C (41°F). Sufficient indoor swim areas are needed when seasonal daytime air temperature regularly falls below 15°C (59°F), regardless of whether outdoor water is heated. This is particularly true for family groups rearing pups that may be held indoors for 4-5 months during cold temperatures. Heating of indoor housing pools is not necessary if the ambient air temperature is maintained at recommended levels.

Each individual institution will have to determine whether their exhibit must be heated or chilled and the most efficient way to do that.

Turnover Rate: The size and capacity of a life support system is based on turnover rate (the given unit of time it takes the total amount of the exhibit water to pass through the filtration). This is usually given as gallons per minute (GPM).

The turnover rate is based on the volume of the exhibit combined with the amount of organic matter likely to be put into the water by the animals and the environment (leaves and branches, etc.). For example, a hippopotamus pool is likely to require a much higher turnover rate than an otter in the exact same sized pool. The dirtier the water is, the likelier a higher turnover rate should be employed.

A proper turnover rate is crucial as a system with a turnover rate that is too low will always be laboring to catch up, and a system with a turnover rate that is too high is wasting money.

Filtration:

Biological Filtration: This is the use of bacterial colonies to convert ammonia to nitrite and then nitrate through nitrification.

In aquariums with fish, ammonia and nitrite are harmful and should be kept as close to 0.0 as possible. With the exception of certain invertebrates in salt water systems, nitrates are not considered harmful. Otters should have no problem with ammonia and its derivatives, but an excess of nitrates in an otter pool is undesirable as they are a primary source of nutrients for algae.

Nitrate levels can be kept in check with water changes or by a number of different denitrification methods.

Mechanical Filtration: Mechanical filtration is the mechanical removal of particulate matter suspended in the water column before it decomposes.

The matter is moved to the filter which is then cleansed. Until the filter is backwashed, the particulate matter is still in the system. Rapid sand, screen, drum, bag, diatomaceous earth, protein skimmers and bead filters are most commonly used in this application. Water clarity is most directly affected by the quality of the mechanical filtration.

Chemical Filtration: The use of any filtering substance designed to chemically attract pollutants in order to remove them from the water column.

- Activated Carbon

- Reverse Osmosis is a method of filtering water by pressing the water against a semi-permeable membrane that sits inside filter housing. This membrane allows water molecules to pass through, but not others. Minerals, trace and other elements are removed producing pure raw water.
- Ion Exchange resins are a water filtration medium comprised of natural and synthetic resins in a flow-through pouch. They selectively remove ammonia, nitrite, and nitrate from freshwater.

Part Two: REQUEST FOR PROPOSALS (RFP) [ABRIDGED VERSION]

OTTER EXHIBIT LIFE SUPPORT SYSTEM RENOVATION.

The following specifications shall define the scope of this design/build project for installing a Life Support System's (LSS) mentioned above at the Generic Zoo.

Bidders:

You are hereby invited to submit a proposal to the Generic Zoological Society, Inc. (the "GZS") for the design and construction of:

OTTER EXHIBIT LIFE SUPPORT SYSTEM RENOVATION.

I **Bid Documents**

A. RFP Project Description
B. Project Scope of Work and Specifications attached.
C. Bid Form (attached to this RFP)
D. Contractor's Qualification Form (attached to this RFP)
E. Generic Zoo, Zoo Contractors Policy
F. General Condition form
G. Standard AIA Contract
H. Prevailing Wage Rates

PROJECT DESCRIPTION

Project: Otter Exhibit Life Support System Renovation
The Otter Exhibit Life Support System Renovation project consists of designing and installing a replacement Life Support System for the existing Otter Exhibit as described in the following specifications.

Contractor Responsibilities:
1. Where specified standards are in conflict, the more stringent of the two shall apply.
2. Site verification of specifications during the bid process and post award period.
3. Visit the site to determine the extent of work, which may or may not be shown in the plans.
4. Any Owner property damaged as a result of the work associated with this project shall be restored to its original condition at Contractor's expense. Contractor is responsible for documenting existing conditions.
5. Coordinate the marking of any underground utility lines, which are within the proposed limits of construction.
6. The Site shall be kept clean at all times. Contractor shall keep the project site clean from worker-related debris, including surrounding lay-down areas, parking areas, walkways and lawn areas. All debris shall be constantly picked up and properly disposed to reduce impact on guest experience and so that winds do not carry the debris to other areas of the grounds.
7. The Zoo has adopted a limited smoking policy. All vendors may only smoke in areas permitted by the designated Zoo representatives, and the contractor is responsible for the workmen, including those of sub-contractors, disposing of cigarette butts in identified containers.

Bid Documents:
1. Request for Proposal
2. Project Description
3. Contractor's Qualification Form
4. Bid Form
5. Project Scope of Work and Specifications

6. Generic Zoo Contractors Policy
7. General Condition form
8. Standard AIA Contract

CONTRACTOR'S QUALIFICATION FORM

The undersigned certifies under oath the truth and correctness of all statements and all answers to questions made hereinafter.

