NORTH AMERICAN RIVER OTTER

Husbandry Notebook, 4th Edition; Chapters 7 - 10

(Photo: Dave Mellenbruch)

NORTH AMERICAN (Nearctic)
RIVER OTTER *(Lontra canadensis)*
Husbandry Notebook, Section 2 Chapters 7 - 10©

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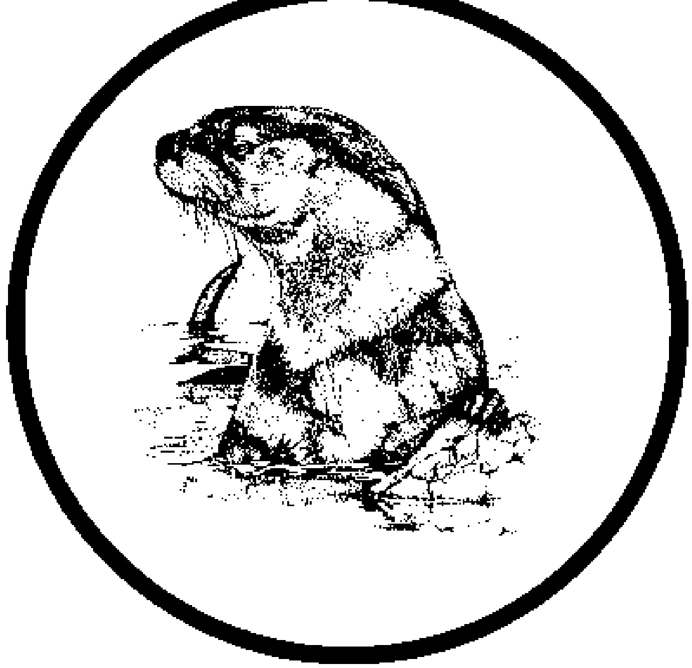

"Alacris ad ludos est."

"It is quick to play"

(Albertus Magnus, 13th Century teacher and naturalist)

North American River Otter Husbandry Notebook
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Photo: Julie Katts

In the days when the earth was new and there were no men but only animals the sun was far away in the sky. It was so far away that there was no summer. It was so far away that the trees and the grasses did not grow as they should.

He-Who-Made-the-Animals saw how it was that there was not enough sun to heat the earth, and so he fashioned a snare. The Sun did not see the snare in his path, walked into the snare and the snare held him fast.

The sun was close to the earth. In fact, the snare held the sun so close to the earth that there was no night. Day after day the sun shone and the earth dried and the grasses withered. There was not enough food or water for the animals and they desperately called a council. "Sun," the animals said, "You give too much heat to the earth."

"Set me free from this snare" the Sun said, "and I will go away."

"But if you go away, then there will not be enough heat." "Set me free," the Sun said, "and I will come to the edge of the earth in the morning and in the evening; then at noon-time I will stand straight above the earth and warm it then."

The animals sat around the council fire and they said, "Who is going to set the sun free?"

"I shall not do it," Wildcat said. "Whoever sets the sun free must go so close to the sun that he will be burned to death." Lynx said, "Whoever sets the sun free must chew the leather thong that holds him; the sun will burn him to death before he can do it." "I shall not do it," said the deer, the wolf and the raccoon.

"I shall do it," Otter said. "How can you do it?" said the animals. "You are too small, your teeth are for fish, and your fur has already burned away." None of the other animals liked the otter because he played too much. They did not think he was brave.

"Let him try," Bear said. "He will burn to death, but we will not miss him. He is of no use to us. He looks silly now that his fur is gone." The animals laughed.

Ignoring the taunts, the otter set off to the place in the sky above the earth where the sun was held by the snare. Otter took many days to get to the sun. The sun burned him. The sun was so bright, Otter had to close his eyes. When he reached the sun, Otter began to chew on the leather thong that held the sun. His skin was burning and blistering, his eyes were hot stones. But, Otter did not stop chewing.

Suddenly he chewed through the leather. The animals saw the sun rise into the sky. The animals felt the cool winds begin to blow on the earth. Otter had freed the sun from the snare.

Time passed. Otter lay in the center of the council ring. There was no fur at all left on his body. His skin was burned and scorched and his flesh was falling off his bones. His teeth were only blackened stumps.

He-Who-Made-the-Animals also stood in the center of the council ring. "Otter," he said, "the animals will not forget what you have done for them. I will see that they do not forget," and he gave Otter new strong teeth, tireless muscles, keen eyesight, and a powerful tail to help him in his hunting and in his play. He did not have to give him bravery. But he gave him new fine fur that was like down on his skin, and a second coat of fur to guard the first so that he would not get cold in water or in winter. Then he gave him joy so that he would always be happy in his otter's life, and Otter has so remained until this day.

An Otter Legend derived from the Cree Indians
Contributed by John Mulvihill
The River Otter Journal Vol. VIII, No. 2, Autumn 1999

Contributors

4th Edition

Thank you to all who contributed to the 1st and 2nd editions as well as the 1997 Husbandry Survey (the 3rd edition was never published). Some of this information is still part of this edition. However, the 2nd edition is available on the IUCN Otter Specialist Group website and the original Otter Lore and other deleted sections can be found there.

Contributors to this new edition include: Helen Bateman, Gwen Myers, DVM, Melanie Haire, Tanya Thibodeaux, David Hamilton, Brian Helton, Lynn Hougle, Jennifer Mattive, Kristina Smith, Mike Maslanka, M.S., Barbara Henry, M.S., Monica Anderson, Nicole Barker, Rachael Chappell, Julie Christie, Kristin Clark, Erin Dauenhauer-Dacota, Erin Erbren, Bethany Gates, Katie Jeffrey, Maggie Jensen, Brett Kipley, Marcy Krause, Tara Lieberg, Hilary Maag, Christine Montgomery, Melissa Newkoop, Melanie Pococke, Josh Prince, Nancy Ramsey, Tami Richard, Karen Rifenbury, Jan Sansone, Ashley Snow, Alicia Striggow, Maicie Sykes, Janée Thill, Marla Tullio, Jen Wilson, Andrea Dougall, Victor Alm, Courtney Lewis, Bill Hughes, Jennifer Galbraith, and the Otter Keeper Workshop Attendees (2004, 2006, 2008, 2010, 2012). Thank you to the zoo and aquarium people who contributed photos and to the professionals who gave permission for use of their photographs: Dave Mellenbruch, Haley Anderson, Graham Jones, Gary Woodburn, Debbie Stika, and Herb Reed.

USER GUIDE

INTRODUCTION

***Lontra canadensis* is most commonly known as the North American river otter but also will be referred to here as the N.A. river otter, NARO, and Nearctic otter.**

As soon as the first edition of the North American River Otter Husbandry Notebook was completed additional information became available – that is the way projects of this nature all work. I have no doubt it also will be true for this edition. Each edition should be used as a beginning point when looking for an answer to a particular otter problem or question. Our approach to captive husbandry should be as dynamic as the animals in our care. **This 4th edition includes updated information. Since publication of the last edition significant work has been done on otter reproductive physiology, contraceptive recommendations have changed, and there have been some changes made to recommended routine veterinary care. These changes as well as additional enclosure, training and enrichment information have been included in this digital update of the NARO husbandry notebook. All deleted information and sections (e.g. North American River Otters in European Institutions) are still available in the 2nd edition. The 2nd and 4th editions are available at otterspecialistgroup.org, Otters in Zoos, etc. link (OZ Task Force – Otters in Zoos, Aquariums, Rehabilitation, and Wildlife Sanctuaries).**

Where possible, all measurements and weights have been put into the English and metric systems. This is not true for the weights tables, however. There is some duplication from one chapter to another; some information on a given topic may only appear in one location. This is inconsistent but an attempt was made to at least provide some basic information on pertinent topics where appropriate so a reader would not have to go to all of the sections. For example: there is pup development information in the Reproduction section and Hand Rearing.

Many thanks go out to all of the people who have shared ideas with me over the years, too many of you to name here however, your contributions have all been helpful and have been incorporated in some way in this manual. The notebook has been split into three sections allowing the inclusion of more photos while trying to keep the file sizes manageable. They are as follows:

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CHAPTER 7 Captive Management

*"Otters in general are most attractive exhibits in the zoological garden, their antics in the water being especially engaging. The races of **L. canadensis** and **L. lutra** are best suited for northern institutions, since their hardiness permits them to be shown in permanent outdoor installations. The first essential is a pool of fresh, clean water, deep and long enough for swimming, diving, and play. Moderately running water is certainly preferable, if not actually essential. There should also be sufficient land area to allow reasonable space for the explorations in which the otter seems to delight. For, while we tend to think of the otter in terms of water, the animal really spends much more time out of that element than in it. A dry, clean shelter, well packed with clean straw, will complete the requirements, at least as far as the welfare of the otters is concerned."*

(Crandall 1964)

Introduction

The basic requirements for a river otter exhibit have been understood for a long time, at least since Crandall was writing in 1964. However, zoological institutions were not always familiar with what Crandall had to say. As a result, many old river otter exhibits were poorly designed and did not address the species behavioral or physiological needs. Additionally, in the past the zoological community had a tendency to think: small mammal – small exhibit. Wallach & Boever (1983) gave minimum dimensions for otter (species not specified, figures taken from the AZA Animal Health Committee) as 10.98 m^2 floor space for one animal and 12.80 square meters floor space for two animals; minimum height is given as 1.22 meters. More recently the AZA publication, Minimum Exhibit Requirements (1997) gives 231 $ft.^2$ as a minimum enclosure requirement for two animals, with additional space 'probably needed' for breeding pairs.

After looking at 25 zoos and observing more than 100 otters, Duplaix-Hall (1975) concluded that the lack of breeding success could be linked to three factors: "*…the enclosure, the diet, and the otters themselves."* Duplaix-Hall believed that most often, the lack of success was due to one of the first two, inadequate enclosures and/or poor diet.

As of the writing of the 1st edition of this manual there had been improvements in our captive management of river otters however, U.S. and Canadian zoos were only beginning to experience anything more than hit and miss breeding. The reasons for this appear to have been one or more of the following:

- Inadequate enclosure and den design
- Insufficient enclosure space,
- Insufficient numbers of animals to allow for mate selection;
- A lack of the resources needed to implement an appropriate breeding arrangement protocol,
- Paired animals that were introduced at too young an age which may have caused them to become too familiar with one another,
- A lack of interest in breeding this species

Over the last decade significant improvements have been made in our ability to breed Nearctic otters in zoos, aquariums, and wildlife sanctuaries. This improvement is due to greater cooperation between facilities, good population management led by the AZA Studbook Keeper, improved enclosures and pair management, as well as better understanding of river otter reproductive physiology and otter behavioral needs.

Understandably, it is not possible, or desirable, for every institution housing river otters to breed them.
However, it is advisable that the otters are housed and cared for in a manner that does not preclude breeding if it becomes a desired goal. Proper diet, adequate enclosure facilities, and good management

practices are important to the health and well-being of otters. The recommendations contained in this section are based on a literature search, historical records, 20 years of personal experience and visiting over 60 otter facilities, as well as input from over 100 professionals who work with Nearctic otter in *ex-situ* situations. Many variables should be considered when designing or modifying otter enclosures; there is no definitive size or design, but there is a body of experience; as well as creative ideas and options not yet attempted.

Housing and Enclosure Requirements

MINIMUM SIZE

Duplaix-Hall (1975) recommended a minimum enclosure size of 15 x 10 meters, or 150 m^2 (1614 $ft.^2$). Reuther (1991) requires a minimum exhibit size of 100 m^2 (1076 $ft.^2$) for *L. lutra* (Eurasian otter) that are sent on "loan contract" from the Otter Zentrum in Hankensbüttel, Germany. After visiting over 60 otter exhibits worldwide I agree with Duplaix-Hall's recommendation of a minimum of 150 m^2. Having said this, I will add that I have seen exhibits smaller than this that offer the otters a highly varied and enriched environment. These include Columbus Zoo and Central Park Wildlife Conservation Center; both of these facilities offer good quality land space, bushes and trees for shade, soil and natural vegetation for digging and rooting around in, as well as streams that run through the land area. However, it is suggested that all new exhibits be designed with a minimum of 150 m^2 of useable land/water surface; if larger groups are planned for space should be increased.

Although there is no definitive way to establish a species' minimum spatial requirements, the river otter is an active animal, adapted to traveling long distances [daily movements from 2.4 to 42km (1.5 – 26 miles) up to 42km for a dispersing males in one day (Melquist & Hornocker 1983)], and curious by nature. For these reasons, otters are best kept in environmentally complex exhibits where they can be offered a variety of behavioral choices to include: a long, complex water/land interface for exploration and object manipulation; a variety of substrates and vegetation; resting sites; holts or denning sites; pools; logs or other high spots for grooming and as sprainting spots; digging pits; leaf litter piles; trees, shrubs, grasses; deadfall piles, rafts, floating logs, or islands, etc. It is possible to provide these options, to a limited extent, in exhibits smaller than 150m^2 but these exhibits will prove difficult to maintain and enrich over time. Inquisitive animals will quickly become overly familiar with small, un-enriched environments; the result of this familiarity is generally, excessive sleeping, or, abnormal repetitive behaviors such as rotational swimming, pacing, and self-directed aberrant behaviors. Although a larger exhibit does not guarantee these behaviors will not occur, it does provide the animal with more behavioral options, room for exhibit-mates to interact or not-interact, and offers the management team greater enrichment and education choices.

LAND/WATER RATIO AND INTERFACE

In 1975 Duplaix-Hall set down guidelines that are frequently cited as the desired standard. These guidelines were based on an assessment of the river otter exhibits she had seen, a historical literature review, field experience with some otter species, and gut instinct (pers. com.). Her recommendations were an exhibit size as listed above, a land to water ratio of 4:1, a turf to shrub ratio of 2:3, and a containment barrier at least 1.80 meters (6 ft.) high with an 80cm (2.6 ft.) smooth overhang. These guidelines are still considered valid today. A land to water ratio of 3:1 also is acceptable if the exhibit is large. The land to water ratio is critical because the river otter actually spends more time out of the water than in the water.

- Dry land area is important because this is where the animals groom, sleep, rest, play, and eat their food.
- Pools are important because this is where otters play, hunt for food, most frequently breed, sometimes defecate, and, where our visitors expect to see them.
- The length and complexity (e.g. convoluted, interrupted with deadfall, shrubs, rock piles, trees, digging pits, etc.) land/water interface is of primary importance because this is where otters are known to spend the majority of their time.

