ASSOCIATION OF ZOOS AND AQUARIUMS

Standardized Animal Care Guidelines

OTTER

(Lutrinae)

Created by the:

AZA Small Carnivore TAG

In association with the:

AZA Animal Welfare Committee

Standardized Animal Care Guidelines for Otters

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Disclaimer:
The use of information within the Standardized Animal Care Guidelines should be in accordance with all local, state, and national laws and regulations concerning the care of animals in captivity. The Recommendations contained herein are based on the current art and science of animal management, and are provided as best practices that may positively influence the welfare of the animals. The recommendations do not identify exclusive management approaches, diets, medical treatments, or procedures. The Standardized Animal Care Guidelines do not represent specific 'standards' of care. Flexibility in management and care is needed to address the specific needs of individual animals, and potential limitations of individual institutions. However, it is hoped that by identifying best practices and animal care recommendations that these guidelines will help to eliminate those limitations in the future, and maximize the welfare of the animals.

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Standardized Animal Care Guidelines for Otters

Introduction

The AZA Small Carnivore Taxon Advisory Group (TAG) has designated five semi-aquatic otter species for management under the Otter Species Survival Plan (SSP). These are the Asian small-clawed otter (*Aonyx/Amblonyx cinereus*), African (Cape) clawless otter (*Aonyx capensis*), giant otter (*Pteronura brasiliensis*), North American (N.A.) river otter (*Lontra canadensis*), and the spotted-necked otter (*Lutra maculicollis*). This document will address keeping these five otter species in captivity. These otter species exhibit varying needs and degrees of sociality. Many of their captive 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); dusty.lombardi@columbuszoo.org
- International Giant Otter Studbook Husbandry and Management Information and Guidelines 2005, 2nd Edition (Sykes-Gatz 2005); v.gatz@cityweb.de
- North American River Otter Notebook, 2nd Edition (Reed-Smith 2001); jan.smith@columbuszoo.org

Asian small-clawed otter (*Aonyx/Amblonyx cinereus*): The Asian small-clawed otter was originally known as *Aonyx cinerea* then *Amblonyx cinereus*. ISIS uses *Amblonyx* but the IUCN/SSC Otter Specialist Group uses *Aonyx cinereus* based on work by Koepfli and Wayne 2003. 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 captive 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).

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 captivity, 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).

<u>Giant otter</u> (*Pteronura brasiliensis*): This single species in the genus *Pteronura* is one of four species of otter found in South America. Their 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 their 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 captivity. 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 in 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. Threats include habitat destruction and degradation, over-fishing, illegal hunting, mining, and water and land pollution.

<u>North American river otter</u> (*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 captivity (Ben-David et al. 2000). In the wild, males do not participate in pup rearing; in captivity 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 (in prep.) 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. 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).

<u>Spotted-necked otter</u> (*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). 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.

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 1992). Listed on CITES as Appendix II (www.cites.org).

1. Abiotic Environmental Variables (address both exhibit and off-exhibit holding)

1.1 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 the animals the opportunity to select for their comfort.

A. cinereus: The ideal air temperature is between 22.2-24.4°C (72-76˚F). If Asian small-clawed otter 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.

A. capensis and *L. canadensis*: 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.

L. maculicollis: 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.

P. brasiliensis: 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 section 4.4. 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 section 4.4), 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 (50°F) (Sykes-Gatz 2005; V.Gatz, personal communication). The following recommendations are provided for giant otters:

- Exposure to air temperatures at or below ~10°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 must be provided in all climates (Sykes-Gatz 2005).

1.2 Humidity:

Since otters should always 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 section 1.4.1), natural substrates, and bedding material (see section 1.4.5) to aid the otters in proper coat maintenance, and allow for adequate drying of their pelts and feet.

1.3 Illumination

1.3.1. Identify light intensity, spectral, and duration requirements

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.

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).

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.

1.3.2. Address the impact of and need for daily changes in light intensity and seasonal changes in light intensity and duration

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.

1.4 Space

1.4.1. Behavioral repertoire, space requirements, and complexity.

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 may have a somewhat lower land to water ratio 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* require a land/water ratio of 3:1 for large, complex exhibits. As exhibits decrease in size below 232m^2 (2500ft^2), a greater land/water ratio is recommended. For a 149m^2 (1600ft^2) enclosure, Sykes-Gatz (2005) recommends 70% land area, and for a ~74m^2 (800ft^2) a land area of 75% is suggested. Sykes-Gatz (2005) offers a simple formula 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. These behaviors are among the most favored and frequently performed terrestrial activities in captivity, 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 due to stressful situations (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. Adequate land area and substrates on which otters can groom are considered key to the captive care of otters. In order to maintain healthy thermal properties of their coats, otters must frequently groom its fur replacing the air layer trapped within the under-fur (Dunstone 1998). See section 1.4.5 for species-specific requirements.

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., as these 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 for very shy animals should be made.

<u>Exhibit complexity – aquatic</u> (see also section 1.5): 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 off of 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. Furnishings should allow otters, especially pups and animals carrying pups, 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. See section 1.5.2 for recommended pool temperatures. Ample, extensive dry land areas must be provided to allow all otter species to dry off completely.

Exhibit complexity – terrestrial:: 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).

1.4.2. Minimum inter-individual distances that must be maintained and that will influence size of space.

See section 2.2.1.1.

1.4.3. Identify appropriate furnishings to accommodate an array of locomotory and foraging behaviors as well as resting and sleeping.

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. 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 old 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.

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 must be allowed ample grooming opportunities and surfaces on which they can rub to prevent matting, and aid in this annual coat replacement process (see substrates). 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 captive 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 land area, pool, and shoreline furnishing to maintaining dry areas in giant otter enclosures. 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. 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) 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 soil conditions, which can lead to health problems such as described in section 3.2.3.

Existing exhibits that do not meet these guidelines can be relatively easily and inexpensively modified. Sykes-Gatz (2005) and Hancocks (1980) are two of the many resources available with information on naturalizing old exhibits. Artificial and hard enclosure surfaces can be naturalized with the methods and substrates described for *P. brasiliensis* in section 1.4.5. These 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 nor more or less than 40-45°, and be located behind and near the water area shoreline (otters cannot dig deep enough in flat terrain and 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 Section 1.4.6 for enclosure barrier considerations to prevent escape by digging. See Appendix A for additional information on giant otter exhibit furnishings, substrates, etc.