QUALIFICATION OF LIFE SUPPORT SYSTEM CONTRACTOR

1. In order to be the Life Support System Contractor on this project, bidders are to submit the following information to the Zoo not less than ten (10) days prior to the bid opening date.
2. Experience
3. Statement certifying that the Contractor has been in business a minimum of Six (6) years and has extensive experience in the construction of mechanical life support systems for live animal exhibits of similar size and scope.
4. Contractor must have experience designing and building similar LSS.
5. References of at least one (1) recent project at an AZA Accredited Zoological Facility within the last year, demonstrating experience and ability to install projects of similar size and complexity as those described in the RFP. Include the name of the person responsible for the project, phone number, and approximate contract amount.
6. Contractor shall submit full documentation of his/her construction crews and lead personnel, detailing the experience of each person listed and their ability to perform all phases of the work to the Zoo's satisfaction.
7. Site Superintendent must have supervised ten (10) or more LSS installations during his/her employment with the Contractor
8. Site Superintendent must have experience in installing ozone systems
9. Site Superintendent must show proof of 30-hour "OSHA 500" safety certification course.
10. Contractor shall be a member of the Association of Zoos and Aquariums.

The Zoo reserves the right to require additional information and/or request a visit to completed work to make a determination of the bidder's qualifications to produce work as described in the Construction Drawings and Specifications.

Project Identification: Life Support Systems Installation

Name	Title
Company Name	Phone
Address	City/State/Zip
Fax	FEIN
Signature	DATE

(If more space is needed, please answer on the back of this sheet)

Is your organization licensed to do business in the State of……..?

Have you ever failed to complete any work awarded to you? If so, note when, where, and why? (If more space is needed, please answer on the back of this sheet.)

Please list all work which your firm will be self performing.

List Subcontractors. Use back of sheet if necessary.

List four or more projects executed by your firm within the past three years that were similar in nature and scope to this project. Additional projects may be listed on a separate sheet.

A) Project Name: Location: Year:

Project Cost: Owner Name and Phone Number:

B) Project Name: Location: Year:

Project Cost: Owner Name and Phone Number:

C) Project Name: Location: Year:

Project Cost: Owner Name and Phone Number:

D) Project Name: Location: Year:

Project Cost: Owner Name and Phone Number:

If you have State of….. and/or….. MBE/WBE certification for you or any subcontractors, provide Certification Numbers, Expiration Dates, and Disciplines/SAIC numbers for which you or they are certified.

Provide names of key personnel to be employed on this project. Indicate the projects listed above with which they were involved.

Name	Years Experience	Years w/ Firm	Projects listed	Project Role
1.				
2.				
3.				
4.				

If you wish, attach photographic documentation of projects listed in above that illustrate work that you have completed that is most comparable in style, technique and workmanship to the project.

BID FORM

Generic Zoological Society, Inc.

Life Support Systems Installation Request For Proposals

Deliver To: Construction Department
Generic Zoo
Smallville, USA, Planet Earth

In submitting this bid, the Undersigned declares that they are the only person, or persons, interested in said bid, that it is made without any connection with any person making another bid for the same contract, that the bid is in all respects fair and without collusion, fraud or mental reservation, and that no employee of the OWNER is directly or indirectly interested in said bid, or in the supplies or work in which it relates, or in any portion of the profits thereof.

The Undersigned also declares that they have examined the Request For Proposals, including the drawings and specifications contained therein, and that by signing this proposal, they waive all right to plead a misunderstanding regarding the same.

The Undersigned agrees to submit a bid bond, payable to the Generic Zoological Society, Inc., in the amount of five percent (5%) of the total bid amount, along with the bid.

The Undersigned further understands and agrees that they are to furnish all material, equipment and supervision to complete entire work for the indicated project and to accept in full compensation therefore the stipulated sum or sums as stated herein.

On acceptance of the proposal for the construction portion of said work, the Undersigned does hereby agree to provide the Generic Zoological Society within ten (10) days Payment and Performance bonds in the amount of one hundred percent (100%) of the total bid price to provide construction services for the consideration named herein.

The Undersigned agrees to hold open this Bid Proposal for a period of ninety (90) days following the submission of this Bid Proposal.

The Undersigned agrees to provide evidence of insurance coverage along with their bid submission, including areas and amounts such as umbrella insurance, general liability, automobile liability, garage liability, excess liability, workers' compensations and employers' liability. On acceptance of this proposal for said work, the Undersigned agrees to provide the Generic Zoological Society, Inc. with a Certificate of Insurance adding the Generic Zoological Society, Inc. as an Additional Insured.