The importance of the complexity of the land/water interface cannot be stressed enough; in the wild otters spend very little time along smooth, barren shorelines. The complex shoreline interface is where otters spend the majority of their time (~ 60%, Reuther 1991). This should consist of convoluted shorelines broken up with rock piles, deadfall, downed trees attached to pool sides, docks, etc. Well placed shrubs and trees also should be incorporated to the land/water interface with hollows designed for denning and visitor viewing. Many exhibits, large and small, can be enhanced by the placement of logs, branches, rock outcroppings, mud banks, etc. sticking out into pools.

> "*Looking at the space using behaviour of the Eurasian Otter in captivity it has to be realized that the length of the banks seems to be more important than the water-land-ratio. More than 60% of the total activity happens in an area of 1.5 – 2.0 meters* (5 ft. – 6 ½ ft.) *left and right of the water-line."* (Reuther 1991)

CONTAINMENT BARRIERS

Otters are diggers and are known to climb, therefore sinking perimeter fences/walls at least 2.6 feet (80 cm) into the ground is advisable and containment walls should be unclimbable and at least six feet (180 cm) high (temperate facilities need to take into account snow levels). If the containment barrier is chain-link fencing, it should be topped with an unclimbable overhang (Duplaix-Hall 1975, Foster-Turley 1990, Reuther 1991). Hot-wire can be used effectively but caution should be taken to ensure that animals cannot reach the hot-wire from the water.

Studies conducted on *L. lutra* at Otter Zentrum (Reuther 1991) showed that the Eurasian otter can "*...jump well leaping a distance of 130cm* (4.27 ft.) *in height when jumping from the ground to a platform, 160 cm* (5.25 ft.) *in width when jumping from one platform to another and 90 cm* (3 ft.) *in height when jumping out of the water on to a platform if there is a possibility to push off from the bottom."*

There are numerous reports of climbing otters; personally, I know of several. We should not be trying to completely prevent them from climbing, just controlling what and where. Placement of trees and the design of solid surface containment walls (gunite, rock, etc.) should be considered carefully.

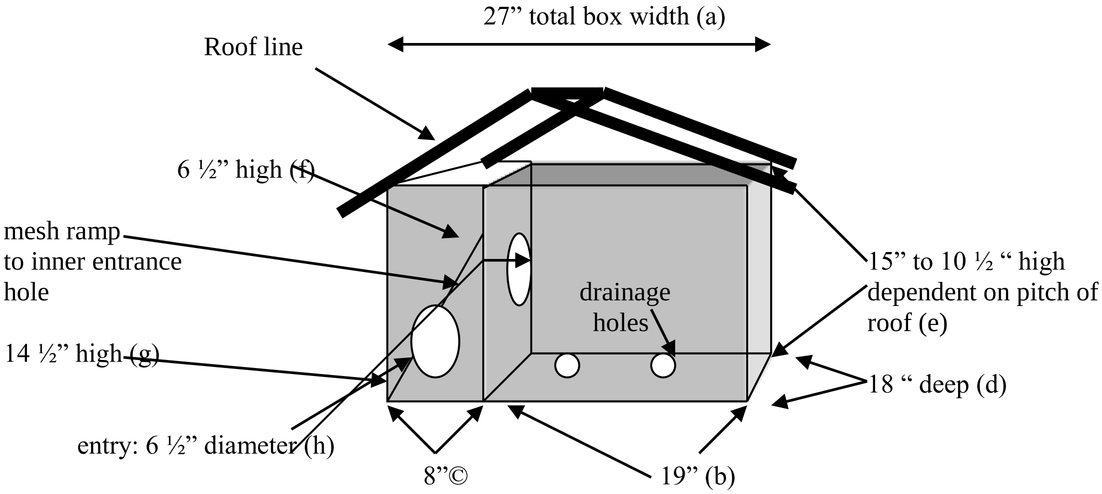
DENS/HOLTS

There should be at least a one to one ratio of dens to animals. Dens should be large enough for an adult animal to turn around and curl up in comfortably. Denning areas and/or nest boxes are important and can be made from PVC tubes, plywood, cement blocks or more natural materials such as hollow logs and 'constructed' log jams (make sure the logs are secure and will not collapse onto the animals). Photo: Pueblo Zoo on-exhibit den.

Suggested den box dimensions include: 30" x 30" x 17" (.76 m x .76 m x .43m) (Wallach & Boever 1983 citing AAZPA [AZA] Animal health Committee), and, 75 cm x 75 cm x 50 cm with a 22 cm entrance (2.46 ft. x 2.46 ft. x 1.64 ft., entrance is 8.66 in.) (Duplaix-Hall 1975)

Sample Denning Boxes
See also Section 1, Chapter 6, Pupping Boxes for additional examples and photo.

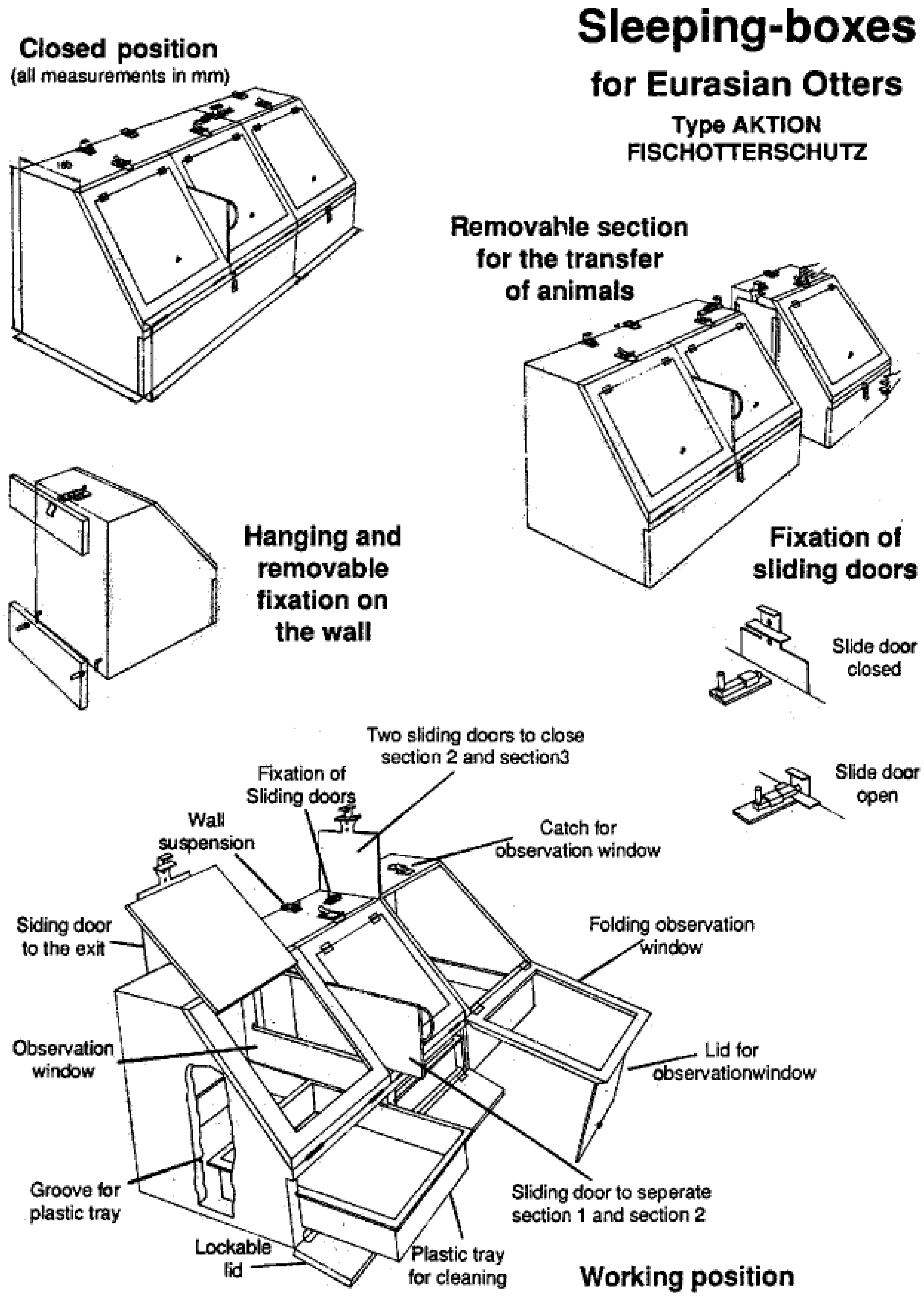
John Ball Zoo
This nest box is used off exhibit in the night quarters, especially for the female when she was due to give birth. The rest of the year this box is alternated with an air kennel and a 30” (76.2 cm) diameter heavy-rubber tub with, or without, a lid constructed out of the end portion of a soft drink barrel.



(a) 68.58 cm	(b) 48.26 cm	(c) 20.32 cm	(d) 45.72 cm	(e) 38 cm to 26.67 cm
(f) 16.5 cm	(g) 36.83 cm	(h) 16.5 cm		

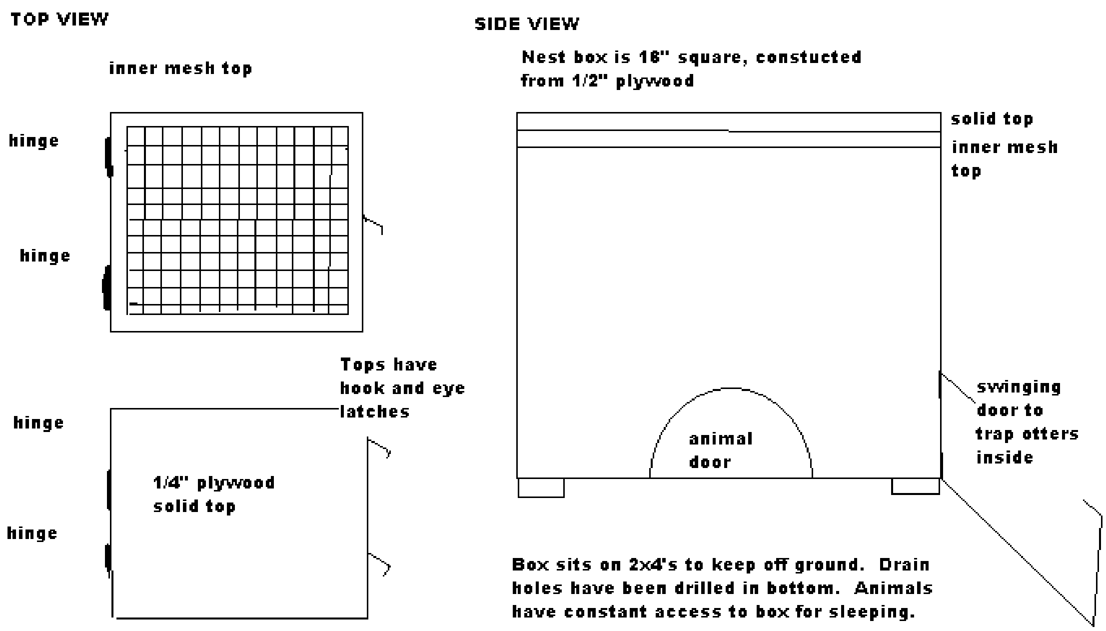
(Nest box pictured below with the top up; the top is hinged on one side and held with a hasp on the other side.)

Otter Zentrum den box, Hankensbüttel, Germany



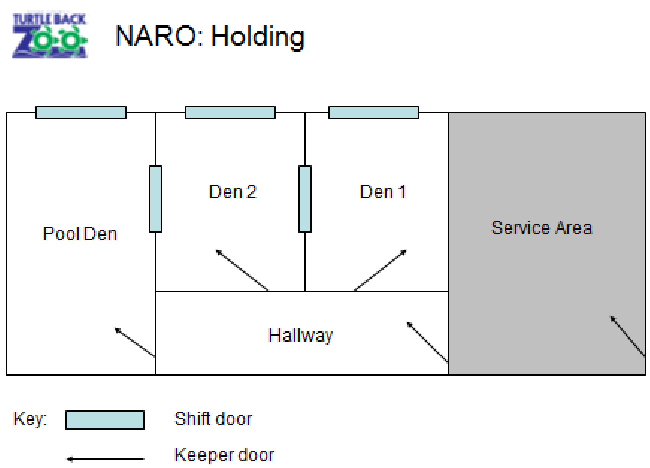
Sleeping-boxes for Eurasian Otters, Type AKTION FISCHOTTERSCHUTZ.

Stone Mountain Zoo
Contributed by: Sandy Elliot, Lead Keeper. Nest box design, allows animals to be locked in and moved.



OFF-EXHIBIT HOLDING

Night holding facilities may be used as an alternative to on-exhibit dens, if, the exhibit has sufficient hiding places to provide the animals with a sense of security and shelter. The Turtleback Zoo's diagram of their otter holding is provided as an example of good holding design. Designs should offer egress to the exhibit from all dens as well as shifting between dens. Separating walls should be solid but provide for visual introductions when necessary. Ideally, the number of individual dens should equal the number of otters you anticipate holding, plus a pupping den. Off-exhibit holding varies greatly in size based on its intended use. Whenever possible design off-exhibit holding that is satisfactory for at least short term maintenance of sick or new individuals, or animals separated for other management reasons.



If at all possible, it is preferable that animals be given access to holding at night but not locked in. Some facilities require that all animals be secured in holding at night. In these cases it is advisable that the animals be given access to: a nest box or sleeping area per individual (otters most frequently choose to sleep together but provisions must be made for social dynamics); pool; fresh drinking water separate from the swimming pool; dry bedding, and rotating furnishings and institution approved enrichment items.

Some institutions offer expanded off-exhibit outdoor areas that include pools and yards; one of these is the Pueblo Zoo, photos shown here:

Photo 1: Off-exhibit outdoor yard (70’ x 90’) with an 800 gallon semi-buried stock tank as a pool and chain link containment (buried and with flashing to prevent digging/climbing). This is an excellent idea offering multiple management options and room for managing new animals, family groups, and discordant individuals. **Photo 2**: Off-exhibit den site made from plastic culverts embedded in the hillside.