1.4.4. Address the need for and appropriateness of visual, acoustic, and olfactory barriers within the space.

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 (see also section 2.2.3). 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 section 4.4 for further information.

In the rare cases when bonded pairs or other group members must 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.

1.4.5. Identify appropriate substrates and nesting/bedding materials if required.

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 show 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) 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). Some facilities have successfully used fleece and blankets (Ben-David et al. 2000, 2001a,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.

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.

P. brasiliensis: 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 sand or large size tree bark mulch/chips) that are deep 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. More 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 with sand or mulch at least 40-60cm (16-24") deep, or soil hillsides, to allow for deep digging. See Appendix A 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 should 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 and replaced with sand or tree bark mulch. Care should be exercised to ensure that all nest box substrates remain dry. Damp conditions may have contributed to otter pup mortality in at least one institution. After parturition, otters may remove 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).

1.4.6. Address mechanisms for the provision of change and variation in the environment.

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.

1.4.7. Address issues, such as scent marking, that may influence how and how often space is cleaned.

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.

1.4.8. Identify number of air or water changes/hour required

<u>Air changes</u>: 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 on 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 changes: 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 400ppm or lower (see section 1.5.1), which is the standard set for seal rehabilitation pools. A level of 100ppm is considered safe for humans. All chemical additives should be monitored daily and recorded.

Many municipalities add chlorine to their water; in these cases, 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 1ppm for prolonged periods (24 hours or more), and that levels should be maintained at 0.5ppm or lower, preferably at a reading of zero (see also section 1.5.1).

1.4.9. Identify necessary measures for safety and containment.

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.

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.

1.4.10. Address issue of transport, identifying (in accordance with IATA)

<u>Crate requirements</u>: All possible relevant regulatory agencies always 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.

<u>Pre-shipment exams</u>: All otters should receive a thorough pre-shipment physical examination (see section 3.2.2 for more details). 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 (APHIS form #7020). Institutions using MedARKS should provide the receiving institution with electronic copies of the medical records.

1.4.10.1. Type of transport container

Always consult the International Air Transport Association (IATA) guidelines for specific requirements for shipping containers. 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. All shipping crates must 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 must be covered with securely fixed weld mesh and all ventilation openings covered with wire mesh. The door must have secure fasteners at the top and the bottom. A curtain that can be raised and lowered and does not block ventilation must be fixed over the door to reduce light inside the container. A dropping tray must be fixed to the floor and filled with absorbent material. There must 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 must be correctly labeled. If the container has wheels, they must 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.

<u>*P. brasiliensis*</u>: Sykes-Gatz (2005) states that *P. brasiliensis* should not be transported in hard plastic containers in situations other than within an institution. 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. Transport crate floors constructed of many long slats of wood running horizontally and fixed over the top of the drop tray has proven successful at keeping otters clean and dry, as well as safely contained on long journeys. Slats should be placed close enough together to prevent the otter's feet from becoming caught in-between and going through the slats. If a flooring of this type is used, no substrates need be used. 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.

1.4.10.2. Appropriate size of transport container

Crates should be large enough to allow the animal to stand up and turn around. 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.

1.4.10.3. Provision of food and water during transport

The crate must allow 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.

<u>*P. brasiliensis*</u>: Sykes-Gatz (2005) recommends that giant otter be provided with drinking water at all times during transport.

1.4.10.4. Provision of bedding or substrate in transport container

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.

1.4.10.5. Mechanism(s) for separating animal from urine and feces during transport

In order to separate urine and feces from the animal, a drop tray must be fixed to the floor of the crate and filled with absorbent material.

1.4.10.6. Identify appropriate temperature range during transport

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).

<u>*A. cinereus*</u>: *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.

<u>*L. canadensis*</u>: *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.

<u>*P. brasiliensis*</u>: *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).

1.4.10.7. Consider appropriate light levels and how to minimize noise during transport

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.

1.4.10.8. Address appropriate group size or need for separation of individuals during transport

Otters should be transported separately. *A. cinereus* have been transported successfully in groups when the animals are less than six months of age.

1.4.10.9. Consider need for handler/veterinarian access to animal during transport

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, and the crate should be transferred to the nearest zoo or veterinary clinic prepared to handle the animal.

1.4.10.10. Consider maximum duration of transport allowable before temporary transfer to "normal housing" is required.

Otters should not be maintained in shipping containers for longer than 24 hours without food and water.

1.4.10.11. Address appropriate timing of release, size and type of enclosure at transport destination

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.

1.5 Water

1.5.1. Acceptable water quality parameters.

Otters are semi-aquatic mammals, using bodies of water for foraging, transportation corridors, mating (typically), cleaning, and "play-type" behavior. 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. Ultra violet sterilization has proven helpful in inhibiting algae build-up, particularly when combined with regular cleaning of poolsides. 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.

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, as 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. A turn over rate through the rapid sand filtration of at least 30ml (1oz) per hour is recommended. 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.

Coliform bacteria: 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 500ppm, or a MPN of 1000 coliform bacteria per 100ml water.
- Fecal coliform count should not exceed 400ppm.

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

Chlorine: 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).

<u>Algae control</u>: 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.

- *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.).

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

1.5.2. Appropriate means of presentation of water, and appropriate placement of water sources for terrestrial and semi-aquatic organisms.

<u>Temperature</u>: 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. Wünnemann (1995a) indicates that water temperature may be more critical than air temperature in determining the amount of time *P. brasiliensis* spends swimming. See Appendix A for information on pool design recommendations for giant otters.

1.5.3. Address issues of depth and need for variation in depth and/or current

Exhibit pools should have varying depths, offering opportunities for animals to forage in the shallow water and swim/dive in deeper water. Additionally, the pool should have sloping banks to allow for easy ingress and egress by pups, parents carrying pups, or old animals. Shorelines should be complex and curving, as opposed to straight lines and uninterrupted. As a method of containing exhibit substrate, a low, 4-inch curb (10.16cm high) constructed of flat/sloping rocks or thin logs connected with brackets can be placed along the shoreline (Sykes-Gatz 2005). 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 unpublished data). 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.

<u>*P. brasiliensis*</u>: *P. brasiliensis* pools should have deep area of at least 100cm (3.28ft), as well as shallow areas. Shallow areas are often the most frequently used by the otters. Depths of 150-200cm (5-6.5ft) and deeper are highly recommended to allow a wider range of swimming and diving behaviors.

2. Biotic Variables

2.1 Food and Water

2.1.1. Identify appropriate containers and protocols for the provision of food and 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.