The base bid price of the proposal shall be inclusive of the following:

- Price to provide the design of the LSS
- Price to complete the installation of all LSS piping, equipment, accessories, valves in accordance with these specifications.
- A one-year warranty from substantial completion on all parts, labor and material.
- Performance and payment bonds on the construction phase.
- Factory representative onsite for initial start-up of all systems.
- All labor at Davis-Bacon Act Prevailing Wage Rate.
- Detailed listing of all equipment, labor and materials.
- Guarantee of performance of LSS to meet criteria.
- LSS shall be designed and built to maintain the exhibit animals in a healthy condition (see 1.04B).
- LSS shall be designed and built to maintain high water clarity suitable for overhead viewing.

<u>OTTER EXHIBIT LIFE SUPPORT SYSTEM RENOVATION</u>

A. PROJECT SCOPE/SPECIFICATIONS

1. Contractor
    a. Design and Install a new Life Support System (LSS) for the Otter Exhibit at the Generic Zoo. This scope of work shall be split into two contracts. The first phase shall cover the design of the LSS. The deliverables for the first phase shall include completed designs to meet the project budget and a proposal to provide the

construction of the LSS. The second phase shall include the construction of the LSS.

b. Remove all existing LSS equipment and piping. Please provide an alternate to perform this work.

c. Piping will connect to existing main drains, surface skimmer, returns, waterfall, overflow, waste and domestic water lines.

d. 8 hours LSS Startup and Zoo staff operator training.

2. Zoo

a. Exhibits will be emptied of all water and animals.

b. The Zoo is responsible for the integrity and functioning of existing underground and through-wall piping and electrical service.

c. Provide all electrical connections from existing electrical panels. (See Alternates)

B. BASE EXHIBIT DATA

Otter Exhibit

a. 20,000-gallon fresh water pool.

b. Exhibit pool is outdoors.

c. Fresh water provided by existing domestic water supply.

d. Exhibit maximum capacity to be two river otters.

e. Exhibit filtration consists of sand filtration with manual backwash operation.

f. Exhibit disinfection currently done by bromine additions.

C. LSS DESIGN PARAMETERS

1. The LSS equipment will be sized to meet or exceed the following minimum requirements. If the minimum requirements appear insufficient, provide an explanation for concerns on the sizing. Any additions, deletions, or substitutions must be approved by the designated representative of the Zoo.

a. Turnover rate for the Otter system shall be at least 60 minutes.

b. System shall have flow meters appropriately placed to monitor the turnover rate.

c. Waterfalls in the exhibits shall be on a separate loop (with a dedicated pump) from the LSS.

d. Both systems shall have skimmers adequately sized and located to deal with the heavy leaf litter problem. Skimmer openings shall be screened to protect against accidental intrusion by live exhibit tenants.

e. If sand filters are specified, they will be sized to less than or equal to 10 gpm/sqft of filter area at 100% of system flow. Backwash shall be manual or hydraulic in operation.

f. Exhibit disinfection shall be provided by an automated ozone injection treatment system.

g. Ozone system will be sized for a 20% side-stream and a contact time of 2-3 minutes. It shall include an ozone destruct unit. There shall be no residual ozone in the exhibit.

h. There shall be a trickle filter sized for the full system flow at 15 gpm / sq. ft (of tower cross section). The tower shall have enough height to provide adequate gas exchange and prevent air entrainment in the exhibit.

i. All domestic freshwater supply lines shall have flow meters.

j. All equipment shall be able to be disconnected with unions or flanges from the system.

k. All equipment will be fully bypassable.

l. Valved water sampling ports will be provided in the LSS equipment area.

m. Two primary filtration pumps that will each have the capacity to run the LSS at design capacity will be installed so that a full backup pump is accessible with the turn of two valves. Piping on the suction side of the filter pumps shall be sized to handle the designed system flow at less than 4 ft/second. Piping on the discharge side of the filter pumps shall be sized to handle the designed system flow at less than 6 ft/second. Gravity piping will be sized to accommodate the design flow at less than 2.5 ft/sec.

n. Contractor to verify new equipment will fit in the area of the existing pump room obtained by removing existing equipment.

o. Contractor to verify prior to bid that existing piping to remain is appropriately sized for new LSS.

p. Contractor to install all systems according to local code and arrange for all inspections.

2. The LSS contractor will be required to guarantee the performance of the LSS once installed. If in the opinion of the bidding contractor, the above criteria will not meet the quality standards set forth here or meet the LSS contractor's own standards, those deficiencies should be detailed in the proposal. The design must allow the system to achieve the following parameters under normal operating conditions :

Parameters	
Temperature	(appropriate to species specific range)
Calcium Hardness	100- 200
ORP	300-400 pool, 750 - 800 contactor
Ozone in Water	0 ppm
Total Alkalinity	80-120 ppm
Dissolved Oxygen	90-100% saturation
Free ammonia	0
Nitrite (NO_2)	<0.1 ppm
pH	7.5 - 7.8
Coliforms (MPN)	< 1000

D. EQUIPMENT AND MATERIALS (ANY ADDITIONS, DELETIONS OR SUBSTITUTIONS MUST BE APPROVED BY THE DESIGNATED REPRESENTATIVE OF THE ZOO)