BEDDING MATERIAL

It is advisable to provide some kind of bedding material on, and off, exhibit year around, particularly when the exhibit substrate is gunite, concrete or something similar. The roughness of these surfaces can be tough on foot pads in the absence of material for the otters to dry off on. Straw, wood-wool, hay, grass, sedges, pine needles, and leaves have all been used. Bret Sellers of Oregon Zoo has a caution regarding the use of conifer shavings such as pine or fir; he says that these stripped their otters’ coats of their water repellency due to the natural turpentine found in these products. His advice is to use Beta chips made from autoclaved maple and alder chips if wood shavings are the bedding material you choose (per. com.).

HIDING PLACES

Areas where the otters can feel protected are important as resting places. These can be depressions near logs, hollow stumps, bushes, solid walls away from viewing areas, etc. When designing exhibits keep this in mind; a well-designed space should offer a sleeping area that will attract the otters, yet leave them at least in view of the public. As previously mentioned, this can be achieved by creating a depression with some sort of shade covering and providing bedding. A word of caution, otters like to build beds, they will haul material from one spot to another which may drop into the pools. The best way to counteract this is to design a bedding location back far enough from the water so that it is not an issue, or keep a good long-handled skimmer on hand!

WEATHER PROTECTION

It is important to provide shaded area for animals exhibited outside.

DRAINAGE

Proper drainage is very important. There should be enough dry area in the exhibit to allow all animals room for rubbing and drying their coats. Inside holding facilities should be provided with dry areas as well as built in pools or water tubs for swimming. Nest boxes should be provided with adequate drainage to allow bedding to stay dry.

SUBSTRATE/TOPOGRAPHY

Land to water ratio should be around 4:1 to 3:1. This ratio unfortunately is seldom the case. The majority of facilities have a land to water ratio of 3:2 or 1:1; older facilities may even be weighted the other way. This is because historically river otters were regarded as aquatic mammals instead of semi-aquatic. It is preferable to provide a variety of substrates in the exhibit area. If an exhibit is all concrete the animals will not have a suitable surface for adequate coat maintenance. Otters are diggers – while this should be kept in mind, and exhibits designed so animals cannot dig out, offering a variety of natural substrates in which the animals can dig aids in coat care and provides natural behavioral opportunities. The Pueblo Zoo has had excellent results using digging pits of soil or sand. The substrate is turned regularly prompting renewed interest by the otters due to the fresh scent and looseness of the soil. Duplaix-Hall (1975) cautioned against the use of sand substrates, unless they are of a non-abrasive quality. In particular, builder's sharp sand is dangerous because it can wear away the otter's guard hairs. It is recommended that as far as possible different substrates should be made available, to include: soil, non-abrasive sand, gravel, rocks, mulch, leaves, pine needles, grass, river rock, etc. As previously mentioned, simply turning over the soil will attract attention and provide scent, tactile, and behavioral enrichment.

SPECIAL FURNISHINGS

In addition to a pool, preferably of varying depths, logs, rocks, grass, bushes, bedding choices and a variety of substrates are all important to the maintenance of healthy otters. These items not only provide stimulation by provoking investigatory behavior, but they are important to the maintenance of a healthy coat. Water falls, sprinklers, shallow streambeds, periodic flooding of shore lines, underwater entrances to holts, log jams, stumps, complex root systems and structures, as well as islands all add variety and environmental enrichment.

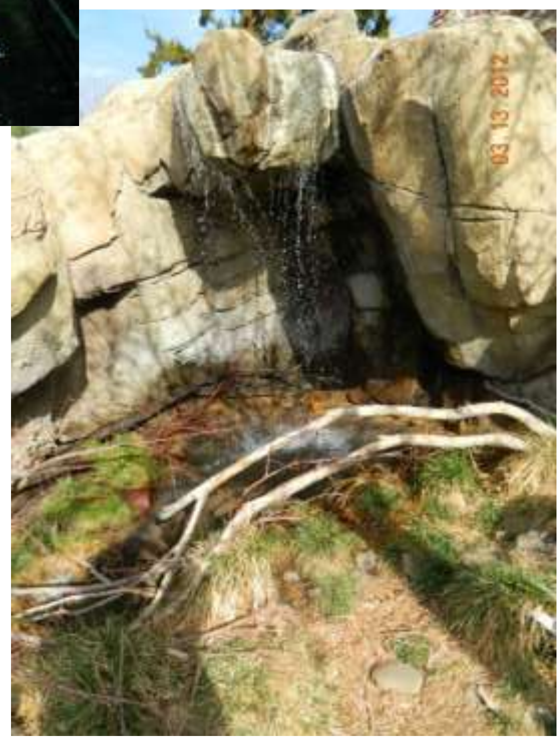
Feature Photos

Audubon Park Zoo – bridges and Jacuzzi jets (otter playing in jets)

Coyote Point Zoo - hammock

Sedgewick County Zoo – hollow logs and deadfall

Pueblo Zoo – waterfall and stream which runs into lower pool; rockwork, grasses, and deadfall



Denver Aquarium – underwater swim-through features

WATER SOURCE/TREATMENT

Fresh drinking water should always be available. Drinking containers should be cleaned daily and disinfected at least every other day.

Pool, or swimming, water should at least be monitored for chlorine level. Many facilities also monitor pH level and some monitor Coliform levels. Water may be changed daily or re-circulated through some sort of filtration system. What is important is the maintenance of potential disease causing vectors at concentrations below health hazard levels. Vacuuming of outdoor, re-circulating pools can be very effective at keeping algae growth and debris under control. The important thing to remember with otter enclosure water features is that most facilities are concerned with water clarity versus water quality. Water quality is taken care of with most water treatment systems (ozone, biological filters, sand filters, etc.) however, to provide clear water for underwater viewing can be complicated in outdoor environments. The key is the design of the filtration system in addition to placement of the pool which should be done in conjunction with experts experienced in otter aquatic exhibit systems.

What sort of water treatment system is the most effective, and safe for the animals is still subject to some debate. In general, it is this author's opinion that every method available carries with it some risk: natural flowing streams may carry pathogens or pollutants; ozone systems may be set, or designed improperly; chlorinated systems are now fairly widely accepted as detrimental to the animals' health and the water repellency of their coats; dump and fill systems are potentially expensive and may be utilizing chlorinated municipal water; filtration systems using chlorine free water may not do as good a job at keeping the water clear for underwater viewing. Whatever the solution chosen by your facility, please keep all of the information provided in this chapter in mind. At this time, the safest, and most effective systems appear to be properly designed ozone systems, de-chlorinated dump and fill, or de-chlorinated filter systems using a secondary filter medium for algae control.

Algae Control and Underwater Viewing

Underwater viewing offers a unique look at this semi-aquatic animal. Problems presented include keeping the water clear enough for viewing and algae growth. The 1st edition of the husbandry manual suggested the use of chlorine, maintained at .05ppm or less, as a means of controlling algae. Further research has uncovered concerns about the use of chlorine as an algaecide due to its potential impact on the otters' coat quality and its role as a possible carcinogen (see Chlorine). Unfortunately, some of us are still restricted to the use of chlorine. If this is the case, these precautions should be taken: 1) Chlorine should be added only when the animals are not in the pool. 2) Animals should not be allowed back into the exhibit until the chlorine level is lower than .05ppm – this level is ARBITRARY, future research may reveal this to be too high, or it may be safe at slightly higher levels. The best policy is to monitor your otters' coats water repellency quality closely. 3) Research alternate methods of algae control that will work in your institution. 4) The addition of sodium thiosulfate will neutralize the chlorine. (Photo: Graham Jones, Columbus Zoo)

Note: Boness (1996) raises questions regarding the effectiveness of using chlorine as a disinfectant (particularly for true aquatic species which the otter is not) if the breakpoint chlorination approach is not used. To use this approach effectively in a pool where ammonia may be continuously added via animal urines it is necessary to continuously monitor chlorine levels.

"Breakpoint chlorination is generally believed to be the most effective technique of chlorine disinfection. When chlorine is first added to a system, it reacts with ammonia and nitrogenous waste to form combined chlorines. With time and increased chlorination, the ammonia levels decline, so that the addition of more chlorine results in the formation of a free chlorine residual. The point at which the combined chlorine residual is at a low is called the breakpoint. Breakpoint curves are unique to each water system because they are dependent on the initial concentrations of nitrogenous material and other oxidizable substances in the water.

Breakpoint chlorination is possible in an aquatic mammal facility, but, because ammonia is added continuously via the animals' waste products and because the animals are in the water all the time, careful monitoring of chlorine levels is required to stay beyond the breakpoint once it is achieved. The relatively high chlorine residuals that might be required to stay at breakpoint should not be a problem because free chlorine appears not to be toxic, but if breakpoint is lost and these high residuals become mainly combined chlorine, one has a serious problem. Depending on the concentration of nitrogenous material added to the pool on a daily basis (a direct function of the number of animals in the pool), whether or not there are precursors of trihalomethanes present, and the bacterial load of the system, it may be more feasible in an aquatic mammal exhibit just to use a low level of combined chlorines to keep bacteria levels down.

Chlorine should always be administered to the pool through a high-quality injection system. Manually adding any type of chlorine is unsafe and does not properly distribute the chemical in the pool….Because of the known and probably negative effects of chlorine on animal health, alternatives should be considered."

Chlorine

"Chlorine is a very active oxidizing agent, and it readily reacts with ammonia and other nitrogenous materials to form chloramines, or combined chlorines.

"Free chlorines are much more effective disinfectants than combined chlorines….Combined chlorines are more toxic than free chlorines, but free chlorines used in the presence of humic and fulvic acid or some algae can produce carcinogenic trihalomethanes. And, even though chlorine is a good bactericide, many protozoans, yeasts, cysts, and viruses are resistant to it." (Boness 1996)

There is a great deal of debate over the use of chlorine as an algaecide. However, after completing my own, simple water tests and observations, questioning over 50 institutions worldwide and speaking with several water quality experts, I have come to the conclusion that chlorine levels of .5ppm are potentially harmful and levels above this have a definite impact on the water repellency of otters' coats. I have left a recommendation of .05ppm if chlorine is used or present in the water supply however, all facilities are strongly urged to explore other methods of algae control and water treatment.

Additionally, LaBonne (per. com.), Boness (1996), Oliver (1980), Briley et al. (1980) raise questions about the presence of Trihalomethane, a chlorinated organic compound and *"…a volatile substance…"* (LaBonne per. com.) that is a known carcinogen in aquatic mammal pools. *"The concentration will depend upon how much dissolved organic material is available, the concentration of chlorine (how much is free and how much is combined), the water temperature, and the filtration equipment (copper, ozone, biofilter, sand filter, etc.)."* (LaBonne per. com.)

Barley Straw

Some institutions have had success controlling algae growth using barley straw. It is available commercially packaged specifically for use in streams and ponds (example: Aquatic Ecosystems, Inc.; http://www.aquaticeco.com/subcategories/2786/Barley-Straw). Aquatic Ecosystems claims a ½ lb. bag per 1,000 gallons of water works up to 6 months when placed where the water can flow over it in a pond or in the sump or filter. Research on how, and if, barley straw effectively controls algae is inconclusive with results perhaps being dependent on the size of the water body. The activity of barley straw is typically regarded as being algistatic (prevents new growth of algae) versus algicidal (kills existing algae) (Lembi 2002).

HUMIDITY
Due to the semi-aquatic nature of these animals, humidity should not be a problem in properly designed exhibits. Lack of sufficient land area for drying-off on is more often the problem. A relative humidity of 30 – 70% is recommended for mustelids in general.

LIGHTING REQUIREMENTS
Animals not housed out of doors year around, or a full 24 hours a day, should be provided with a varying photoperiod which can be easily set up with timers. Indoor facilities should provide full spectrum lighting, if possible, in addition to the varying photoperiod.

TEMPERATURE
Indoor facilities should be kept at an ambient temperature below 70 - 75ºF (15.5 - 18º C) (Wallach & Boever 1983). Animals housed indoors should be provided with a thermal gradient within the exhibit. This will allow for the selection of a comfortable temperature by each individual animal (Moore, unpubl.). Obviously, the temperature of outdoor facilities cannot be controlled however, shade in the warm months and shelter from inclement weather in the cold months is very important. Dry bedding material should always be provided in, or near, the otters' denning facilities.

VENTILATION
"*Indoor exhibits should have negative air pressure of 5 – 10 air changes per hour of non-recirculated air. Separate ventilation systems should be provided between exhibit and visitor areas to reduce air (and odor) transmission, and potential disease transmission, between humans and animals"* (Moore, unpubl.).

CAPTURE AND HANDLING
Many facilities are training their animals for routine husbandry procedures; target training can be very useful in reducing stress on the animal and the keeper staff. See Training Section for information on training for hand injections which is the preferred method of administering vaccines/anesthetics. These squeeze/transfer cages are best used if the otters are trained to enter them calmly and willingly.

McCullough et. al. (1986) describes a squeeze box they used on otters. Serfass (1994) gives the design of a transport tube used by the Pennsylvania State Wildlife Agency. Reuther (1991) includes the design of a squeeze cage used at Otter Zentrum in Hankensbüttel, Germany. Air Kennels (or similar), squeezes designed in-house, and smaller sized squeeze cages are probably were the most frequently used methods of capturing and containing otters in zoos; currently most facilities train their otters to willingly enter a containment box or squeeze. Other capture methods include nets or catch-poles. Due to their loose skin, it is not advisable to hand catch these animals; it could lead to keeper injury and undue stress on the animal. If it is necessary to hand hold an otter, gloves should be worn. For anesthetizing information see the Health Care section.

Sample Capture boxes, Training chutes, and Squeeze Cages

Oregon Zoo Squeeze cage (LGL Animal Care Products, In., College Station, TX; http://www.lglacp.com/transfer_restraint_cages.htm) used for weighing as well as transport

Dickerson Park Zoo – injection/physical inspection chute

National Zoo (made in-house)
Dimensions: 34” long, 18” high, 18” deep. The length of the bars that lift up are 21” across.

Cleaning and Waste Removal

Food bowls and feeding stations should be disinfected daily; water bowls should be cleaned daily and disinfected at least every other day. Enclosures and holding facilities should be cleaned daily and disinfected as necessary. Do not disinfect every den, or the entire exhibit, at the same time, (it is preferable to leave the animals' scent on something); disinfecting of dens and sprainting sites should be done as necessary. Because scent is important to this species, nest boxes and exhibit furniture should not be cleaned as frequently as other surfaces. When these items are cleaned do not do all of them on the same day. In the AZA's Minimum Exhibit Guidelines (1997), Moore suggests not more than 25% be cleaned at any given time. Soiled or wet bedding should be removed and replaced daily. Pools should be kept free of accumulated feces or discarded food. Waste and trash should be removed in a timely fashion to minimize odor, disease hazards and pest infestation. The enclosure design should facilitate the drainage of excess and/or cleaning water.