2.1.2. Identify appropriate foodstuffs and feeding schedules

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 captive *Lutra lutra* consuming between 11.9-15% of their body weight maintained a healthy weight. Ben-David et al. (2000, 2001a,b) reported success using 10% of a captive *L. canadensis'* body weight as a guide for the basis of their maintenance diet. See section 3.1.2 for sample diet for the various otter species.

<u>Feeding schedule</u>: 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 times daily. *P. brasiliensis* should be fed 3-5 times per day, and these more frequent feedings are also effective with the other species. 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 (see also section 2.1.4). Timing of foraging opportunities and items offered should be varied to prevent habituation.

- *P. brasiliensis*: Food for *P. brasiliensis* should not be scattered 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.

2.1.3. Address the provision of variability in food type and presentation (e.g. spatial and temporal dispersal of food resources)

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 to 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.

- *A. cinereus*: The AZA Otter SSP has specific diet recommendations for this species (see also 3.1.2) that should be obtained from the AZA Otter SSP Chair, Dusty Lombardi (dusty.lombardi@columbuszoo.org).
- *A. capensis*, *L. canadensis*, *L. maculicollis*: See section 3.1.2 for sample diets and nutritional information. 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 so 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).

2.1.4. Address opportunities for animals to process food in ways similar to their wild counterparts, and consider mechanisms that enable animals to work for food

Live prey: 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 only. 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.

Puzzle feeders: There are a variety of puzzle, 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 Section 5.7 for other enrichment items used, including non-food items.

2.2 Social Considerations.

2.2.1. Group Composition, including as appropriate

2.2.1.1. Suggested age and sex structure of social group

A. cinereus: *A. cinereus* are monogamous with both sexes helping to raise the offspring. Unlike *L. canadensis*, the otter parents and offspring must be housed together. Older siblings help raise younger ones. It is recommended that these otters be held in adult pairs, adult pairs with offspring, or single sex groups. 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 captivity, pairs have been separated during parturition and early pup rearing.

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; M.Ben-David, personal communication; S.Shannon, personal communication). For example, Stevens & Serfass (in prep.) 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.

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, with 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).

Zoos and aquaria 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,b) maintained 15 unrelated males in one enclosure for 10 months with little to no problem. In general, if a group of animals are exhibited, the following groupings are recommended: all males; one male with multiple females (1.2); or equal opposite-sex pairings (2.2, 3.3, etc.), which have been successful in some cases. All female groupings, or even pairs of females are not recommended, unless they are sisters, mother/daughter, or have been introduced at a young age. 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).

L. maculicollis: This species is best housed as pairs or family groups. While several field researchers have reported seeing large groups (10-20 individuals) of *L. maculicollis* (Proctor 1963; Kruuk & Moorhouse 1990; Kruuk 1995), 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.

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 captivity, 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 in captivity, 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.

2.2.1.2. Temporary isolation of parturient females and young, or of males, and corresponding adequate and appropriate space for animals when removed

If it is necessary to separate animals, 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 barrier 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: In the wild, males do not participate in pup rearing (Melquist & Hornocker 1983; Rock et al. 1994). In captivity, parturient females should be given privacy (particularly for primaparous 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 (e.g., St. Louis Zoo, Homosassa Springs, and others). 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, generally at 60-75 days (Reed-Smith 2001).

Actual gestation is calculated at roughly 70 days (Bateman in prep.); 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 primaparous 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 section 2.2.5).

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 the Brookfield Zoo 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 (see section 2.2.1.5). Animals separated for extended periods should be put through a standard introduction (see section 2.2.5), 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.

2.2.1.3. Seasonal separation of sexes. For those species that are truly solitary, seasonal introduction of sexes

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 captive study (N=13.14 animals) attempted to determine breeding associated behaviors in captivity 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).

2.2.1.4. Nursery groups (groups of mothers with most recent young)

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.

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.

2.2.1.5. Forced "emigration" of adolescents

A. cinereus: Adolescents are not forced from the group. In captivity, 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, have been observed in the wild (J.Reed-Smith, personal observation).

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 captivity, 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 captive-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 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.

2.2.1.6. Multigenerational groups (e.g. many primates, elephants)

See section 2.2.2.1.

2.2.1.7. Groups deriving from cohorts (e.g., dolphin male pairs)

See sections 2.2.2.1 and 2.2.2.8.

2.2.1.8. All male groups

A. cinereus: All male (and all female) groups are not seen in the wild, but are found in captivity. Groups should be formed as early, and at as young an age as possible, and 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.

- *All-female groups*: 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,b; G.Blundell, personal communication; S.Shannon, personal communication). 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.

- *All-female groups*: 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 do well; sisters, or females introduced at an early age may be compatible for years. However, if one animal must be separated, they may not re-establish this social equilibrium upon reintroduction (Reed-Smith 2001). In one field study, an all female clan was identified, but this is the only reporting of this social situation (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).

- *All-female groups*: 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).

P. brasiliensis: Single sex groups, typically two males, have been kept together successfully. However, they must be raised together or introduced slowly to monitor any signs of potential aggression.

- *All-female groups*: There are a few reports of adult females living together. The introduction of adult females to one another is not advised (Sykes-Gatz 2005).

2.2.1.9. Daily and life stage variation in patterns of social affiliation

For life stage variation in social affiliation patterns, see sections 2.2.2.1-2.2.2.5, 2.2.2.8, and 2.2.4.1.

2.2.2. Group Size, including

2.2.2.1. Minimum and optimum group sizes

A. cinereus: In captivity, Asian small-clawed otters are monogamous, with both members of the pair helping to raise the offspring. Unlike *Lontra* species, the breeding pair and their offspring must be housed together. One breeding pair can produce two litters a year, with up to seven pups in each litter. Older siblings help raise the younger ones and a family group of these otters can build up to 15 or more in a year. Similar sized groups are sometimes found in the wild (Foster-Turley 1990). It is recommended that this species be held in adult pairs, adult pairs with offspring, or in 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).

A. capensis: This species has been held in pairs and in groups of 1.2 in captivity (Reed-Smith & Polechla 2002).

L. canadensis: 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, multiple pairs. 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).

Multiple pair groupings should be monitored closely for signs of stress in subordinate animals. Multiple male, one female groupings 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: Optimal group size will vary with the size of the exhibit. This species has been observed in groups of 10-20, as singletons, and all group sizes between (Proctor 1963; Kruuk & Goudswaard 1990; J.Reed-Smith, personal observation). In the wild, it is thought that this species typically live as pairs, with both parents participating in pup rearing. This has not been the case in captivity, 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).