E. EXECUTION

1. Inspection

a. Examination: The contractor shall examine surfaces for conditions that will adversely affect execution, permanence and quality of work.

b. Unsatisfactory Conditions: The contractor shall correct unsatisfactory conditions before proceeding with the work.

c. Operating Instruction: Upon completion of work and acceptance of operation and maintenance manuals, the contractor shall provide bound operational instruction books to the owner.

d. Instructions shall include the operation of the treatment system for a period of 8 hours, at a time designated by the owner. Upon completion of such instruction, the contractor shall obtain from the owner a dated and signed statement certifying the completion of such instruction.

e. Project Close out requirements:

1) The contractor shall provide the following items as prerequisite to the issuance of certificates for final payment and formal acceptance of the project:

a) As Built Drawings
b) Reproducible Record Drawings.
c) Valve Identification Chart.
d) Maintenance and Operating Manuals.
e) Operating Instructions and Certified Statement.
f) Certified Statement of Successful Test.

2. Installation

a. Pipe and Equipment Installation

1) The contractor shall install all piping, valves, and equipment in a manner and in locations to avoid obstructions and keep openings clear. Pipe and/or equipment shall not be installed where it will present a potential tripping hazard or below 7'-0" above finished floor, where it would be a potential head knocking hazard. Installation shall permit direct access to all valves and pieces of equipment that will require maintenance. The contractor shall make any changes as directed by the owner, at no additional expense, which may be necessary in order to accomplish this purpose.
2) Before being placed in position, all pipe, pipe fittings and accessories shall be cleaned, and shall be maintained in a clean condition. Piping shall be installed and aligned in accordance with the Drawings with a tolerance of + 1/8-inch in the horizontal and vertical directions.
3) All work specified and not clearly defined by the Drawings shall be installed and arranged as directed and in a manner satisfactory to the owner.

b. Installation of Pressure Filter Gravel and Sand
   1) Preparation of Filters: Before placing the media, the contractor shall determine that all holes in the underdrain system are open.
   2) Placing of Gravel: The gravel shall be deposited in such a manner as to avoid endangering the header laterals. Each layer shall be brought up to the required elevation and made level over the entire filter bed area, and shall be smoothed down to a true surface. Care shall be taken not to injure equipment piping and coatings in the filter units by walking on or dropping gravel upon them.
   3) Placing of Sand: In placing of the sand in the filters, extreme care shall be taken to avoid disturbing the internal piping. Extreme care shall be taken to protect the internal filter coating. Sand shall be placed to the appropriate depth specified and shall be level.

c. Installation of Piping
   1) All PVC Pipe will be transported, stored and installed with regard to manufacturer's recommendations. Bolting of PVC flanges shall be in accordance with manufacturer's recommendations and shall not be unduly stressed through the use of excessive torque while tightening bolts. Use of torque wrench will be required.
   2) All life support piping shall be flushed clean prior to connection to equipment or tanks.

d. Installation of Pumps and Motors
   1) Pumps and motors shall be installed in accordance with the Manufacturer's Recommendations.
   2) Pumps shall be installed with an isolation valve on the suction side of the pump. This valve shall be a true-union ball valve unless it is 4" or greater, in which case it will be a gate valve. A compound gauge shall be installed between the pump and the isolation valve.
   3) Pump shall be installed with an isolation valve on the discharge side of the pump. This valve shall be a butterfly valve. Between the pump and the isolation valve, a swing check valve is required. A pressure gauge shall be installed between the check valve and the isolation valve.
   4) Unless otherwise noted, all pumps shall be installed on concrete housekeeping pads.

e. System Startup
   1) The contractor shall provide one full-time mechanical technician who is familiar with the system for a period of 3 days to aid the owner in starting and operating the system.

PART 3: DISINFECTION

Bromine: Bromine is unsuitable for outdoor usage as it is broken down very quickly by sunlight

Ozone-Chlorine comparison: The common perception is that ozone systems are more elaborate and expensive than chlorine. In truth, both systems will require that water parameters be measured. If the operator starts with more or less a blank slate, the level of sophistication required to safely and effectively operate and maintain a properly designed chlorine disinfection system vs. the level of sophistication required to safely and effectively operate a properly designed ozone disinfection system may just about be equal.

Indeed, one could argue that the chlorine system operator needs a higher level of sophistication and dedication and needs to work harder and be more attentive than if they had an ozone system. As we are talking about otters, when comparing an ozone system to a chlorine system, the latter system is more likely to harm the animals (or their human caretakers).

Ozone: If you have a well designed, installed and maintained system, the operator will keep the various filters and probes clean. They might occasionally turn a few dials but other than that, operation consists primarily of monitoring the system and recording data. Every year or so you should get a visit from an ozone tech for the more serious maintenance issues depending on your system, but that is about it.

Advances in design now allow us to use much smaller dosages of ozone than in the past with the resulting decrease in safety concerns.