Enclosures Examples

Otter enclosures range in size and cost. To illustrate this I have selected two outstanding examples that represent different positions on the cost spectrum however, both provide more than the minimum suggested enclosure space and make excellent use of space, complexity, natural substrates, and holding options.

Pueblo Zoo

Exhibit

Square footage: 2,250 cubic feet; 60% land to 40% water; 8600 cubic feet of water
Enclosed in "Rock wall" that leans inward for containment

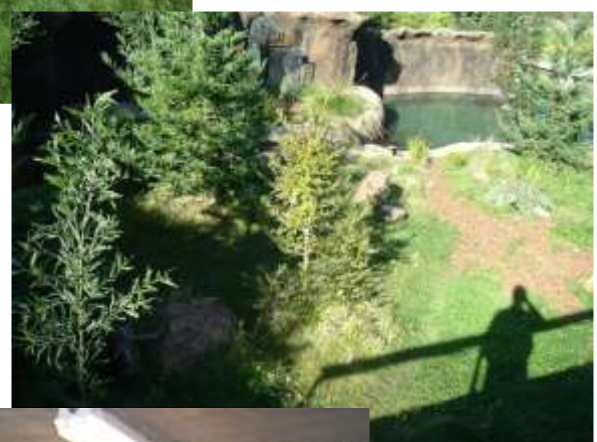
Pueblo Zoo built this exhibit approximately 9 years ago. It was designed to allow for breeding and management of two groups if required. To maintain the vegetation staff periodically replants plugs and reseeds grasses. The otters have created trails through the grasses and make good use of two digging pits (sand and soil) that are frequently dug out or turned over (serving to attract the otters' attention with new smells and fresh soil). Daily enrichment is supplemented by novel feeders and scatter feeds for portions of the diet, as well as training (see enrichment and training chapters). Otters are given access to the exhibit, off-exhibit enclosure, and holding dens overnight (as one group or two during pre-breeding separation). (Photos: Jan Reed-Smith)

Off Exhibit Enclosure
Space- 70’x 90’
Water source is an 800 gallon semi-buried stock tank. The perimeter is chain link fencing with the bottom few feet buried at an inward angle. There is a sheet metal flashing approximately two feet off the ground to prevent climbing. Enclosure dens created from partially buried plastic culverts

Holding
The holding space was intentionally built smallish to be used for maternal denning and temporary holding only. Holding dens are each provided with tough rubber bins (holes drilled in the bottoms for drainage) and/or rubber tubs. Bedding used includes straw, shredded paper, cardboard chips, and materials brought in by the otters. The room is 15’X 15’; Smallest den: 3’x 5’; Largest den: 5’x 5’; 4 Dens total. Two shift doors to exhibit; 2 shift doors to patio leading to off-exhibit enclosure. Access to both sides (exhibit and off-exhibit yards) is through squeeze cage. Otters are not locked inside the holding building overnight but given access to both, or at least one outdoor enclosure. Photos: M. Pocock, one side of holding dens and back patio/yard.

OAKLAND ZOO

Exhibit: Approximately 3000 $ft.^2$ of natural substrate (grass and soil) with trees, bushes, rocks, branches, and two 30,000 gallon pools. The holding consists of four stalls (10 x 10 ft.) provided with fire-hose hammocks and at least one Vari Kennel; igloos also are used as an option. One of these stalls is a sound-proof whelping den with an attached den box (this stall has always been selected by the female for giving birth; den box is similar to Otter Zentrum example). Each stall has a 50 gallon, above ground pool. The otters are allowed access to the enclosure and holding dens at night. (Photos: Andrea Dougall)



Holding dens

Animal Management

IDENTIFICATION OF INDIVIDUAL ANIMALS

Generally each animal has a slightly different shaped rhinarium (nose pad); this can be used to identify some individuals visually from the outside of the exhibit. Many otters also have some sort of spot pattern on the upper lip area. Scott Shannon (personal communication) used these “moustachial maculations” to identify individuals in the otter population he studied for many years in Northern California. Behavioral cues and coat color variations also may prove to be useful identifiers. However, all of these methods require patience, experience, and familiarity with the animals. Temporary pup identification can be achieved by the clipping of a small patch of hair in a different location for each pup.

Permanent identification should be done in a manner consistent with the holding institution’s policy. Options include: transponders, tattooing on the inside of a hind leg, or tattooing on the interdigital membrane of two hind toes a method used by Melquist & Hornocker (1983). See Health Care for transponder placement information.

PHYSIOLOGICAL AND BEHAVIORAL INDICATORS OF SOCIAL STRESS, HARASSMENT, ILLNESS, ETC.

If an animal is being harassed by exhibit mates it may show some of the following symptoms: wounds, stereotyped or abnormal repetitive behaviors, hiding, self-mutilation, loss of appetite, poor coat condition, hair loss, screaming. Because these also can be indicators of illness it is sometimes difficult to determine the true cause. If an animal shows any of these symptoms an environmental/exhibit-mate problem should be thoroughly checked out. This may be difficult to do and could necessitate 24 hour observations. Once the problem has been identified immediate steps should be taken; if one animal is harassing another the harasser should be temporarily removed. Re-introduction of this animal should be done slowly and with close supervision. Causes of hair loss also can be very difficult to determine; in addition to considering illness or parasites in cases of hair loss, over-grooming, hair plucking, and limb or body sucking are all factors that need to be considered. See Exhibit-mate Aggression.

FEEDING

Due to their high metabolic rate and rapid digestion (Iversen 1972, Toweill & Tabor 1982, Estes 1989, Davis et. al. 1992, Kruuk 1995, Spelman et al. 1997) otters should be fed at least twice a day, three, or more, is preferable. This prevents consumption of spoiled food, accommodates their rapid digestion of food as well as their high metabolism and can stimulate increased activity. Note: The consumption of spoiled food can lead to enteric problems, something otters are very susceptible to. See Section 3 Enrichment, Abnormal Repetitive Behaviors (ARBs or stereotypies) for information on use of feeding cues and their possible mitigation of pre-feeding related ARBs associated with more frequent feedings and training sessions.

Duplaix-Hall (1975) found that in the wild, river otters rarely ate more than 500 grams of food at a time but that they would eat approximately 20% of their own body weight daily. See Diet/Nutrition.

Weight

The ideal weight for all animals will vary and should be established on an individual basis. Subcutaneous fat is not widely distributed but is located primarily at the base of the tail and caudally on the rear legs. Smaller deposits are located in the axillary regions and around the external genitalia” (Baitchman & Kollias 2000). AZA Otter SSP Nutritional Advisors and individual institutions have been working on a weight matrix. The following photos are provided as a guideline:

Thin (Photo: Gary Woodburn, wild otter)

Good condition (Photo: Dave Mellenbruch, wild otter)

Heavy (Photo: Jan Reed-Smith, captive otter)

New Animal Introduction and reintroductions

Introduction of new animals can take anywhere from one day to several months; it just depends on the animals. It is advisable to plan on proceeding slowly via sight and smell first, gradually building to a physical introduction. The animals will generally give you behavioral cues as to what their reaction to a physical introduction will be.

It is possible to introduce adults, some of these introductions work, some do not. Two factors appear to be very important; 1) Both animals have a place to get away, 2) Introductions take place in neutral territory. An alternate, but slightly less preferable method is to introduce them in the home turf of the animal likely to be the more submissive (personal observation & K. Butkiewicz pers. com.).

Problem introductions should be done slowly, back up a few steps if need be. Be aware that some animals will just never get along (especially females) and an introduction may have to be abandoned. The reverse of this also is true, some animals hit it off right away and an introduction can be accomplished in just a few days, or in some cases, hours. Several facilities have had problems reintroducing an animal after an extended absence from the exhibit due to illness. Again, this usually occurs with females and these reintroductions may need to be treated as if the animals were meeting for the first time. An introduction plan should be laid out in advance, for example:

- ✓ Where; holding or outdoor enclosure. If at all possible do intros where an animal cannot become cornered.
- ✓ When; preferably before opening, and when there is no external stimuli such as construction or grounds cleaning, etc.
- ✓ Who will be there (staff, supervisor, veterinarian, volunteers).
- ✓ What you will need to separate animals in case of aggression (e.g. baffle boards, hose, net, catch pole, etc.).
- ✓ When early introductions will be ended; preferable to end when animals are getting along or ignoring one another.
- ✓ How and who will handle extended introduction periods. Will they be monitored, for how long, and by whom?

- ➢ Phase 1: Introduce new animal to the facility alone (holding and exhibit) and, exchange scents between otters.
- ➢ Phase 2: Allow visual access without possibility of physical interaction (i.e. side-by-side dens with small mesh wire). Otters also will be able to smell and hear one another. Monitor for affiliative or agonistic behavioral clues.
- ➢ Phase 3: Allow brief physical introductions which should be continuously monitored.

Affiliative grooming (Photo; Montreal Biodome)

- ➢ Phase 4: Increase length of physical introductions with continuous monitoring gradually switching to periodic monitoring.
- ➢ Phase 5: Once staff is confident the otters are compatible they can be left together unmonitored or overnight.

Introducing Females to Females

Introducing adult females to unfamiliar adult females can be difficult, if not impossible. There also have been reports of previously compatible females becoming aggressive to one another after extended separations. Having said this, in the years since publication of the last Husbandry Notebook edition several institutions have had great success introducing females, particularly younger females to old females. These introductions should be planned in advance with well-defined roles for the staff and emergency procedures identified in case of aggression (females have been known to try and drown newly introduced otters, both sexually immature males and other females). This introduction approach can be used for all otters, regardless of sex; it is just that male/male and male/female introductions may go more quickly. It always should be remembered that some introductions may never be successful and the most important key is a staff who knows the animals involved.

M. Rabon and C. Zewe, Houston Zoo's John P. McGovern Children's Zoo (2010) reported on their introduction process which was based on experience gathered from other successful institutions. An adapted version of their plan outline for their introduction approach is provided on the following page.

Female/Female River Otter Introductions –Approach Outline

Phase One – In Quarantine (30 Days):

- Have staff develop a relationship with the new otter while in quarantine by visiting at end of the day. Start training the new otter husbandry behaviors.
- Introduce new otter to resident female's scent by bringing bedding or used enrichment items to quarantine (with quarantine supervisor's approval).
- Begin, or continue, training sessions with resident otter; particularly recall and station. In this situation training sessions were to occur on exhibit.
- Add smaller mesh covering to exhibit holding dens, if required, to prevent injury to paws, etc. during introduction howdy sessions. Install video cameras for future monitoring when otters are left alone.
- Ensure otters can be separated in holding during introductions if required. Modify dens if required.

Phase Two – Howdy Time (2 – 4 weeks):

- Move new otter to holding. Leave resident female on exhibit for a few days to allow the new otter time to adjust to holding.
- Start howdy process by alternating otters on exhibit during the day. Leave one otter outside at night and one in holding.
- Start short, supervised howdy sessions in side-by-side dens.
- Continue training begun in quarantine. Begin on-exhibit training with new otter.

Phase Three – Introduction (one week to months):

- Introductions should first take place in holding if possible; this requires a set-up where an otter cannot be trapped by an aggressing animal. If this is not possible in holding then animals should be given access to exhibit and holding. Introduction area should be provided with visual barriers, escape routes, non-food enrichment.
- First introduction should take place when zoo is not open to the public and area disturbances are minimized (i.e. no grounds cleaning, construction, etc.). Observers should be limited to staff involved in introductions, with trainers ready to enter the exhibit if required.
- Have all necessary intervention equipment ready and available, such as hose, net, baffle board, CO^2 fire extinguisher, air horn, crate, etc. Have a plan identified as to which intervention should be used first, by whom, and under what circumstances.
- Begin with short introduction sessions; stop before aggression occurs if possible. Gradually increase session length based on otters' behavior. These sessions should be continuously observed by trained staff. Once otters are showing signs of getting along or non-aggressive avoidance volunteers can be used with qualified staff nearby.
- Continue to separate at night until the otters' behavior indicates they are past aggressive interaction.
- If otters are left together overnight video camera tapes should be reviewed for aggression, submissive, stress-related, or avoidance behaviors.

Phase Four – Success

- All successful introductions should be monitored by well-trained staff for any changes in individual otter behaviors, stress, and/or dominance.

Introducing Male to Family Group

In the wild males do not participate in pup rearing. However, in captive settings they are routinely reintroduced to the female and pups. The concern for when this should occur is primarily based on the female's acceptance of the male. No incidences of the male attacking the pups have been reported. The suggested process is as follows:

- Pups should be swimming well before the male is introduced to the family group (intros typically at 3 – 6 months of age).
- If the male has been moved to another exhibit he should be returned to the otter enclosure and rotated onto the exhibit separately from the family group for several days to allow the female to become accustomed to his presence and scent in the enclosure and holding.
- If the female appears comfortable and unconcerned about his presence they should be allowed visual, but not physical access in holding. Staff familiar with both animals should assess the female's reaction; if she appears comfortable or exhibits affiliative behavior towards the male introduction can be attempted.
- Introduction of the male to the female, without the pups present, should be attempted first. This can occur in holding or by giving the male access to the exhibit and opening the female's den door. This will allow her to enter the enclosure and interact with the male yet return to the den and pups if she wants.
- If she behaves aggressively the introduction should be stopped and tried again ~ a week.
- Once the pair demonstrates no aggression between them the male can be introduced to the pups. Typically, if the female is comfortable with the male's presence the male/pup introduction goes smoothly.

Exhibit-mate Aggression

Aggression between otters can occur. Often it is brief and results in a loud squabble with little actual fighting. Behavior such as this should be noted but typically is not of great concern unless it occurs with increasing frequency or intensity. Aggression which leads to physical fighting, and at times attempted drowning generally only occurs during initial introductions, if an otter has been removed for an extended period, or during breeding season if multiple males are in the exhibit or the female is not interested.