P. brasiliensis: Giant otters are highly social and live in family groups. In the wild, mated pairs bond for life and all family members, including offspring from the parents' previous litters help to care for the pups (Duplaix 1980; Schenck & Staib 1994). Typically these older offspring range in age from 1-2 years of age.

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 captivity, 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 must 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 captivity (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 section 4.4 for additional information on pup rearing in this species.

2.2.2.2. Inter-individual distances required

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.

2.2.3. Conspecific groups, consider the need for/influence of adjacent groups, or similar taxa, on territorial species

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.

<u>*A. cinereus*</u>: If a group of Asian small-clawed otters must 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).

2.2.4. Mixed species groups

2.2.4.1. Identify appropriate species

<u>*A. cinereus*</u>: 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.

<u>*A. capensis*</u>: 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.

2.2.4.2. Identify key environmental elements for each species

All individuals housed in mixed-species exhibits should be routinely monitored for stress, injuries, and to ensure they are getting adequate food and water. See section 1.4 for details on necessary environment elements for otter species.

2.2.4.3. Identify interspecific inter-animal distances required

No generic information is available on inter-animal distances, and this will vary with the species housed with otters. 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.

2.2.4.4. Address appropriateness of single-sexed groups

No information is available on the advisability of maintaining single or multi-sex groups in mixed species exhibits.

2.2.5. Introductions

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.

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), 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. 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).

<u>*L. canadensis*</u>: 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. Unsuccessful introductions 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).

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), must 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 are common. During introductions/reintroductions, females are more commonly reported to have injured or killed males and even other females than have males. However, males have been known to 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) provide greater detail on the recommended introduction procedure for this species.

2.2.6. Human-animal interactions

2.2.6.1. Identify acceptable forms of human/animal interaction

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 (see sections 5.1 and 5.2). It should be kept in mind that otters are capable of inflicting severe bites, especially sexually mature females, and have been known to turn on their trainers.

2.2.6.2. Address both animal and keeper safety

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.

3. Health and Nutrition

3.1. Diet

3.1.1. Identify existing standards for nutrient requirements for all life stages if available

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 by (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 1 lists dietary nutrient ranges for otters.

Table 1: 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 10IU/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

The table provided in Appendix B 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 B also provides a description of the nutrients listed in these tables.

3.1.2. Provide sample recommended diet(s) for all life stages based on nutritional requirements and identify body condition norms as determined from wild animals, if possible

The one best diet for any of the captive otters 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, Dusty Lombardi (dusty.lombardi@columbuszoo.org), 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*.

- 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

<u>*L. maculicollis*</u>: 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.

<u>*P. brasiliensis*</u>: 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*), channel catfish (*Ictalurus punctatus*). Fish species marked with an asterisk (*) can be used as training reward or for vitamin delivery.

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 a 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

3.1.3. As appropriate address the influence of the following variables on dietary requirements

3.1.3.1. Age (infant, juvenile, reproductive adult, senescent adult)

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 a captive 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).

3.1.3.2. Body size

An animal's diet should be developed to maintain optimal weight for their species, size, and age.

3.1.3.3. Reproductive status

Lactation diet: 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 and 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 captive 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, or Chiclidae (tilapia) (21-32% fat DMB – Toddes 2005-2006 analysis), which are the low-fat fish commonly fed at the Philadelphia Zoo.

3.1.3.4. Seasonal changes in ambient temperature

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.

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).

3.1.3.5. Seasonal changes in body condition

An animal's weight should be regularly monitored and diets adjusted accordingly. At this time, further research into seasonal physiological changes is required.

3.1.3.6. Seasonal changes in nutritional requirements

An animal's weight should be regularly monitored and diets adjusted accordingly. At this time, further research into seasonal nutritional requirements is required.

3.1.3.7. Activity levels

An animal's weight should be regularly monitored and diets adjusted accordingly. While otters should carry some body fat, and not be kept artificially thin, they are prone to gaining excessive weight in captivity.

P. brasiliensis: 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.

3.1.3.8. Health status

Weight loss diet: 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 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.

3.1.4. Address issues of palatability, texture, processing, etc. that will encourage species-appropriate appetitive behaviors.

All otters will benefit from receiving live fish/crayfish (from approved sources), at least as enrichment on a weekly basis (see section 2.1.4). 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. See section 3.1.2 for additional information.

3.2. Medical management

3.2.1. Quarantine and hospitalization

Quarantine: 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.

- *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 first, and then 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 must 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 3.2.2).
- *Final exam*: During the last week of quarantine, a thorough examination should be conducted as outlined in section 3.2.2. 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)

3.2.1.1. Identify problems arising from isolation of social taxa and suggest possible mechanisms for avoiding these problems

P. brasiliensis and *A. cinereus* should be housed in social groupings, as far as possible, during quarantine. See section 2.2.1 for additional information.

3.2.2. Preventive medicine (testing, vaccinations, parasite control, etc.)

Medical examinations: It is recommended that all animals should have at least a biannual 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 bears 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*, biannual 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 captive species. 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, 1986). 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. Dose 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.

<u>Parasite control</u>: 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 for parasite testing are provided in the table below:

Table 2: Parasite testing protocols

Parasite	Testing protocol
External parasites	Regular inspections during any physical examinations
Internal parasites	*Annual fecal examinations*: direct smear, fecal flotation, & sedimentation or Baermann techniques.

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

- *Parasite treatment*: The following table (Table 3) provides a list of anthelmintic products that have been used safely in a variety of mustelids:

Table 3: 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

Identification: 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 interscapular 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 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.

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

3.2.3. Management of (hereditary) diseases or disorders

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 fur within their coats rather than a layer of blubber for thermal insulation. 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 captive 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., 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).

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

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

Cause of death	**Causal factors**
Heart disease	- Heartworm/death from heartworm treatment; Acute myocarditis; Myofiber degeneration
Renal failure	- Etiology unknown; Amyloidosis; Pyelonephritis
Hepatic lipidosis	-
Adenocarcinoma	-
Transitional cell carcinoma (bladder)	-
Peritonitis	- Secondary to intestinal perforation from foreign body; Secondary to GI perforation from ulcers
Diarrhea	- Unknown etiology; Clostridial endotoxin; *Helicobacter* (also causing vomiting, weight loss); *Salmonella*
Gastric dilatation with volvulus	-
Pneumonia	- Often without identifying underlying cause
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 mistreatment towards pups due to stress from human disturbances or inappropriate enclosure 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).