The oxidation process with ozone occurs in the contact chamber away from the animals, and any residual ozone is unlikely to be anywhere near as harmful, if at all, to the otters as chlorine residuals. The by-products of ozonation can be dealt with.

Chlorine: In the United States, many municipalities add chlorine to their water, and readings from tap water of 1ppm or higher are possible. For this reason, many aquariums run their city supplied water through carbon filters prior to use. While otters generally show no adverse effects from these levels, it is not known what the overall impact is to their health and the water repellency of their coats. For this reason, the AZA Otter SSP recommends that otters should not be exposed to chlorine levels higher than 0.5ppm for prolonged periods, and ideally, chlorine should be kept at a non-detectable level. The addition of sodium thiosulfate will neutralize any residual chlorine. The most efficient and effective way to do this is to use an automated system (www.polarispool.com/products/details.asp?ID=34).

This would cost you about $5000.00 to buy and install. Because chlorine is most effective between the pH of 7.2-7.8, such automated systems draw from sodium hypochlorite barrels and muriatic acid barrels. Without an automated system, the pool will have to be tested daily and chemical adjustments made manually. Even with the automatic controller, this system is inherently more labor intensive than an ozone system.

The oxidation process with chlorine occurs in the water with the animals that are exposed directly to the chemicals. Therefore an overdose of chlorine or muriatic acid would affect the animals. Increasingly, evidence is mounting that chlorinated pools may cause health problems in humans (www.swimming.about.com/od/allergyandasthma/a/cl_pool_problem_3).

Therefore all things being equal, one has to wonder how sensible and ethical it is to continue using chlorine with otters. Chlorination produces carcinogenic byproducts know as Trihalomethanes (THM) (www.epa.gov/enviro/html/icr/gloss_dbp). Sooner or later, this may become a regulatory issue and we have no way of removing THM from the process.

In the end, the decision on the type of disinfection to use is an equation. Your particular institution may have a set of circumstances (access to highly trained personnel and sophisticated water quality measuring equipment, inexpensive sources of sodium hypochlorite and muriatic acid, lots of cheap labor, etc.) that will sway the equation towards chlorine. And in cases where algae is out of control, chlorine may be the most immediate and cost effective solution.

However, all things being equal, our experience has been that when you add up the total cost of doing it correctly over a number of years, having an ozone disinfection system works out to be cheaper, less dangerous, and far less labor intensive. Time is money.

Basic Manual Chlorination Device: In manually applied situations, the two most common forms of chlorine used are granular and liquid.

Granular: Granulated chlorine is designed to dissolve slowly over time so it needs someplace (typically a skimmer or a floating chlorinator) where it can dissolve away from direct human or animal contact.

DERBY DUCK FLOATING CHLORINATOR

Large capacity holds up to six 3” tabs (or 1” tabs or sticks). Adjustable to regulate the amount of chlorine dispensed. Made of heavy duty resin – child resistant lock.

Liquid: In its liquid form (sodium hypochlorite) the chlorine is more concentrated. It would be used if the need is to raise the levels of chlorine quickly. Sodium hypochlorite is very corrosive and proper procedures (and suitable personal protection) need to be taken when using it.

Gas: Chlorine gas is also commercially available, but this is typically for industrial applications and should be considered too involved and dangerous for most institutions.

Automated Chlorination Device: The Polaris Watermatic Pro System L-1 and L-2SC are complete systems for feeding sodium hypochlorite (liquid) and muriatic acid (liquid). Easy to install, operate and maintain, these systems include controllers that plug into any 120V outlet and provide built-in receptacles for the peristaltic pumps. The L-2SC System further provides easy-to-use graphic displays and adjustable-rate pumps.

Benefits:

- Complete liquid system
- Integrated Flow Cell
- Easy to install and maintain

Drawbacks:
- Need to buy, handle and store toxic chemicals such as sodium hypochlorite and muriatic acid.
- Exposes animals and workers directly to potentially caustic substances.

Chlorine Removal: Chlorine can be used, if necessary, when the otters are not present. Chlorine is a volatile chemical and sufficient aeration can lower levels dramatically in twenty four hours. In situations where time is an issue, sodium thiosulfate can be added and run through the system for an hour before the otters are allowed access again. To remove chlorine with sodium thiosulfate:

Amount Sodium Thiosulfate (grams) = aquarium volume (gal) x 0.0038 x [7 x tested chlorine level (mg/l)]

Example:
Problem: 20,000 gallon tank. Chlorine test results show 0.5mg/l of chlorine in the system following water change and refill. To remove the chlorine you will need ____ grams of sodium thiosulfate.