It appears that more actual fighting has occurred between female/female cage-mates than male/male or male/female pairings. Mixed-sex groups (2.2 or more) are exhibited, but, they may require a long introduction period particularly if the animals are adults. In mixed-sex groups with multiple males aggression may increase during breeding season and should be monitored for. Males are frequently reintroduced to family groups, the time frame ranges from 3 to 6 months. Males left in an exhibit with a lactating female should be provided with enough room to stay out of her line of sight until she decides he may approach the pups. See Behavioral Indicators of Social Stress.

WHAT SORT OF GROUP TO EXHIBIT

Otters in the Wild

Most frequently zoos exhibit a pair, this is fine, and will probably continue to be the norm, but is not believed to be the typical social grouping found in the wild.

Typically, female otters are found alone (Blundell et. al. 2000), or, with: 1) pup(s); 2) offspring from the previous year (probably female, but it is not known for sure); 3) pup(s) and a "helper otter" which is generally believed to be a female (Rock et. al. 1994, Blundell 1999 & Blundell et. al. 2000), or 4) with a male (Blundell et. al. 2000) during the breeding season.

Blundell (2000 and pers. com.) has found females joining groups of males in marine environments for some part of the year. Most males are found, by themselves or in a group of males; less frequently, but observed, they are found with a female or with a female and pups (Blundell et. al. 2000). It is difficult to observe otters in the wild so it has been hard to determine the otters' social history. However, recent, and on-going studies, are showing the river otter, particularly males, to be more adaptable socially than previously thought.

All Male Groupings

All male groups do very well. The Virginia Marine Museum exhibited a group of five males and researchers at the University of Alaska, Fairbanks housed 15 males together without any problems. (Harshaw per. com., Ben David per. com.) There are frequent reports from the wild of all male groups (for example: Blundell 1999 pers. com., Shannon 1999 pers. com., Melquist & Hornocker 1983, Blundell et. al. 2000), in fact, all male groups containing up to 20 individuals have been seen.

Male/Female Pair

As stated earlier, this is the most frequently seen captive grouping. Although pair living is not the normal social structure for otters in the wild they seem to adapt to living in pairs in *ex-situ* situations. An option to explore, even if a facility is not interested in breeding their river otters, is rotating the pair through the exhibit. The male can go out for a while, then the pair, then just the female or any variation. This mimics their natural social state a little more closely, will help stimulate activity in the exhibit, and provide an enriched behavioral environment for them. (Photo: Haley Anderson)

Multiple Pairs

Facilities have kept multiple pairs in three ways: all together (Lowry Park Zoo & Nashville Zoo @ Grassmere), males and females separately (Minnesota Zoo, St. Louis Zoo), or, separate pairs (Little Rock Zoo, St. Louis Zoo). See Section 1, Chapter 3, page 21 for groupings currently held in AZA institutions.

All Female Groupings

In a zoo, it is generally wiser to not exhibit multiple females together unless they are related or introduced at a very young age. Several facilities that have been successful with multi-female groups have had problems arise when one animal has been reintroduced after a brief absence. However, as with everything, there are exceptions. Some multi-female groups get along for years (the individual relationship history of many of these groups is unknown, this is not true for all), and have no trouble adjusting after brief separations. Related females kept together tend to do well over the long term.

Multiple Female-Single Male

Knoxville has been successful with 1.2 (females came from the same source, same age, same acquisition date but it is not known if they are related). These animals are together 24 hours a day. There are a few other facilities that have housed multiple females with a single male. If this group structure is selected you should watch for signs of stress because, the females may fight with each other or team up against the male.

It is possible to house 1.2 but exhibit the females separately from the male by rotating them through the exhibit. If this method is chosen adequate off-exhibit holding should be provided. The male can then be introduced to the females during the breeding season if desired. If space allows and this rotational method is chosen it may be advisable to house 2.2 to allow for mate choice. See female introduction protocol.

Multiple Male-Single Female

There is not a good reason to exhibit multiple males and one female unless something happens to one of your animals. If this is the case, the group should be monitored closely to ensure the female is not being traumatized by too much attention from the males. If possible, rotating animals through the exhibit is recommended. This could include different pairings such as 2.0, 1.1, 0.1, and the other 1.1 pairing.

Family Groups

Males are often reintroduced to a female and pups with great success. If the male is reintroduced the pups should be swimming well and the pair should be introduced on their own to determine if the female will accept the male. When pups sexually mature they should be sent to another facility.

GERIATRIC/IMPAIRED OTTERS

Modifications to the exhibit and holding may make it easier for older otters to manage their environment. These could include ramps, steps, softer substrates, “beds”, hammocks, etc. Chondroprotective agents can be tried and pain should be managed. It is particularly important that the weight of older otters be appropriately managed and that annual physical exams are performed to monitor bloodwork, dental condition, radiographs, etc. (G. Myers per. com.)

MIXED-SPECIES EXHIBITS

North American river otters are not routinely kept with other species. I have heard rumors of a Canadian facility housing them with raccoons and/or fox but have not been able to confirm this. They also have been kept with sea lions and in one case with beavers; how long these mixed-groupings endured is unknown. There have probably been other mixed species groupings, both successful and unsuccessful but I have not been able to find any further information.

RECORD KEEPING

In general, most facilities have record keeping procedures in place. Information on an animal’s behavior, training, enrichment, food intake, weight, health, reproductive status, vaccinations, etc. should be maintained to facilitate its proper care. All facilities should participate with the AZA North American River Otter Studbook Program (D. Hamilton, DHamilton@monroecounty.gov). The AZA Otter Care Manual has more detail on record keeping practices for member institutions.

SHIPPING

The IATA regulations are subject to constant review, so the current Live Animals Regulations Volume at the time of any animal shipment should be consulted. This information is provided as a guideline only. The sample crate below and all information contained in this section come from the International Air Transportation Association (IATA) Live Animals Regulations (LAR), 29th Edition, 2002, page 299-302.

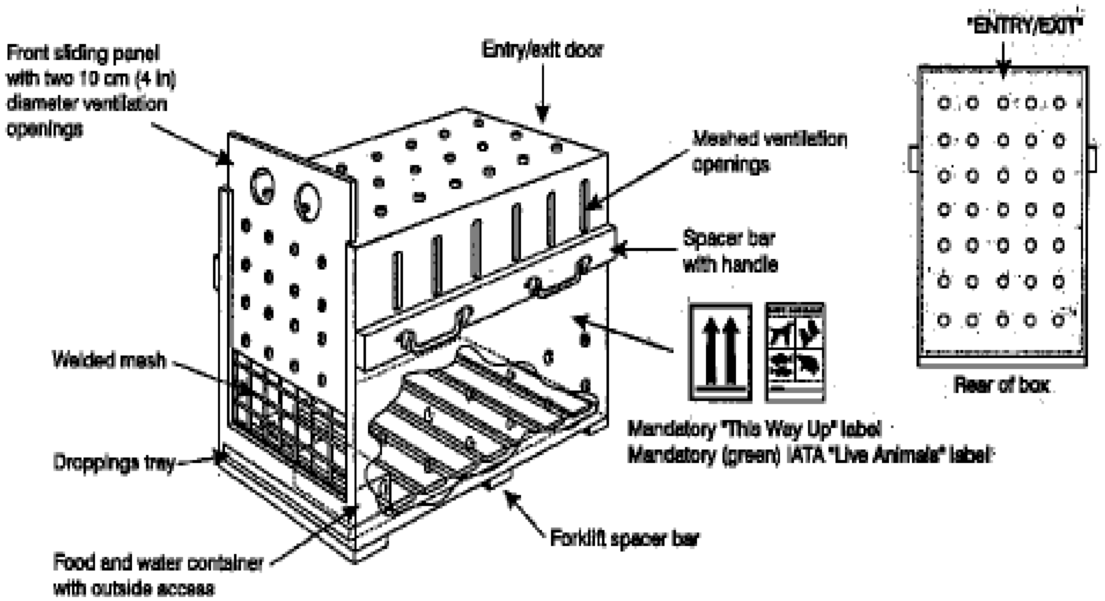
> *“The height of the container must allow the animal to stand in a natural position with its head extended and the width must permit it to turn around and lie down comfortably. The actual measurements will vary with the species involved.*
>
> *“The frame must be made from solid wood or metal parts bolted or screwed together. It must be constructed so that it cannot be damaged from continual biting or scratching at the corners. If the total weight of the container plus the animal exceeds 60kg (132 lb.) metal bracing must be added to the frame.*
>
> *“The sides and door must be made of metal or solid wood. The front of the container must be constructed of weld mesh. The mesh must have a diameter that will prevent the animal protruding its nose or paws to the outside. The whole front must be covered by a sliding shutter which can be raised and lowered to permit feeding and watering. It must have two observation holes of at least 10 cm (4 in.) in the upper part and ventilation holes, with a minimum diameter of 2.5 cm (1 in.), spread over the remainder of the surface in order to give good ventilation but at the same time leave the animal in semi-darkness.*

"The floor must be slatted, over a leak-proof droppings tray.

"The roof must be solid wood or metal with ventilation openings over its surface.

"A sliding door must be provided, it can be made from the weld meshed ventilation front if required. It must have a secure means of fastening so that it cannot be opened accidentally.

EXAMPLE:



Container Requirements 82 apply to otters.

"The main ventilation front must be supplemented by meshed openings along the upper part of the container walls and/or holes with a minimum diameter of 2.5 cm (1 in.) spread over the top third of the sides and the whole of the back and top. These holes must be spaced both horizontally and vertically at intervals of approximately 10 cm (4 in.) center to center. It is essential that there is some ventilation provided in the lower third of the sides for the removal of harmful waste gases.

"The total ventilated area must be at least 20% of the total area of the surface of all four sides. More ventilation and the use of larger meshed openings is permitted but the animal must not be able to protrude its nose or paws to the outside from any opening.

"If the mesh is fixed to the interior of the container all sharp edges must be protected.

"Spacer Bars/Handles must be made to a depth of 2.5 cm (1 in.), must be present on the sides of the container as shown in the illustration. (See illustration)

"Food and water containers must be provided with a means of access from the outside.

"Forklift spacers must be provided if the total weight of the container plus the animal exceeds 60 kg (132 lb.) (IATA Live Animals Regulations 26th Edition, p296)

Regulations have obviously become fairly rigid. Hard plastic pet containers can be used 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 of the larger containers must have secure fastenings at the top and the bottom;*
- *"A curtain, that can be raised and lowered and does not impede 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 made in order that the total ventilation area is at least 20% of the four sides;*
- *" Food and water containers must be fixed inside with access from the outside;*
- *"The container must be correctly labeled."*

When shipping an animal, especially across international borders, check to see what types of special requirements/restrictions may be in place. For example, some countries will not accept crates in which straw has been used as a bedding material.

All animals should be shipped one to a crate. Lactating mothers should not be shipped.

Sample Shipping Crate

Photos: Boonshoft Museum

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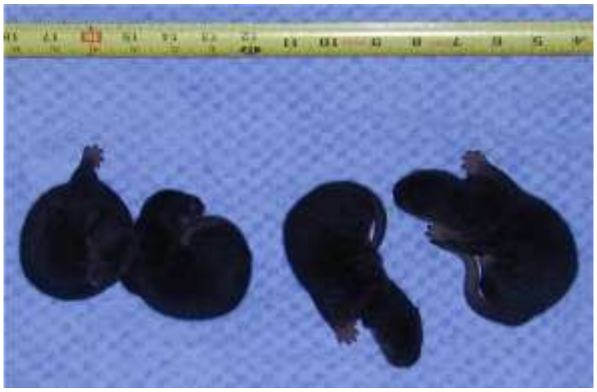
CHAPTER 8 Hand-rearing

General Information



See Health Care Section for vaccination information and Section 3 Rehabilitation of Otters for additional details on hand-rearing pups. Although mothers may successfully give birth, there are times when they are not able to properly care for their offspring, both in the wild and in *ex-situ* populations. Fortunately, animal care staff should be able to assist with the rearing of these offspring if necessary.

(Photo: Florida Aquarium pups, day 1, P. Blum)

Hand-rearing may be necessary for a variety of reasons: rejection by the parents, ill health of the mother, or weakness of the offspring. Careful consideration should be given as hand-rearing requires a great deal of time and commitment (Muir 2003). Before the decision to hand-rear is made, the potential for undesirable behavioral problems in a hand-reared adult should be carefully weighed (e.g., excessive aggression towards humans (rare in most otters), inappropriate species-specific behavior, etc.) and plans made to minimize deleterious effects on the development of natural behaviors as far as possible. This will require an extensive time commitment on the part of staff. Things that should be considered include: fostering or relocation of the young to another facility that has pups of a similar age, exposure to species-specific sounds, teaching the pups to swim, companionship for singletons, 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. There is a 'Neonatal Examination and Monitoring Protocol' provided later in this section. Offspring that are not receiving milk will be restless, possibly calling continuously, may be hypothermic, and scattered around the enclosure. Another indicator of trouble would be the female moving around the exhibit continuously while carrying the young; this could mean she is not comfortable with the denning provided, or there is a problem with her or the pups (Muir 2003). If it is necessary to remove offspring because of an exceptionally large litter, it is best to remove two of the largest pups. The temptation is often to take the smallest, but they stand the best chance if raised by their mother. Hand-rearing of singletons is more likely to lead to severe imprinting on humans than if they have a conspecific to play with (Muir 2003). The AZA Otter SSP recommends that singleton pups being hand-reared be placed together, if at all possible. To date, fostering has been attempted once

with otter pups and was successful. A pup was taken from a female with no milk and sent to another facility where their female was already nursing pups (Columbus Zoo and Beardsley Zoo). In these cases, the AZA Otter SSP management team should be consulted first. Columbus Zoo also has been successful at stimulating milk production in a female and placing pups on teats for first nursing (Photo previous page: Columbus Zoo). Young otters removed for hand-rearing should not routinely be reintroduced to the parents with an expectation of acceptance. Introductions of hand-reared animals should follow procedures specified in the standard introduction protocol.

PUP DEVELOPMENT

The following information provides a summary of pup development. Consult Section 1, Chapter 6 Reproduction and Section 3, Chapter 13 Rehabilitation for additional information.