Illness seen in this species, most often when housed in sub-standard conditions include: skin lesions (particularly of the tail and hind legs) due to inappropriate substrates. 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 over the age of 4 years. Again, the causal agent appears to be continued exposure to hard surfaces. Other physical problems caused by exposure to hard or continuously damp conditions include; foot pad abrasions, irritation of the foot's webbing, and poor coat condition (Sykes-Gatz 2005).

3.2.4. Appropriate capture, restraint and immobilization techniques and training for routine and non-routine procedures.

It is recommended that anesthesia be given 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 animal. 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 entubating the animal and maintaining it on Isofluorane (Ohmeda Pharmaceutical Products Division Inc., P.O. Box 804, 110 Allen Rd., Liberty Corner, NJ 07938) or Halothane (Fort Dodge, 9401 Indian Creek Parkway, Ste. 1500, Overland Park, KS 66210) anesthesia. Otters are relatively easy to entubate, and this method is preferred when it is necessary for an animal to be immobilized for a lengthy procedure.

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 entubated 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, plus it 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

3.2.5. Management of neonates and geriatric animals

Otter pups can develop health issues suddenly, and they must 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.

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).

<u>Diarrhea/constipation</u>: Digestive upset is a common issue with hand-reared neonates, and may be associated with several factors (Meier 1985): a) inappropriate milk formula; b) feeding frequency; c) overfilling the stomach which can cause bloating; and d) 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 must 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.

3.2.6. Management during pregnancy

At this time, the AZA Otter SSP has no specific health treatment recommendations for pregnant females. See section 4 for reproduction information.

4. Reproduction

See Appendix C for breeding information on *A. cinereus*, *A. capensis*, and *L. canadensis*.

4.1. Identify seasonal changes in physiology and behavior associated with reproduction and address management implications of such changes

A. cinereus: These otters are non-seasonal, and are believed to be induced ovulators. 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).

Breeding pairs must 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 (68-74 days reported by H.Bateman, unpublished data). 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.

A. capensis: This species does not appear to have a specific reproductive season (Mead 1989). Breeding in the northern hemisphere (Ohio) 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 captive breeding situation, receptivity by the female lasted one day (Oregon Zoo, 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 captive 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.

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 (H.Bateman, in preparation).

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 remain distended until the end of the breeding season (Liers 1951; Reed-Smith 2001; Bateman in prep).

There is evidence that breeding season varies somewhat with latitude (Reed-Smith 1994, 2001; Bateman et al. 2005). 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). During this time, captive observations 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).

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. 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. 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. Successful copulations must be at least of several minutes duration. 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 of 42-46 days (Reed-Smith 2001; Reed-Smith in prep.). 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 10.5-12 months (Liers 1951; Reed-Smith 2001); actual gestation is about 70-72 days (Bateman, in preparation). 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).

A postpartum estrus occurs soon after parturition lasting the same 42-46 days of a typical estrus. Hamilton and Eadie (1964) give the postpartum 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). 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 is involved in on-going research.

L. maculicollis: Schollhamer (1987) reported that females at the Brookfield Zoo 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, and 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 captivity (R.Willison, personal communication). 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). Captive 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 work in Tanzania has not been able to validate this observation (J.Reed-Smith, personal observation).

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 captivity, 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. Captive 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.

4.2. Address hormonal tracking as a mechanism for identifying reproductive state, and assessing feasibility of introduction for solitary species

Research utilizing these 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 (H.Bateman, unpublished data). Helen Bateman, at Cincinnati Zoo's Center for Research in Endangered Wildlife (CREW) and reproductive physiology advisor for the AZA Otter SSP, should be contacted for more information: helen.bateman@cincinnatizoo.org.

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.

4.3. Address timing of introductions for individuals of solitary species

Not applicable for these species. If male and female *L. canadensis* are housed alone, the pair should be introduced after the first signs of estrus appear, or when the female shows unusual interest in the male's scent or enclosure.

4.4. Address provision of and describe facilities for parturition and as appropriate, management of females during isolation or denning.

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.

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"), and be large enough for an adult animal to move around in, and for 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. 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 are very susceptible to any human disturbance, especially within the surroundings of the natal den, and this can have fatal consequences for the young (Sykes-Gatz 2005). They also are sensitive to poor or sub-standard enclosure conditions. Several steps have been recommended to reduce stress to the reproductive pair and improve pup-rearing success (Sykes-Gatz 2005):

- Build a positive keeper-animal relationship and allow only familiar staff to work with the otters prior to and 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 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.
- Monitor the female’s weight during the pregnancy; if the female becomes uncomfortable with this routine it should not be forced.
- Isolation of the natal den and limitation of all human activity in the vicinity prior to the birth and until staff determines the pair and young have bonded and the young are thriving is very important and cannot be stressed enough.
- 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, both natural (e.g., dug by the otters) and man-made. The recommended size is 43-97ft^2 (4-9m^2). Ideally, pupping boxes just large enough to hold the adults 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 Hagenbeck Tierpark (Flügger 1997). Captive *P. brasiliensis* 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 thermoregulate 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 by contacting Sheila Sykes-Gatz at v.gatz@cityweb.de.

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 necessarily 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 and non-stressful situations, it may appear that both parents sometimes treat their pups roughly. This kind of behavior may be carried out whether parents are in the nestbox, on the land, or in the water. This is especially evident when parents 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 are not actually too rough with their pups.

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 captive-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 is known as 'Eskimo rolling' (Sykes-Gatz 1999-2006). Pups also may be gently pushed/pulled into the water to encourage swimming.

Following is a list of parental behaviors observed in captivity 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 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, can be abnormal.
- Excessively forceful throwing or pushing 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 is 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 a problem with lactation, such as insufficient milk production.
- Biting, hitting, or laying on the pups; and drowning pups

Further information on these abnormal occurrences can be obtained from Wünnemann 1995a,b; Flügger 1997; Autuori & Deutsch 1977; Corredor & Muñoz 2004.

4.5. Address what, if any, circumstances might warrant hand-rearing and identify acceptable hand-rearing and reintroduction protocols.

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 must 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 Appendix D 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). However, *P. brasiliensis* are known to carry their pups around as part of normal rearing (see section 4.4).

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 singletons pups being hand-reared be placed together, if at all possible. To date, fostering has not been attempted with otter pups but should be tried if a suitable female is available at another facility. In these cases, the AZA Otter SSP management team should be consulted first.