Solution: 20,000 x 0.0038 x (7 x 0.5) = 266 grams Sodium Thiosulfate

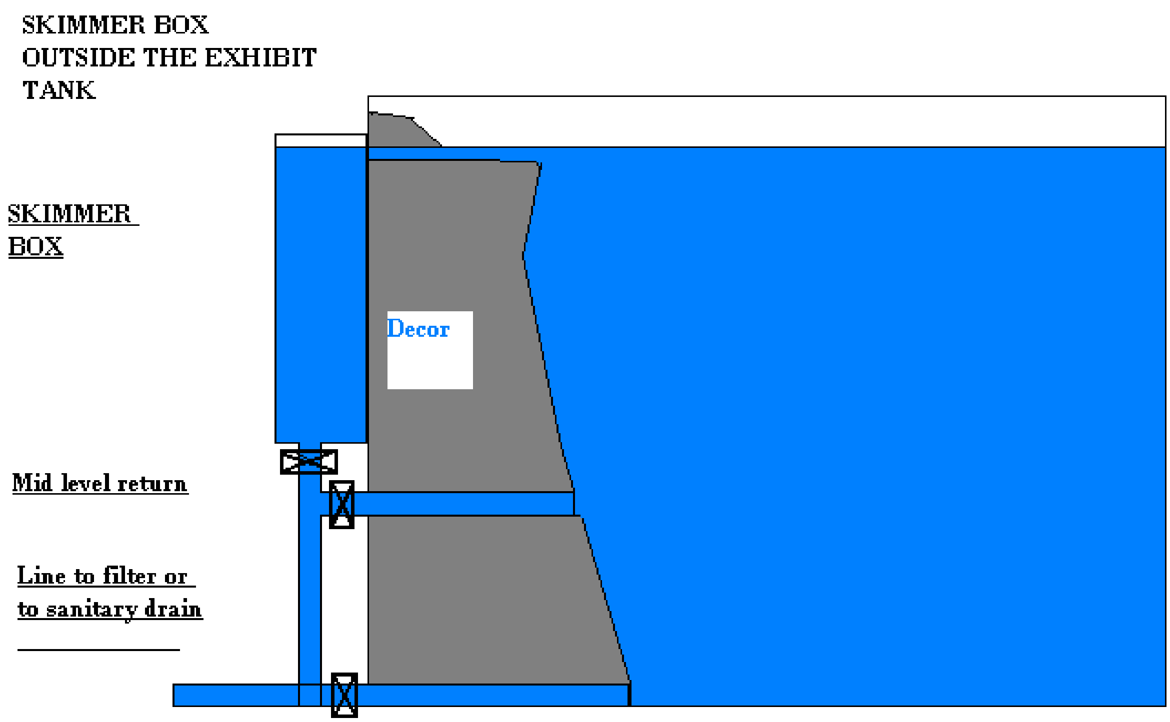
Part 4: SKIMMER BOXES

All skimmer boxes should be protected with appropriate screening to prevent live animals from getting trapped, though this should not be a concern with healthy animals. Screening also prevents foreign objects from harming pumps. A skimmer box can be placed outside the exhibit tank or inside

There are commercially available skimmers typically found in pond and swimming pool supply houses, but they are usually too small for most large otter exhibits so chances are skimmer boxes must be fabricated by the installer. Ideally, the pool should have outflows at three points:

A drain from its lowest point
A midlevel drain
A skimmer box drain

Skimmers should be mandatory, especially in outdoor pools. They aid in the removal of floating organic material (leaves, twigs, etc.) most commonly associated with such pools.



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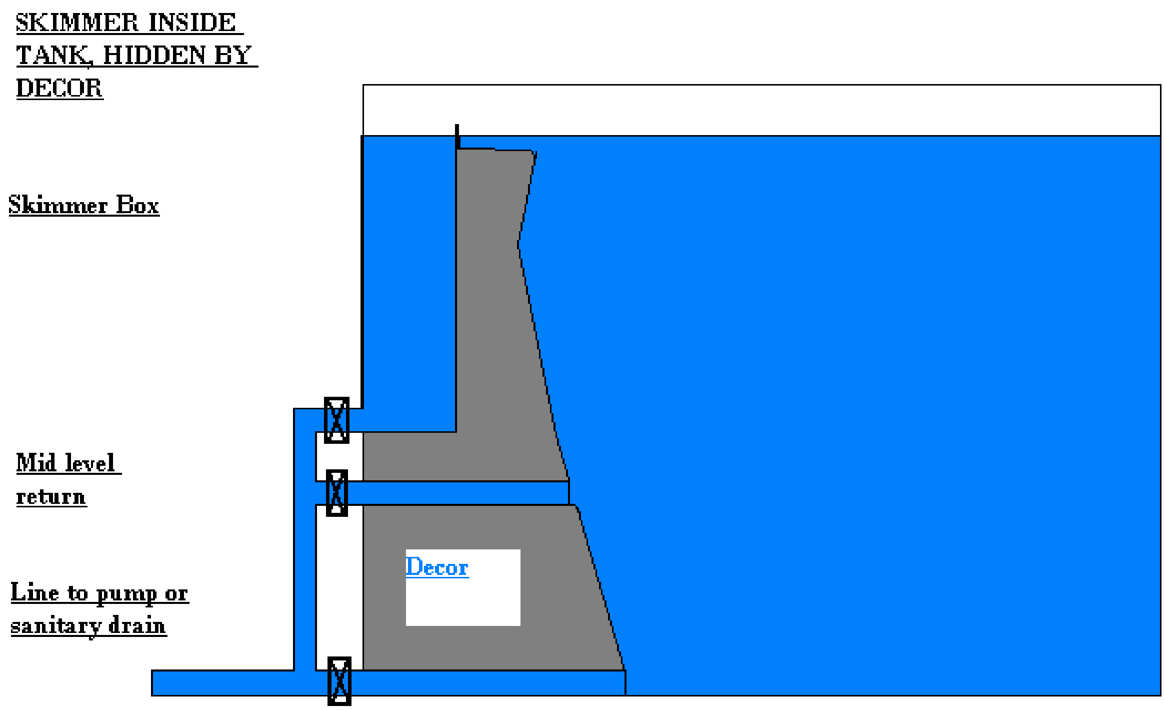
SKIMMER INSIDE TANK, HIDDEN BY DECOR
Skimmer Box
Mid level return
Line to pump or sanitary drain
Decor