- 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 begin erupting at about 12 days
- Eyes fully open at 28-35 days
- Eyes focus ~50 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, range 38 to 70 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, may start as early as 75 days

SWIMMING, TERRESTRIAL ACTIVITIES, AND BEHAVIORAL STIMULATION

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

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

Physical Care Protocol

Incubators provide the best source of warmth. Heat lamps are too intense and can be dehydrating. In an emergency, hot water bottles wrapped in a towel may be placed in a box with the pups nestled next to it, or they can be warmed slowly by placing them next to your body (Muir 2003). Pups may feel more secure if wrapped in layers of towels; this also aids in keeping them warm (Muir 2003). Pups should be dried after feeding/bathing to prevent hypothermia until they are proficient at self-grooming. The normal body temperature for pups is unknown (for adults: 38.1 – 38.7°C (100.6 to 101.7°F); baseline = 38.4°C (101.1°F) (Spelman 1999), but the animal should feel warm to the touch.

Altricial young are unable to self-regulate their body temperature during the early postnatal period and require an external source of warmth. If an incubator is not used, it may be necessary to place a heating pad, set on low, under the housing container until the pups are able to thermo regulate. Meier (1986) and

Wallach & Boever (1983) recommend 29.4-32.2°C (85-90°F) and 50-60% humidity as the desired incubator setting for neonate mustelids. The temperature should be gradually reduced to room temperature, 21.2-23.9°C (70-75°F), over the course of about three weeks (unless the neonate becomes ill). Litters of pups are less likely to need additional ambient heat since huddling together may provide an adequate amount of warmth. External temperatures should be closely monitored to prevent hyperthermia. Rapid and/or open-mouth breathing, restlessness, and hair loss are indication of an external environment that is too warm.

Pups should be stimulated to urinate and defecate, at least 4-5 times each day for several weeks, generally before feeding. However, some animals may respond better to post-feeding stimulation. The genitals and anal area are rubbed gently with a finger, towel, or damp cotton to stimulate the pup 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.

NEONATAL EXAMINATION & MONITORING GUIDELINES

(from Read & Meier 1996)

Categories should be recorded daily while pup(s) are in incubators and monitored regularly after that. The optional laboratory values should be recorded if analyzed.

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

FEEDING AMOUNT AND FREQUENCY

Initially, the animal should receive only an electrolyte solution for the first 2-3 feedings, depending on how compromised it is. This is to rehydrate the animal and clear the stomach of the maternal milk. The artificial formula should be started at a diluted concentration, generally at a 1:4 ratio (mixed formula: water) for another 2-3 feedings. It generally takes about 72 hours to get the animal on full-strength formula by gradually offering higher concentrations. Typically, 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) $^{0.78}$ (1972) is used rather than Kleiber's equation of 70 x body weight (in kg) $^{0.75}$ (1947) typically used for other species. Therefore, for a 200g river otter, the BER would be: $84.6 \times 0.2^{0.78} = \sim 24$ kcal/day.

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

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

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

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

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

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

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

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

Otter pups should only be fed if the pup is hungry and suckling vigorously. Weak infants may be hypothermic, dehydrated and/or hypoglycemic. Do not offer anything by mouth until the body temperature is within the normal range for its age (i.e., warm, not hot, to the touch; adult normal body temperature range is given as 38.1 – 38.7°C (100.6 to 101.7°F); baseline = 38.4°C (101.1°F) (Spelman 1999). Electrolytes can be offered orally if the pup is suckling, or subcutaneously if it is too weak; 2.5-5% dextrose can also be given to raise the pup’s glucose level. More research is required to determine body temperature norms for young of all the otter species. 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. (Photo: Florida Aquarium nursing orphan, P. Blum)

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

Feeding Techniques

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

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

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

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

Otter Milk, Milk Replacer and Selecting a Formula

"It should be noted that artificial milk replacer cannot be expected to perform as well as mother's milk. Rather, the goal is to produce a positive growth rate to enable the neonate to wean itself to a solid diet.

"In order to provide a neonate with proper nutrition and, hence, facilitate its growth while it is being fed an artificial milk replacer, one should remember the following concepts: (1) Protein, fat, carbohydrates, energy, vitamins, and minerals must be supplied in amounts and proportions that support a positive growth rate. (2) The amount fed at each feeding should not exceed the maximum comfortable stomach capacity. (3) The number of feedings per day should be adequate to supply daily energy requirements considering the finite amount that can be delivered at each feeding. (4) Every effort should be made to promote weaning to a complete and balanced solid diet when physiologically possible." (Evans 1985)

There have been a number of formulas used over the years to hand-raise otters. Keep in mind that the dams milk composition is presumed to change over the course of lactation and that, as of this writing, we do not know when these changes occur. For this reason, when hand-rearing otters it is important to closely monitor the pups' progress and re-evaluate the number of feedings and formula offered on a case by case basis. It is important that the artificial milk formula matches the maternal milk in protein, fat, and carbohydrate composition as closely as possible.

OTTER MILK COMPOSITION

Water: 62% Fat: 24.0% Protein: 11% Carbohydrates: 0.1% Ash: 0.75%
(Ben Shaul 1962)

Solids: 38% Fat: 23.9% Protein: 11% Carbohydrates: 0.1% Ash: 0.75%
Energy(KCAL/ML): 2.83 (Evans 1985)

Solids: 38% Fat: 63% Protein: 28.9% Lactose: 0.3%
(Pet Ag, Inc. 1993 Zoological Nutritional Components Milk Matrix Formulation & Mixing Guide. From: Jenness R. & R. E. Sloan 1970)

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)

It should be noted that no data is available as to when during the process of lactation milk samples were collected. Further, it has been pointed out that sample size and how the data was collected is not specified resulting, possibly, in a high margin of error. However, Evans (1985) comments: *"Despite the sources of error, practical experience has shown that these data generally are adequate for formulation of artificial milk replacers."*

MILK REPLACER & FORMULA OPTIONS (HISTORICAL RECORDS)

General Formulas

Esbilac and Milk Matrix 30/55 (also known as Multi-Milk available from *Zoologic® a division of PetAg®, 261 Keyes, Hampshire, IL 60140, 800-323-0877*), combinations of these, and combinations of these formulas with other additives have all been used. (Foster-Turley 1985, Lowry Park Zoo pers. com., Cain-Stage 1992, and many others). In Great Britain, a product called Lactol has been used successfully. (Johnstone 1978)

Davis (1983, unpublished): This information comes from a conversation between Joe Davis, IUCN/SSC Otter Specialist Group Advisor, and Joan Ryskamp, John Ball Zoo (6/22/83).

> *"He recommended Esbilac at a mixture of 1 part Esbilac powder to 3 parts water. Of the reconstituted Esbilac he suggested 2 tbsp. of liquid per 4 oz. of body weight, or, 1 fluid once of liquid per 4 oz* (113 g) *of body weight. For every pound of weight gain increase daily amount offered by 4 oz. He apparently did not recommend offering more than 6 oz*. (170 g) *per feeding.*

"Mr. Davis suggested using a rubber ear syringe for feeding because it is stronger than a regular nipple, it won't collapse, and it can be forced into the mouth if necessary.

"As solids are offered, he suggested adding rendered chicken fat and powdered Esbilac to Nebraska Feline diet. The additives are required because the feline diet does not have enough calories. For 1 lb (453 g) *of feline add, 2 tbsp. of rendered chicken fat and 3 to 4 tbsp. of powdered Esbilac. If meat mix will not be consumed quickly he strongly suggested adding 2 tbsp. of active cultured yogurt to prevent rancidity.*

"Pups should be pot-bellied, if they appear lean, increase milk.

"Wean at four months. If a weight loss is seen for two days there could be a severe problem."

Cain-Stage (1992): This formula was first published in the Wildlife Health News, 1992. M. Cain-Stage works with the rehabilitation center H.A.W.K.E., Inc. in St. Augustine, Fl.

"Multi-milk (one part powder:2 parts water)
½ oz. (15 ml) *liquid whole milk whipping cream*
½ tsp. white Karo syrup
One egg yolk
½ eye dropper of liquid HiVite multi-vitamins

"Formula is made fresh daily and heated portions should not be reheated. The formula is mixed in a blender and stored in the refrigerator until used.

"A composition more like the natural milk can be achieved using Multi-milk, Esbilac, and heavy whipping cream. The ration is as follows:
one part powdered Esbilac
two parts water
one part Multi-milk
one part heavy whipping cream

"…river otters have also been successfully raised using Esbilac mixed as shown on the carton."

Diana Sevin from the Bayou Otter Farm provided this formula that she uses for raising young otters:

½ c. Fowl starter/grower crumbles
½ c. Rice cereal
Mix with 1 c. Esbilac & 1 tsp. cod liver oil

MILK REPLACER & FORMULA OPTIONS (CURRENT)

The following two tables provide information on the nutritional content of otter milk, and on the nutritional composition of selected substitute milk formulas/replacers.

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

Product	Solids %	Fat %	Protein %	Carbohydrates %	Ash %	Energy (KCAL/ML)
3:1*	22.93	11.63	8.00	0.89	1.52	1.56
4:1*	22.90	11.60	7.86	0.71	1.49	1.55
1:3*	22.70	9.81	8.75	2.67	2.13	1.51
1:4*	22.60	9.65	7.54	2.84	1.39	1.43
Multi-Milk:Esbilac++						
1:1*	34.22	15.95	11.55	2.67	2.16	2.24
3:1*	34.40	17.45	12.00	1.34	2.28	2.33
4:1*	34.35	17.40	11.79	1.07	2.24	2.33
1:3*	34.05	14.72	13.13	4.01	3.20	2.28
1:4*	33.90	14.48	11.31	4.26	2.09	2.15

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

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

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

Preferred formulas

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

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®, is a group of beneficial gut bacteria that also break down milk sugars in the digestive tract. Follow label instructions for these products.

Haire (2011 and Section 3 Rehabilitation) states:

> ***Formula Note**
> *Recent change (2009) in the manufacturing process of Esbilac powder has been causing some growth and digestibility problems in squirrels, opossums and raccoons for some wildlife rehabilitators using this milk replacer. Problems regarding this product with other wildlife species have not yet been reported or published to author's knowledge.*
>
> *Pet Ag®, manufacturer of Esbilac and the Zoologic Milk Matrix line of milk replacers, reminds wildlife rehabilitators that using Esbilac on wildlife is "off label" usage and they recommend that instead rehabilitators use the Zoologic Milk Matrix products such as Zoologic 33/40 since it is manufactured and labeled for use in wild orphan mammals.*
>
> *Wildlife rehabilitators are advised to know about these issues in order to make informed decisions on the formulas we choose to feed. Current updates on milk replacers, feeding practices, and information on gastrointestinal conditions in wildlife are available at www.ewildagain.org.*

The following formulas used successfully by rehabilitators working with orphaned otter pups are provided in Haire (2011 and Section 3 Rehabilitation):

- *1 part powdered Esbilac® + 2 parts water + Lactobacillus (Avian Benebac™) powder (1t/cup of formula) (provided by M. Haire)*
- *1 part powdered Esbilac® + 2 parts water + 1 part heavy whipping cream + 1 part Multi-Milk® (provided by M. Caine-Stage)*

- *2 part liquid Esbilac® + 1 part whipping cream*
- *Multi-Milk ®30/55 until eyes open, than;*
  2 parts liquid Esbilac + 1 part Multi-Milk® (Provided by S. Beckwith)
- *Canned Esbilac® (as is)*
- *1 part powdered Esbilac® or Milk Matrix® 33/40 + 1 part powdered Multi-Milk® or Milk Matrix® 30/55 + 2 parts water*
- *Multi-Milk® 30/55 until eyes open then transition to Esbilac® (Zoologic milk substitute 30/55 has low level of lactose)*
- *Esbilac® 2 T/4 oz BW divided into 5 - 7 feedings every 2 - 3 hours until 10:00pm*
  4 weeks old consume 1 oz/feeding 4 - 6 x/day
  6 weeks old consume 2.5 oz/feeding 4 x/day (provided by Blasidell)

Feeding Young Pups and Weaning

FEEDING YOUNG PUPS (NEONATES TO SIX OR TEN WEEKS)

Very young pups are more difficult to raise, this information can be used as a guideline if pups are pulled when slightly older. It is preferable that neonates, in particular, are raised by experienced personnel. Care must be taken to closely monitor the animal's growth, development, and coat condition. **Do not** over feed, feed too quickly, bottle feed holding the pup on its back, forget to stimulate to urinate and defecate, allow severe diarrhea to continue, or change more than one formula/diet item at a time when trying to resolve a problem. **Do** keep the pup warm, clean, feed the age/weight appropriate amounts (frequent, small feedings for very young animals), wean to solids as soon as possible, keep detailed records, groom and maintain the pups coat, and teach the pup to swim.

Lowry Park Zoo
In 1994/95 Lowry Park hand raised a number of pups. Their guideline was to offer no more than 30% of the pup's body weight (in formula) in a 24 hour period. Weight data can be found later in this section.

Week 1:	Pups fed 8 – 10 times per day (every 2 –3 hours). 3 – 5ml. were offered at each feeding. Started with Pedialyte then offered Milk Matrix 30/55. The specific gravity (SG) of their urine was tested to monitor the pup's hydration. A SG value of 1.020 is considered normal for adults and 1.012 for juveniles; if it is higher the animal was considered to be dehydrated.
Week 2:	On day 10 switched to 1:1 Esbilac: Pedialyte; on day 11 full strength Esbilac was offered. Consumption at each feeding ranged from 5 – 9ml.
Week 3:	Pups were taking about 7 – 15ml. at each feeding.
Week 4:	Mid-week one feeding dropped so they are now fed seven times in 24 hours. 11 – 20ml. offered at each feeding.
Week 5:	20 – 30ml. offered per feeding
Week 6:	Up to 55ml. offered per feeding.
Week 7:	Same.
Week 8:	Chicken baby food was added on day 54. Pups still being fed seven times per 24 hours. ½ to 1 teaspoon of baby food offered each feeding.
Week 9:	Fed six times per day. 60ml. of Esbilac and 1 Tablespoon to 1/3 jar of chicken baby food was offered at each feeding.
Week 10:	Feedings were gradually reduced to four times per day. They continued to offer 60ml. of Esbilac at each feeding supplemented with chicken baby food which was then switched to a mix of liver baby food and feline diet.

FEEDING OLDER PUPS (SIX TO TEN WEEKS AND OLDER)

Occasionally zoos and aquariums may receive orphaned pups or have cause to take over feeding of weaning or weaned pups. In the case of wild orphans, they are frequently young animals found in the wild when the dam was killed by cars or dogs. Usually these animals are old enough to be following mom so should require minimal formula feeding. With animals in this age range (six to ten weeks) it is important to wean them onto solid foods as soon as possible. Watch the animals coat condition, fecal output and consistency. See Do's and Don'ts under Feeding Young Pups.