<u>Physical care protocol</u>: 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 thermoregulate. 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 must 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 Sutdbook, Husbandry, and Management Information and Guidelines (Sykes-Gatz 2005), respectively. The hand-rearing of giant otters (*P. brasiliensis*) is somewhat different than that of other otter species (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).

<u>Feeding amount and frequency</u>: 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 (MER): 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 MER (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 value 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.

Example 1: Approx. birth wt = 135g.

1. 84.6 x 0.135kg $^{0.78}$ x 3 = ~53 kcal/day
2. 135g x 0.05 (stomach capacity of 5% body wt) = ~7g (ml) can be consumed per feeding
3. With a formula that contains 1.78 kcal/ml: $\frac{53 \text{ kcal/day}}{1.78 \text{ kcal/ml}}$ = ~30 ml/day
4. Based on feeding capacity $\frac{30\text{ml/day}}{7 \text{ ml/feeding}}$ = 4.2 feedings/day (=5)
5. 24 hrs/5 feedings = 5 hours between feedings.

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 and 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.

<u>Feeding techniques</u>: 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.

- *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.
- *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 http://www.otterspecialistgroup.org/Library/TaskForces/OCT.html).

If an animal aspirates formula, the veterinarian should be consulted. 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 a large amount of formula is aspirated, or if fluid is heard in the lungs, Lasix™ (Furosemide) may be given, under the supervision of a veterinarian (K.Grant, personal communication).

Pups will need to be stimulated to urinate and defecate for the first six weeks of life, either immediately before or after feeding. Captive parent-reared giant otters 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.

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 5 provides information on the nutritional content of otter milk, and Table 6 provides information on the nutritional composition of selected substitute milk formulas. See Appendix E for a nutritional analysis of commercial animal milk replacers.

Table 5: 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 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 must be kept as low as possible to prevent gastric upset and diarrhea.

Table 6: 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
Formula #1 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
Formula #2 1 part Multi-Milk® or Milk Matrix 30/55® 1 part water	31.3	17.8	10.4	1.1	1.91

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 must be refrigerated for 24 hours prior to feeding for the enzyme to perform correctly (Grant 2005). *Lactobacillus* spp., in Bene-bac® or Probios®, are a group of beneficial gut bacteria that also break down milk sugars in the digestive tract. Follow label instructions for these products.

- *L. Canadensis*: 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*. 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): a) weight loss; b) diarrhea; and c) 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).

<u>Pup development</u>: 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 F for pup weight charts.

- *A. cinereus*:
    - 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 is 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 primaparous 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 nurse for 6.5-7 months but will begin weaning at roughly 4 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.
- Mother-reared pups may nurse until ~ 8 months of age
- Pups are approximately ¾ the size of adults at 10 months of age, although this will vary

<u>Swimming, terrestrial activities, and behavioral stimulation</u>: 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 at an early age, the more they will interact with as they age. All toys must 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.

4.6. Recommend means and duration of contraception for taxon; include all acceptable alternatives and identify the benefits and drawbacks of each

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 AZA Wildlife Contraception Center (WCC) 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. For carnivores, the AZA Wildlife Contraception Center now recommends GnRH agonists, e.g., Suprelorin® (deslorelin) implants or Lupron Depot® (leuprolide acetate) as safer alternatives. Although it appears 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.

<u>Gonadotropin releasing hormone (GnRH) agonists</u>: GnRH agonists (e.g., Suprelorin® implants, or Lupron Depot®) achieve contraception by reversibly suppressing the reproductive endocrine system, 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 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, 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) must 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.

- *P. brasiliensis*: Contraceptive hormones should not be used in giant otters due to possible side-effects such as endometritis/pyometra and the potential of inhibiting future successful breedings (C.Osmann, personal communication). To date, contraceptives have not been tried in this species.

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.

5. Behavior management

5.1. Identify procedures that have been successful in managing the taxon for routine husbandry.

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 Appendices G and H for a list of commonly trained otter behaviors, as well as relevant cues and criteria.

5.2. Identify procedures that have been successful in managing the taxon for non-routine husbandry.

As mentioned above, 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).

5.3. Identify procedures that have been successful in facilitating introductions. These may include separation of individuals from group, stationing, tolerance while feeding, "howdy" units, visitation gates, etc.

See section 2.2.5. Auditory, visual, and olfactory introduction must 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 this has not been tried with any of the otters.

5.4. Identify facility design considerations, husbandry training techniques, and implementation plan that can be used to elicit desired behaviors in a way that is safe for both caretakers and animals.

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. Exhibits should be designed with mesh at a particular area specifically for training. In addition, if institutional philosophy permits, otters can be a part of an educational talk or keeper talk in a free contact area within their exhibit.

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); there are some species (*A. cinereus*) or cases (*L. canadensis* particularly males) where an institution feels this precaution may not be called for but, these decisions should be carefully evaluated on an ongoing basis. 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.

5.5. Identify those techniques that have been shown to be most effective.

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 Appendices G and H for additional training information.

5.6. Identify technical skills and competencies needed by staff

The following skills are recommended for all animal caretakers involved in the management of captive otters:

- 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.

5.7. Appropriate methods of enrichment for the taxon should be identified if not included in categories 1-3 above.

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 captive 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 follow 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; see also Appendix J). This list includes key questions that should be answered for all enrichment items or programs to assess potential hazards. For example:

1. Can the animals get caught in it or become trapped by it?
2. Can it be used as a weapon?
3. Can an animal be cut or otherwise injured by it?
4. Can it fall on an animal?
5. Can the animal ingest the object or piece of it? Is any part of it toxic, including paint or epoxy?
6. Can it be choked on or cause asphyxiation or strangulation?
7. Can it become lodged in the digestive system and cause gut impaction or linear obstruction?
8. 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?
9. Can it destroy an exhibit?
10. If fecal material is used for enrichment, has it been determined to be free from harmful parasites?
11. Is food enrichment included as part of the animals' regular diet in a manner that will reduce the risk of obesity?
12. When introducing animals to conspecifics or in a multi-species exhibit, are there sufficient areas for them to escape undesirable interactions?
13. Can the manner of enrichment presentation (i.e., one item or items placed in a small area) promote aggression or harmful competition?
14. Has browse been determined to be non-toxic?
15. Do the animals show signs of allergies to new items (food, browse, substrates, etc.)?
16. Does the enrichment cause abnormally high stress levels?
17. 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)?