SKIMMER SLOT


SKIMMER BOX
SKIMMER LINE
DRAIN LINE


SKIMMER BOX
SKIMMER LINE FLANGE
DRAIN LINE

Part 5: ALGAE CONTROL

The presence of algae in an otter exhibit pool has some benefits. A manageable amount can help control nitrate levels. However, if the level of algae exceeds manageable limits, it makes the water unsightly, reduces oxygen levels, and potentially reduces water circulation.

Algae need light, appropriate temperature, and nutrients. Control is a concern, particularly in outdoor otter pools exposed to significant amounts of sunlight. By providing for a suitable amount of shade and controlling the amount of available nutrients, it is possible to keep algae manageable. Therefore controlling algae is best addressed in the design phase of the exhibit when solutions to these concerns can be built into the exhibit design. Some of them can be added after the fact but it is usually more expensive and troublesome to do so.

Light: If the exhibit is located where the amount of direct sunlight is a problem, a suitable shade structure should be found or fabricated. Coloring agents added to the water can limit the penetration of light to the algae growing below the water's surface by coloring the water. It seems most of them turn the water various shades of blue. Without light, the algae cannot grow. If you have Ultra Violet (UV) disinfection devices on your filtration system, coloring agents will render them useless and the colored water may be an aesthetic issue. They claim to have no effects on animals, but blue tinted otters would be very unsettling.

Trees provide shade but they may also provide falling leaves and branches which foul the water. They also may be toxic in some instances. In such cases, it would be worthwhile to mount some sort of structure, such as a net, to deflect the material from the water.

Nutrients: Algae need nutrients to grow. Nitrates and phosphates are the main concerns. Nitrates come mostly from decaying organic material, found in a variety of sources such as animal waste, uneaten/leftover food, or decaying leaves and branches. Good hygiene and a properly designed filtration system will help. In an outdoor exhibit, a raised lip around the pool will divert rain runoff around the pond and help keep dirt out.

Phosphates usually come in the source water or in food for the animal. At this writing, they can be dealt with by filtering the source water to remove phosphates (expensive) or by treating the system with lanthanum chloride in its various commercial forms. To the best of my knowledge, lanthanum chloride is not harmful to otters but you should check with your vet regardless.

If possible, additional floating plants in the exhibit can compete with the algae for nutrients and also reduce the amount of available light.

Mechanical Filtration and UV Sterilizers: Mechanical filtration moves undesirable organic matter from the water column to the filters. This means the algae have a reduced amount of nutrients available to them. The organic material is typically retained in the filtering material, so keeping the filter regularly backwashed and working efficiently is vital.

UV sterilizers are installed in line after the mechanical filter. The cleaner and clearer water helps UV sterilizers function properly. The clearer the water, the more effective the UV. When water is pumped through the UV sterilizer, the ultraviolet light that is emitted will break down the cell wall of the algae and the algae will then die. UV's also have disinfectant capabilities.

UV dose = UV intensity X exposure time.

Make sure to get the appropriate sized UV sterilizer for your pond, and also make sure that the water flow through the UV allows for sufficient exposure time. Note that the effectiveness of UV drops dramatically as the temperature goes down. Of course, in terms of algae control in outdoor exhibits, algae populations are likely to drop down with the temperature.

Water Additives: Most water additives developed for algae control come from the swimming pool and ornamental pond industry. Many of these products are toxic, not only to algae, but to other forms of life. It is important to take this into account when considering them for use in an otter exhibit. Always check the label and the MSDS sheet to make sure that it is safe for the life in your pool.

Additives can either be algiscidal or algistatic. Algiscidal products kill the algae outright. Most metal ion complexes (copper, silver) fit into this category. Other products, such as barley straw, suppress the growth of algae in some fashion and are usually used in a preventative manner. It is important to understand at what stage the algae problem is so that the correct product is used. For example, barley straw, an algistatic substance, is not as effective after the algae in your exhibit have bloomed. At that stage, it might be best to drain the pool, scrub the algae off the surfaces, refill the pool and then use the algistatic substances to help hold down a recurrence of the problem.