> *"Some of litter removed at six weeks (when mature there is no difference in size, weight, or condition when compared with naturally reared companions). To each 8 oz.* (237 ml) *of Carnation or homogenized milk, add 1 drop of Tri-vi-sol, yolk of one egg, 1 teaspoon lime water. Feed every 4 hours at blood heat from 6 weeks of age, then on demand. Shavings of lean horse-meat offered from start."* (Jeremy Harris, Oxnead Hall, Norwich, Great Britain)

The specific zoo/aquarium information listed below was taken from the responses to the 1994 John Ball Zoo North American River Otter Breeding Survey unless otherwise noted.

Audubon Park Zoo

The pups came in at about 2 ½ to 3 months old. At first they were given thinned Esbilac using a standard baby bottle and nipple. They were quickly switched to Esbilac mixed with baby cereal. The next progression was mixing meat baby food with the Esbilac.

Baton Rouge Zoo

The pups came in at about 6 weeks old. They were fed 2oz (59 ml) Esbilac and three to four whole smelt twice a day for about one week. a standard baby bottle and nipple were used. No problem getting the pups to nurse and the pups ate the smelt well. After about the first week, the adult diet (finely ground) was offered and their milk intake was gradually reduced over the next week. No additional supplements were added.

Metro Toronto Zoo

One animal was hand reared from about six weeks of age. Initially, he was fed five times a day (7am, 11am, 3pm, 7pm, 11pm). The formula was an Esbilac/2% milk combination; 1 ½ Tbsp. Esbilac to 3 ½ Tbsp. 2% milk. He also was fed finely ground carnivore mix in meatballs and smelt filets. *"As he got older, the number of feeds per day was reduced. At two months of age he was fed twice a day."*

WEANING

The weaning process should be started when the pup shows interest in solid food, generally at about 6.5 to 8 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. Only add one new food component to their diet every couple of days until they are eating solids well. It is best to be creative, flexible, and not to rush the weaning process. In the case of problems, try different approaches, try them multiple times, and try foods in new ways like bottles, syringes, suction bulbs, bowls, etc. Do not cut back on bottle-feeding to make the pup "hungry". Offer new food at the beginning of the feeding and finish with the bottle (Blum 2004). Situations to watch for during the weaning process include (Blum 2004): weight loss, diarrhea and sucking behavior. If sucking on tails, feet, genitals, etc. is observed between feedings, an additional bottle-feeding should be offered for a few days. R. Green of the Vincent Wildlife Trust recommends putting orange oil on the genitals to discourage sucking; this worked well with *Lutra lutra* and is not harmful to the otter (G. Yoxon, personal communication). Additional information on weaning pups is available in Section 3, Rehabilitation.

Young Animal Health Concerns

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. The following recommendations and cautions come from P. Blum (unpublished Florida Aquarium data).

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.

Diarrhea/constipation: 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
> Penicillin G Procaine: 40,000-44,000 IU/kg q24 hr IM

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.

L. CANADENSIS : HAND-REARED VERSUS MOTHER-REARED PUP WEIGHTS - TABLE

The following table provides a comparison of hand-reared versus mother reared pup weights taken from several institutions to be used as a guideline.

L. canadensis : Hand-reared versus Mother-reared Pup Weights - Table

Age in Days	LO* 100645	LO* 100646	LO* 100647	LO** 100651	LO** 100652	LR 3762	LR 3763	LR 3764	LR 4177	LR 4178	JBZ 301165	JBZ 301169	JBZ 301561	JBZ 301560	JBZ*** females
1	110.0g	118.0g	110.0g		170.0g	170.0g	170.0g	168.0g	168.0g	168.0g					
2	106.0g	120.0g	109.0g	177.0g	184.0g	177.0g	190.0g	179.0g	182.0g						
3	103.7g	119.7g	114.3g	220.0g	220.0g	198.0g	204.0g	193.0g	193.0g						
4	107.6g	123.7g	115.3g	241.0g	241.0g	213.0g	221.0g	204.0g	213.0g						
5	111.4g	127.0g	116.5g	276.0g	255.0g	248.0g	263.0g	241.0g	249.0g						
6	113.5g	135.5g	123.5g	298.0g	291.0g	262.0g	291.0g	249.0g	272.0g						
7	113.7g	138.0g	115.0g	333.0g	326.0g	298.0g	322.0g	266.0g	286.0g						
8	113.2g	137.4g	124.4g	354.0g	354.0g	333.0g	344.0g	288.0g	308.0g						
9	110.5g	135.8g	122.5g	376.0g	376.0g	347.0g	372.0g	325.0g	347.0g						
10	111.6g	142.4g	135.6g	404.0g	404.0g	383.0g	397.0g	353.0g	364.0g						
11	115.3g	145.7g	137.0g	425.0g	418.0g	397.0g	412.0g	364.0g	378.0g						
12	123.0g	165.5g	152.5g	453.0g	446.0g	411.0g	445.0g	398.0g	414.0g						
13	122.0g	173.2g	159.4g	475.0g	467.0g	439.0g	473.0g	414.0g	431.0g						
14	136.0g	195.0g	176.2g	496.0g	496.0g	454.0g	459.0g	428.0g	448.0g						
15	147.0g	223.0g	200.5g	539.0g	531.0g	489.0g	498.0g	437.0g	462.0g						
16	152.5g	240.0g	220.0g	574.0g	560.0g	517.0g	554.0g	454.0g	468.0g	499.0g			533.0g	672.0g	
17	171.5g	256.0g	240.5g	595.0g	595.0g	546.0g	560.0g	473.0g	512.0g	532.0g					
18	183.6g	296.5g	271.5g	624.0g	617.0g	560.0g	599.0g	496.0g	526.0g						
19	199.5g	321.0g	290.0g	645.0g	624.0g	609.0g	633.0g	515.0g	535.0g						
20	218.5g	346.0g	305.5g	680.0g	666.0g	637.0g	636.0g	549.0g	568.0g						
21	233.0g	367.0g	330.0g	440.0g	450.0g	687.0g	687.0g	652.0g	672.0g	566.0g					
22	252.0g	397.0g	352.0g	483.8g	478.5g	780.0g	765.0g	723.0g	694.0g	619.0g			721.0g	912.0g	
23	280.0g	429.0g	388.0g	484.0g	492.0g	808.0g	780.0g	723.0g	734.0g	622.0g					
24	309.0g	467.0g	388.0g	520.0g	515.5g	843.0g	822.0g	758.0g	756.0g	644.0g					
25	343.0g	500.0g	453.0g	575.0g	543.0g	858.0g	822.0g	772.0g	806.0g	669.0g					
26	346.0g	490.6g	473.4g	570.5g	545.5g	872.0g	829.0g	772.0g	829.0g	711.0g					
27	367.0g	530.0g	483.5g	594.0g	565.1g	872.0g	850.0g	794.0g	871.0g	736.0g					
28	372.0g	535.0g	484.5g	621.6g	592.1g	886.0g	865.0g	815.0g	890.0g	756.0g	862.0g	907.0g			
29	412.0g	595.0g	560.0g	658.0g	610.0g	921.0g	907.0g	872.0g	913.0g	840.0g			952.0g	1.18kg	
30	448.0g	643.0g	553.0g	665.0g	637.0g	978.0g	935.0g	907.0g	969.0g	879.0g					
31	473.2g	660.6g	607.0g	725.0g	680.0g	999.0g	971.0g	928.0g	1.00kg	935.0g					
32	491.0g	652.8g	622.3g	733.6g	690.5g	1.04kg	992.0g	971.0g	1.04kg	969.0g					
33	510.0g	676.0g	632.1g	753.0g	725.0g	1.09kg	1.00kg	921.0g	998.0g	1.09kg	953.0g	1.09kg			
34	543.0g	735.0g	658.5g	801.2g	796.0g	1.11kg	1.08kg	1.01kg	1.13kg	1.04kg					

* Hand raised ** Wild born then hand raised, estimate as to age. *** Weights for four female pups born at JBZ, lightest weight recorded for that particular day, may be from different pups. All four pups survived to adulthood. There is no indication why the LO** pups show such a drastic weight change on day 21

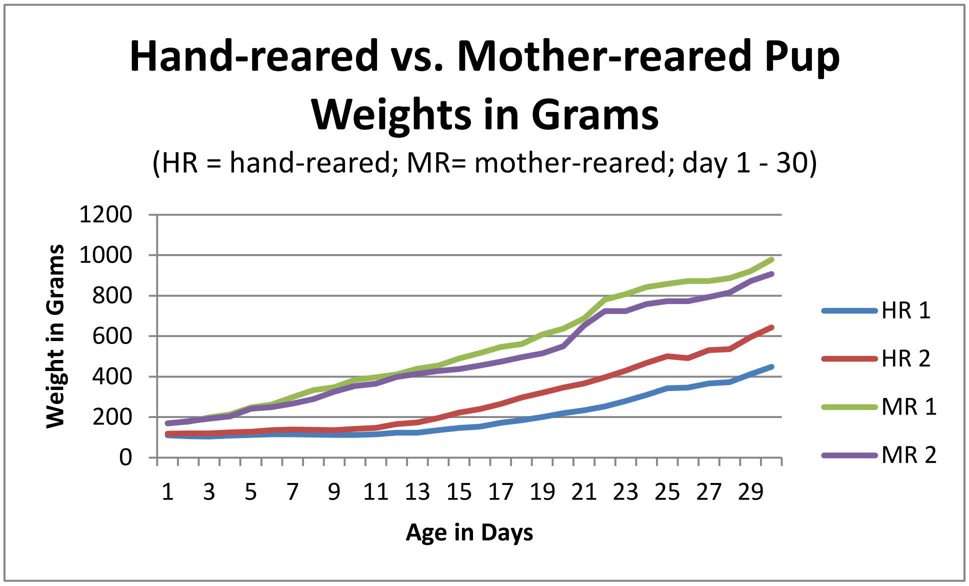
L. canadensis : Hand-reared versus Mother-reared Pup Weights – Table (cont.)

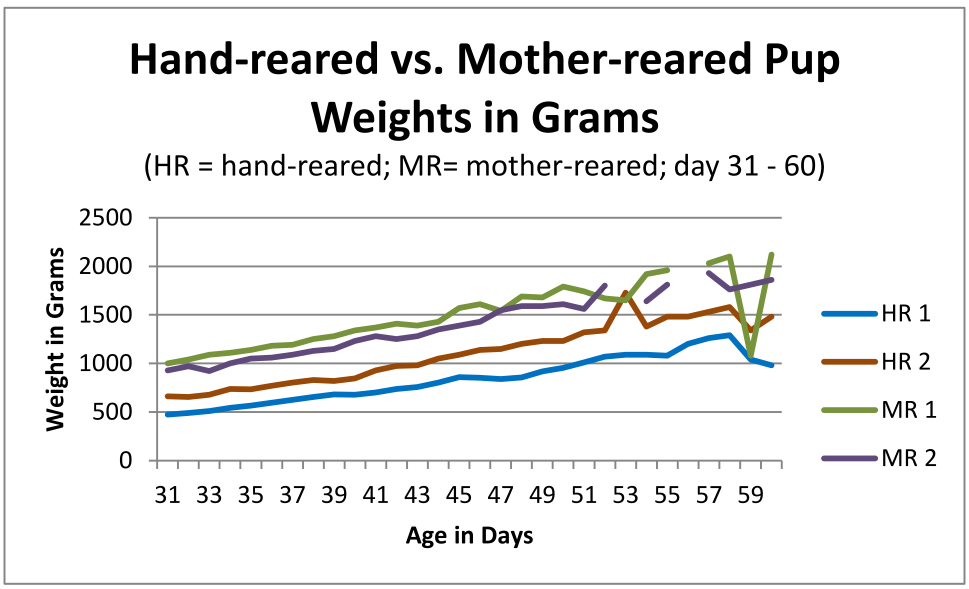
Age in Days	LO* 100645	LO* 100646	LO* 100647	LO** 100651	LO** 100652	LR 3762	LR 3763	LR 3764	LR 4177	LR 4178	JBZ 301165	JBZ 301169	JBZ 301561	JBZ 301560	JBZ*** females
35	564.0g	733.0g	688.0g	808.0g	783.0g	1.14kg	1.11kg	1.05kg	1.15kg	1.06kg	998.0g	998.0g			
36	595.0g	771.0g	725.5g	878.0g	838.0g	1.18kg	1.13kg	1.06kg	1.20kg	1.09kg			1.23kg	1.53kg	
37	625.0g	802.0g	760.0g	913.5g	903.5g	1.19kg	1.16kg	1.09kg	1.22kg	1.14kg					
38	654.2g	828.7g	814.0g	957.0g	898.0g	1.25kg	1.21kg	1.13kg	1.28kg	1.18kg					
39	678.6g	820.0g	827.7g	998.0g	916.0g	1.28kg	1.23kg	1.15kg	1.33kg	1.20kg					
40	677.2g	844.8g	813.5g	1.01kg	951.0g	1.34kg	1.28kg	1.23kg	1.37kg	1.26kg					
41	700.0g	926.8g	865.9g	1.08kg	977.7g	1.37kg	1.35kg	1.28kg	1.39kg	1.25kg	1.20kg	1.30kg			
42	736.9g	972.6g	918.7g	1.11kg	1.02kg	1.41kg	1.32kg	1.25kg	1.43kg	1.29kg					
43	754.8g	980.0g	900.8g	1.12kg	1.07kg	1.39kg	1.35kg	1.28kg	1.46kg	1.31kg					
44	802.0g	1.05kg	981.0g	1.18kg	1.09kg	1.43kg	1.40kg	1.35kg	1.49kg	1.33kg					
45	856.5g	1.09kg	997.0g	1.21kg	1.12kg	1.57kg	1.45kg	1.39kg	1.55kg	1.39kg	1.32kg	1.45kg			
46	852.4g	1.14kg	1.04kg	1.20kg	1.25kg	1.61kg	1.51kg	1.43kg	1.42kg	1.42kg					
47	837.6g	1.15kg	1.08kg	1.29kg	1.61kg	1.54kg	1.47kg	1.62kg	1.45kg	1.39kg					
48	853.7g	1.20kg	1.15kg	1.38kg	1.30kg	1.69kg	1.59kg	1.51kg	1.62kg	1.46kg					
49	917.5g	1.23kg	1.19kg	1.40kg	1.30kg	1.68kg	1.59kg	1.59kg	1.65kg	1.50kg					
50	952.5g	1.23kg	1.19kg	1.41kg	1.32kg	1.79kg	1.69kg	1.61kg	1.69kg	1.53kg					
51	1.01kg	1.32kg	1.26kg	1.47kg	1.39kg	1.74kg	1.62kg	1.56kg	1.77kg	1.59kg			1.76kg	2.07kg	
52	1.07kg	1.34kg	1.27kg	1.12kg	1.87kg	1.67kg	1.53kg	1.80kg	1.62kg	1.60kg					
53	1.09kg	1.73kg	1.61kg	1.84kg	1.63kg	1.65kg									
54	1.09kg	1.38kg	1.28kg	1.50kg	1.40kg	1.92kg	1.74kg	1.64kg	1.85kg	1.68kg					
55	1.08kg	1.48kg	1.38kg	1.46kg	1.46kg	1.96kg	1.71kg	1.81kg	1.90kg	1.72kg	1.63kg	1.72kg			
56	1.20kg	1.48kg	1.39kg	1.80kg	1.51kg				1.90kg	1.74kg	1.54kg	1.68kg			
57	126kg	1.53kg	1.44kg	1.48kg	1.48kg	2.03kg	1.72kg	1.93kg	1.93kg	1.78kg	1.59kg	1.72kg	2.04kg	2.27kg	
58	1.29kg	1.58kg	1.48kg			2.10kg	1.87kg	1.76kg							
59	1.04kg	1.34kg	1.34kg	2.09kg	1.90kg	1.80kg	1.63kg	1.81kg			1.633kg	1.81kg			
60	980.0g	1.48kg	1.46kg	1.80kg	1.70kg	2.12kg	1.93kg	1.86kg							
61	1.10kg	1.54kg	1.48kg	1.90kg	1.80kg	2.15kg	2.00kg	1.84kg							
62	1.34kg	1.51kg	1.40kg	1.90kg	1.85kg	2.24kg	1.95kg	1.88kg							
63	1.50kg	1.90kg	1.12kg	1.90kg	1.90kg	2.25kg	1.20kg	1.91kg	1.59kg	1.81kg	1.59kg	1.81kg			
64	1.60kg	1.80kg	2.0kg	1.90kg	2.26kg	2.00kg	1.93kg						2.04kg	2.27kg	
65	1.60kg	1.90kg	1.80kg	2.0kg	1.90kg	2.23kg	2.01kg	1.96kg	1.66kg	1.81kg	1.66kg	1.81kg			