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 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 *L. brasiliensis*, but applies to all of the otter species due to their inquisitive nature and high-activity level.

<u>More information</u>: 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.

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APPENDIX A

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.

Written by 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 must 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 must 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 must 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. <u>*Each indoor and outdoor giant otter enclosure also needs*</u> 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 must have 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 must 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 must 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 B

Target Nutrient Ranges for Carnivorous Species (dry matter basis)

Nutrient	NRC 1986 Cat[1] Maintenance	NRC 2006 Cat[2] Growth	NRC 2006 Cat[2] Maintenance	NRC 2006 Cat[2] Gestation Lactation	Arctic fox[3] Maintenance	Mink[4] Maintenance	Carniv[5] 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

Description of Nutrients (US National Library of Medicine)

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.

APPENDIX C

Captive Breeding of River Otter (*Aonyx capensis*, *Aonyx cinerea*, *Lutra canadensis*) in North American Zoological Facilities 1980's and 1990's (Reed-Smith and Polechla 2002)

	Aonyx capensis	*Aonyx cinerea*	*Lutra canadensis*
Estrus Cycle	Breeding occurred in Jan, Apr, Nov & Dec for 4 litters produced.	30-37 days. Breeding occurs year-round.	42-46 days unless mating occurs; can occur Nov-Jun
Estrus Length	Peak receptivity lasted 1 day; day before & after consisted of play & close following	1-10 days	Receptivity peaks roughly 6 days apart.
Copulation Freq.	Several times during the 2nd day of breeding behavior	Several times a day.	Several times a day
Copulation Dur.	Not documented.	1-30 minutes, varied.	20-45 minutes, varied. One >60 min. reported
Copulation Position	Not documented.	Dorsal/Ventral most common, also ventro/ventral.	Dorsal/ventral most common, also ventro/ventral.
Copulation Where	In the water.	In the water and on land	Most frequently in the water, also seen on land
Copulation Initiated By:	Male, females would only cooperate for 1 day.	Varies amongst groups, in some it is initiated by male only, in others both initiate.	Both. Female advertises, cooperates only when she is ready. She may initiate with invitations to play chase.
Age at 1st breed.	Males from 2 yrs.10 mos.; Females from 4 yrs. 2 mos.	Unknown	Sexually mature by 2 yrs. Several 2 yr. old males & a 1.5 yr. old female have bred successfully.
Breeding Behav.	Females fought when one was in heat. Signs of estrus shown by male behavior	Increased rubbing, marking.	Female may rub, mark or vocalize to advertise; male/female may initiate w/ play, chase, splashing, genital sniffing, or "butterfly stroke".
Gestation	80, 103 days	Gestations of 62, 64, 65, 72, 83, 84, and 86 days are on record.	332-370 days total gestation, documented for 12 litters. 285-380 (Liers 1951)
Pair management	Breeding was opportunistic. Male kept away from pups by female or separated 1 week prior to expected parturition.	Most facilities started out w/ a pair when 1st litter born. Pair left together all of the time	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	1.2 housed together on exhibit during the day; 0.2 given 2 dens @ night	Some reports of harassment of new pups by older pups (too much play), one reported cannibalism of pups by dam. Most leave all animals together.	Sire can be reintroduced to female and pups after pups are swimming well. Generally done at about 3-6 months.
Signs of parturition	Females gained weight particularly in fold between foreleg and body.	Some weight gain, more time spent in nestbox.	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	Females did not want bedding in their boxes. Given only one box, no problems.	Wooden boxes, hollows under logs and burlap bags have all been used.	Pupping boxes should be filled with dry bedding (straw or hay). A choice of birthing boxes should be available.
Contraception	Two males have been vasectomized due to small gene pool. Females medicated for contraception.	MGA implants in females.	MGA implants and PZP treatment in females. A few males have been neutered.

APPENDIX D

Neonatal Examination & Monitoring Guidelines (Read & Meier 1996)

1. **Vital Signs**
   - Temperature, include activity level
   - Pulse, rate and character
   - Respiration, rate and character
2. **Organ systems**
3. **Weight**
4. **Hydration,** skin tone and turgor
5. **Mucous membranes,** color and capillary refill
6. **Vitality,** response to stimulation, activity levels – type, frequency, duration
7. **Physical condition**
8. **Laboratory values** (optional)
   - Complete blood count
   - White blood cell count
   - Serum chemistries, including blood glucose and blood urea nitrogen
   - Urinalysis and urine specific gravity (recommended)
9. **Urination,** frequency, amount, and character
10. **Defecation,** frequency, amount, and character
11. **Condition of umbilicus**
12. **Total fluid intake,** amount in 24 hours
    - Parenteral fluids, amount, frequency, and type
    - Oral fluids, amount, frequency, type, nipple
13. **Housing temperature**

APPENDIX E

Nutritional Analysis of Commercial Animal Milk Replacers

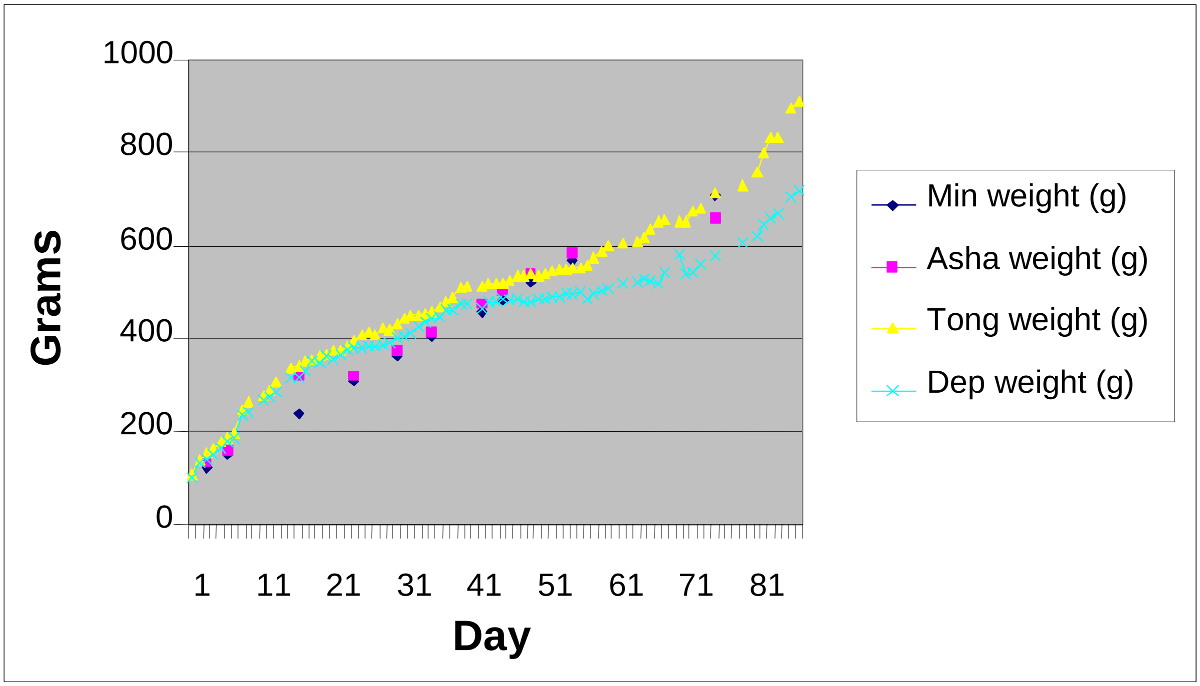
Product	Solids %	Fat %	Protein %	Carbohydrates %	Ash %	Energy (KCAL/ML)
Esbilac						
Undiluted powder	95.00	40.00	33.00	15.80	6.00	6.20
Diluted 1:3*	15.00	6.00	4.95	2.38	0.90	0.93
Diluted 1:1.5*	30.00	12.00	9.90	4.76	1.80	1.86
Liquid product	15.00	6.00	4.95	2.38	0.90	0.93
KMR						
Undiluted powder	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)