The use of bags of barley straw to filter out algae has been quite publicized. The following links to publications by Carole A. Lembi, Professor of Botany at Purdue University, can provide greater insight into Aquatic Plant Management and the use of barley straw in algae control.

www.ces.purdue.edu/extmedia/WS/WS_21.pdf
www.btny.purdue.edu/pubs/APM/APM-1-W.pdf

References and Further Reading:

Dawes, J. The Pond Owner's Problem Solver. Tetra Press. Blacksburg, Virginia; 1999.
May, PJ. The Perfect Pond Detective Book 1. Kingdom Books. Waterlooville, England; 1998.

Appendix O: Female Otter Reintroduction Plan

This plan was developed to reintroduce two females separated for several weeks. It is offered as a template to follow when introducing females or any unfamiliar animals. Introductions should be planned in advance and based on individual institution policies, physical exhibit design, and individual animals.

- Begin with one holding cage between them and visual access through mesh in holding.
- Facility allowed for each animal to have one side of a separated exhibit. They were still with one holding cage between them at night.
- After a few days, the otters were given continual access to side by side holding dens throughout the day and night.
- Otters were switched between exhibit sides every three days to reduce the chance of creating a territory.
- Historically, otters have been introduced in holding then allowed on exhibit.
- The first day of introductions seems to the have the most signs of aggression.
- Start with short periods of introduction in holding, varying the time between 20 – 60 minutes. The time will vary according to signs of aggression.
- Once the otters appear to be more comfortable with each other progress to introducing them in holding twice a day for a period of 30-60 minutes.
- When both introduction sessions start going well, allow them access to the exhibit.
- Watch for signs of aggression and adjust time together accordingly.
- Each otter should be separated with visual access to one another overnight until confident that there is no chance for injury.

Positive behaviors to look for during the introduction are playful wrestling, muzzle touching, social grooming, face pawing, submissive rolling over, resting together, and friendly vocalizations. These vocalizations are chirping, grunts or chuckling.

Aggressive vocalizations are screaming, snarling, growling or grunting. Signs to look for requiring separation include aggressive chasing, aggressive wrestling, tension while dominance mounting, fighting with a lot of screaming, fighting with injuries, or sign of one trying to drown the other in the pool.

Tools recommended having on hand when beginning introductions:

- 3 brooms
- 4 pairs of gloves
- 2 hoses that are ready
- 2 fire extinguishers
- 3 mammal nets
- 1 air horn
- Tongs
- Snake hook
- Extra fish and treats

By: Jessica Foti
Date: June 4, 2006

Appendix P: Otter Body Condition Matrix

Cheryl Dikeman, NAG Advisor

Matrix is still in development, photos of each body condition are being sought.

SCORE	1 Emaciated	2 Poor	3 Ideal	4 Solid	5 Obese
Photo/Drawing					
General Condition	No obvious fat and loss of muscle mass. Lumbar vertebrae all visible, ribs visible, obvious abdominal tuck. Ilia wings pronounced. Poor coat.	Lean, minimal muscle mass	Optimum body fat and muscle tone, well proportioned, ideal coat condition.	Noticeable fat deposits throughout body.	Obvious fatty deposits, no definition between shoulder, stomach and pelvic regions
Neck and Shoulders	Pronounced scapula and lack of muscle over the shoulders, Noticeable shoulder skeletal region.	Visible scapula with little muscle over the shoulders, thin neck. Visible delineation behind shoulders.	Smooth lines over shoulders and scapula. Slight delineation behind shoulder region.	Smooth lines over shoulders and scapula. No delineation behind shoulder region.	No definition, very thickened neck region. Obvious fat deposits over top of shoulders and in neck region.
Abdomen and Waist	Very pronounced waist and severe abdominal tuck	Visible waist behind the ribs. No visible abdominal fat present.	No visible abdominal tuck. Some distinguishable abdominal fat present; however, not obvious.	Some rounding in the abdominal region. Noticeable abdominal fat. Waist is not visible.	Obvious abdominal fat deposits and large protruding waist region. Abdominal fat pad drops below the rib cage.
Hindquarter	Pronounced and very obvious hip and iliac region.	Pelvic bones visible.	Hips and pelvis slightly visible and palpable but not obvious.	No skeletal visibility in hindquarter. Smooth lines over entire quarter.	Fat deposits obvious over hind limbs. Fat pad obvious on tailhead.
Vertebrae and Rib Cage	All vertebrae visible. Visible ribs.	Tops of lumbar and thoracic vertebrae and ribs slightly visible and definitely palpable.	Smooth lines over topline and throughout body. No visible ribs or vertebrae.	Some fat evident over vertebral bodies and/or ribs	Extreme fat pad over rib cage region. Heavy fat deposits over vertebrae.