APPENDIX F

Pup weights of captive bred otters

Asian-small clawed otter (*A. cinereus*) pup weights (mother-reared) at Indianapolis Zoo (N = 1.3)



Spotted-necked otter (*L. maculicollis*) pup weights (mother-reared) at Phoenix Zoo 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, Little Rock Zoo, Grassmere Wildlife Park, John Ball Zoo.

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 G

List of commonly trained behaviors – 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.

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%
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 H

Sample Behaviors and Training Cues for Otters (provided by: *Indianapolis Zoo; **Bronx Zoo; ***Toledo Zoo; ^Santa Barbara Zoo; ^^Point Defiance Zoo; and +Oklahoma City Zoo)

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 placed 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 cage front	'lie"	Palm flat out and facing down. Sweep arm in direction animal should face.	Animal lays down parallel to and 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.
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 or kidneys are 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 cage with palm to push the food into the pool	The animal should jump into the water to retrieve food.
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

APPENDIX J

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 heirarchy.
- 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.

APPENDIX K

Enrichment initiatives commonly provided to captive 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	- Climbing areas (available in all exhibits, i.e., cliffs, ledges)	- Boomer balls and other products like the "spoolie", “bobbin” & "ice cube".	- Fish (smelt, shiners, goldfish, trout, mackerel, tilapia salmon)*	- Ice blocks w/fish, fish-sicles, fish cubes, etc.
- Grass, wheat grass, sedges, etc.	- Logs (on land, submerged, floating; hollow and/or solid)	- Ice blocks, cubes, pops.	- Crayfish	- krill cubes, clam cubes, etc.
- Trees	- Rocks (not artificial)	- Snow & ice	- Crickets	- Frozen or thawed sand eels
- Vines "vine hoops"	- Stream	- PVC cricket feeder	- Giant mealworms	- Fish pieces
- Aquatic plants	- Sticks	- Buckets	- Earthworms	- Chicken necks
- Hay, straw, grass, leaves, wood wools as bedding	- Browse (leafy branches on land and/or floating)	- Blankets, burlap, non-fraying rags, towels	- Freshwater clams	- Mice
- Grass piles	- Slides	- Barrels of water	- Mussels	- Whole-fish -frozen or thawed
- Leaf piles	- Tunnels	- Frisbees	- Krill	- Whole apples/oranges
- Rocks, all sizes for play and manipulation	- Stream bed	- Tubs of water	- Eels- naturally found	- Fruit & berries incl. grapes, blueberries, strawberries
- Knot holes	- Running water	- Carpet over board	- Shrimp	- Small pumpkins and squash
- Bark sheets	- Holts	- Rubber-coated heating pad*	- Aquatic insects - naturally found	- Omnivore biscuits
- Pine Cones	- Jacuzzi-like jets in pool	- Astro turf	- Mice- naturally found	- Monkey chow
- Mud	- Islands in pool	- Floating plastic toys	- Frogs – naturally found	- Pigs ears
- Sod	- Bridges made from logs, etc.	- Phone books	- Grubs	- Frozen blood blocks, cubes, etc.
- Bank over-hangs	- Stumps	- Swim through plastic ring	- Chub	- Hard-boiled eggs
- Floating wood	- Natural fiber mat	- Kids puzzle balls, billiard balls, hard balls	- Minnows	- Day-old chicks
- Blocks	- Movable sand box	- Pieces of PVC pipe and fittings	- Bluegill	- Crabs
- Pine needles	- Logs brought from other exhibits	- Kong chews	- Clams	- Melons
- Other animal urines	- Log ladder	- Metal bowls and pans	- Mud minnows	- Coconuts
- Powdered scents and herbs	- Non-sprayed evergreen trees	- Plastic tubs and bottles		- Frozen feline balls
- Fresh herbs	- Moving soil pots	- Bread tray		- Milk bones
- Extracts, i.e., vanilla, etc.	- Hanging logs with holes for food	- Plastic slide, house		- Screw pine nuts, unsalted peanuts
- Grapevine balls	- Snow piles	- Stock tank		- Krill patties
- Shells	- Piles of ice cubes	- Hanging tub*		- Hamster ball w/ treat
- Turkey feathers		- Warm water hose		- Gelatin Jigglers
- Corn stalks		- Vari-kennel tubs with substrates		- Corn on the cob
- Blowing bubbles into exhibit		- PVC tube hung for climbing in		- Yogurt with fish
- Kudzu vines				- Unsalted ham
- Cow hooves				

* These items should be monitored for safety.

The following list provides more examples of enrichment initiatives offered to otters at the Point Defiance Zoo and Aquariums and Columbus Zoo and Aquarium:

Point Defiance Zoo and Aquarium – 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

Columbus Zoo and Aquarium – 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 (S. Maher, Disney Animal Kingdom)

Enrichment

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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: http://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, http://www.aazk.org/2004enrichnotebookcd.php.

- Fort Worth Zoo's Enrichment Online: www.enrichmentonline.org/browse/index.asp

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