* Hand raised   ** Wild born then hand raised, estimate as to age.   *** Weights for four female pups born at JBZ, lightest weight recorded for that particular day, may be from different pups. All four pups survived to adulthood. There is no indication why the LO** pups show such a drastic weight change on day 21

Hand-reared versus Mother-reared Pup Weights in Grams – Graphs

There is no explanation for the dramatic weight shifts in Mother-reared (MR 2) pup, possibly due to scale error. The data is taken from the previous chart. Weights of hand-reared pups should be catching up to those of mother-reared by 60+ days or once they are weaned.





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CHAPTER 9 Feeding and Nutrition

"Wild animals require the same basic nutrients as their domestic counterparts. For many exotic species that have closely related domestic counterparts (e.g. …mustelids…), nutrient requirements established by the National Research Council (NRC) for domestic and laboratory animals can be a guide to minimum nutrient concentrations in the diet. Although less directly applicable to other species, NRC requirements can still serve as a useful general reference for evaluating the nutritional adequacy of diets for any bird or mammal." (Merck 1991)

General Guidelines

The target nutritional values for otters are based on several sources. The cat is typically used to establish nutrient guidelines for carnivorous animals. The National Research Council (NRC) (1986, 2006), Association of American Feed Control Officials (AAFCO) (1994), and Waltham Center for Pet Nutrition (Earle & Smith 1993) have provided recommendations for cats. A limited amount of information is provided by the NRC publication for mink and foxes (1982), which represents the requirements of another mustelid species. The target nutrient values presented (Maslanka & Crissey 1998) are a range of values reported from various references. As new information becomes available, these ranges will change to reflect knowledge gained. Table 1 lists dietary nutrient ranges for otters. Table 2 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.

It is essential that good quality foods be offered. Long storage times of frozen foods (over 6 – 12 months depending on the food item) should be avoided. Bagged feeds should be stored less than one year (Merck 1991). Specific guidelines for the inspection and handling of fish and/or meat products can be found in the UDSA publications: Crissey and Spencer, 1998.

Primarily piscivorous, otters have high metabolic rates, rapid digestion, and have been found to spend 41-60% of their time involved in feeding or foraging activities (Hoover & Tyler 1986; Davis et. al 1992; Kruuk 1995; J. Reed-Smith, unpublished data). Duplaix-Hall (1975) found that otters (unidentified species) in the wild rarely ate more than about 500g of food at a time and that they consumed approximately 20% of their own body weight daily. Kruuk (1995) reviewed his and other study results indicating that *ex-situ* populations of *Lutra lutra* consuming between 11.9-15% of their body weight maintained a healthy weight. A study conducted by Ben-David et al. (2000, 2001a and b) reported success using 10% of a *L. canadensis'* (*ex-situ* population) body weight as a guide for the basis of their maintenance diet. In general, otters are very active animals and as such require nutrient dense diets. Fats are an important source of energy for these animals (Wallach & Boever 1983). Given the rapid transit time of food through their intestinal tract (see below), and the generally high activity level of otters, frequent, smaller feedings will help encourage activity, and eliminate concerns over food spoiling before it is consumed.

Clean, fresh drinking water should be available at all times. If an otter refuses two or more meals consecutively, they should be monitored for potential health problems.

WILD FEEDING HABITS

As previously stated, free-ranging North American river otters are reported to spend 41-62% of their time engaged in foraging and feeding activities (Hoover and Tyler 1986) in a primarily aquatic habitat. As a general rule, otters will first prey on those species easiest to catch and stay in shallow waters or near the shore. (Sheldon & Toll 1964, Knudsen & Hale 1968, Toweill & Tabor 1982)

N. A. otters are predominantly piscivorous. Researchers have found that the bulk of their diet consists of fish and crustaceans (crayfish); a minor portion of their diet consists of: amphibians (frogs, salamanders, newts and mud-puppies), reptiles, birds (most frequently ducks and divers), aquatic insects, mollusks, and occasionally small mammals (Chanin 1985, Berg 1999). The percentage of crayfish varies seasonally and with geographic location (Grenfell 1974, Chanin 1985, Toweill & Tabor 1982,) becoming the primary prey choice during most of the year. Also see: Feeding Behavior under Natural History.

H. Hansen provided an excellent overview of wild N. A. river otter diet studies in her 2003 dissertation (University of Wyoming); an excerpt is below:

River otters consume a wide variety of fish species ranging in size from 0.8 to 19.5in (2 to 50cm) that provide adequate calorie intake from a small amount of energy expenditure (Melquist and Dronkert 1987). Ryder (1955) stated that river otters feed predominantly on prey in proportion to their abundance but in inverse proportion to their swimming ability. Therefore, slow swimming fishes are preyed upon more often than game fishes when both are equally abundant (Serfass et al. 1990; Toweill and Tabor 1982). Slow-moving fishes include suckers (Catostomidae); sunfishes and bass (Centrarchids); and daces, carp and shiners (Cyprnidae) (Route and Peterson 1988). For example, Berg (1999) found Catostomidae to dominate the diet in the Upper Colorado River Basin in Colorado. Likewise, in other regions of Colorado, Beck (pers. comm.) found common carp (Cyprinus carpio) to be a preferred fish species for the otter.

Some specific examples of fish species that have been found frequently in the otters' diet include: Catostomidae - suckers (Catostomus spp) and redhorses (Moxostoma spp); Cyprinidae - carp (Cyprinus spp), chubs (Semotilus spp), daces (Rhinichthys spp), shiners (Notropis spp and Richardsonius spp) and squawfishes (Ptychocheilus spp); Ictaluridae - bullheads and catfishes (Ictalurus spp). Other fishes that are important in the otters' diet include: fishes that are often abundant and found in large schools such as sunfishes (Lepomis spp), darters (Etheostoma spp) and perches (Perca spp); and bottom dwelling species that are susceptible because of their habit of remaining immobile until a potential predator is close such as mudminnows (Umbra limi) and sculpins (Cottus spp.) (Melquist and Hornocker 1983; Toweill 1974; Toweill and Tabor 1982).

Game fishes, such as trout (Salmonidae) and pike (Esocidae), are not an important part of the river otters' diet (Melquist and Dronkert 1987; Toweill and Tabor 1982). Game fishes are fast-swimming and can find good escape cover, making them less available as prey for the otters (Melquist and Dronkert 1987). However, river otters will
eat trout (Salmo spp), pike (Esox spp), walleye (Stizostedion vitreum), salmon (Oncorhynchus spp), and other game fishes during spawning (Melquist and Hornocker 1983; Reid et al. 1994; Toweill 1974).

Adult river otters can consume 1 – 1.5 kg (2 – 3 lb) of fish per day (Serfass et al. 1990)

Crustaceans
Across North America where crustaceans, especially crayfish (Cambarus spp, Pacifasticus spp, and others), are locally and seasonally abundant, otters may prefer to feed on them more than fish (Route and Peterson 1988). In Georgia, crayfish constituted 2/3 of the prey items in the summer diet and were present in 98% of the summer spraint. In the winter, crayfish constituted 1/3 of the otters' diet (Noordhuis 2002). Tumlison and Karnes (1987) documented a shift in the river otters' diet from fish to crayfish with a shift in water levels in a swamp in Arkansas. During the winter and spring when the water levels were higher, otters preferred to feed on crayfish (73% of scats had crayfish remains) more than fish (Tumlison and Karnes 1987). However, during low water events, crayfish will seek out shelter while fish become more concentrated and highly vulnerable. Therefore, fish are more susceptible to being preyed upon by otters because the easier-to-catch crayfish are more difficult to obtain (Route and Peterson 1988).

Conclusion
River otters' food habits are determined by prey availability (Ryder 1955). This availability may be determined by the following factors: (1) detectability and mobility of the prey; (2) habitat availability for various prey species; (3) environmental factors such as water depth and temperature; and (4) seasonal changes in prey abundance and distribution in relation to otter foraging habitat (Melquist and Dronkert

1987; Route and Peterson 1988). Otters do not seriously reduce prey populations. When an abundant food source diminishes or other prey become available, otters either move to a new location or shift their diet to the most available prey (Melquist and Hornocker 1983). Although other prey species are important to the river otter temporally, the potential limiting factor to the river otter being established as a permanent resident is the availability of fish year-round (Melquist and Hornocker 1983).

GI-TRACT MORPHOLOGY

Otters are semi-aquatic carnivores. As in other mustelids, they possess a simple stomach. They may have a somewhat elongated small intestine like the mink. They do not have a cecum. (Steven and Hume, 1996)

Digestion

Davis et. al. (1992) and Spelman et al. (1997) report that gastrointestinal transit time for a fish meal took anywhere from 2 to 4 hours with a mean of 202 minutes. In 1951, Liers tested otters previously fed a bland diet and found that the exoskeletal remains of crayfish were passed about one hour after consumption.

Target Nutrient Ranges and Nutrient Content of Dietary Component Samples

TARGET DIETARY NUTRIENT VALUES FOR A FISH-EATING SPECIES.

Much of this information is similar to that reported for Asian small-clawed otters (Maslanka and Crissey, 1998). Target values for otters are based on several sources. The cat is typically used to establish nutrient guidelines for carnivorous animals. The NRC (1986), AAFCO (1994), and Waltham Center for Pet Nutrition (Earle and 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 here (Table 1 & Table 2; Maslanka and Crissey, 1998; Henry and Maslanka, 2010) are a range of values reported from various references. As new information becomes available, these ranges will change to reflect knowledge gained.

Energy requirements for otters have not been determined specifically, however dietary energy target values can be based on diets successfully used to maintain captive otters (Table 1). According to survey information (Reed-Smith 1997, Foti 2010), most institutions offer food two or more times per day. Where seasonal dietary alterations occur (in approximately one half of the responding institutions), most involve diet increases during the winter months.

(Photo: P. B.um, Florida Aquarium, First fish.)

Table 1: Target dietary nutrient ranges for North American River Otters.

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 (α)
Thiamin, mg/kg	1-5 (α)
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 B12, mg/kg	0.02-0.025
Choline, mg/kg	1000-3000
Calcium, %	0.6-0.8 (β)
Phosphorus, %	0.6 (β)
Potassium, %	0.2-0.4
Sodium, %	0.04-0.06
Magnesium, %	0.04-0.07
Zinc, mg/kg	50-94
Copper, mg/kg	5.0-6.25
Manganese, mg/kg	5-9
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 (NRC 1986), AAFCO recommendations (1994), Waltham Center for Pet Nutrition recommendations (Earle and Smith 1993), and requirements for mink and foxes (NRC 1982).
** The fat content of fish commercially available in North America typically ranges from 5-40% (Maslanka and Crissey 1998), and North American river otters have been maintained on diets containing 24-30% fat (Reed-Smith 1994), thus an appropriate range for fat appears to fall between 15-30%.
*** The vitamin A requirement for cats is 10 IU/g (dry matter basis; NRC 1985), which represents the upper bound of the range. However, free-ranging North American river otters may consume a higher proportion of fish and may have a higher tolerance for vitamin A due to the high levels which may occur in their natural diet.
(α) 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 100-120 mg thiamin / kg of dry diet are recommended (Engelhardt and Geraci 1978; Bernard and Allen, 1997). (β) The recommended Ca:P ratio is between 1:1 and 2:1.

Table 2: Target nutrient ranges for carnivorous species (dry matter basis) (Henry and Maslanka, 2010)

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

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

NUTRIENT CONTENT OF TYPICAL DIET INGREDIENTS.

As mentioned previously, several basic diet items (including fish and commercially available dry, canned, and frozen diets) have been used to maintain otters in captivity. Meat-based diets have been used in most institutions for growth, maintenance, and breeding. There is a variety of products upon which the diet may be based. Following are tables that describe nutrient concentrations in some commonly used manufactured products that are intended to be nutritionally complete (Maslanka and Crissey, 1998).

Nutrient content of several commonly used meat/nutritionally complete food items (dry matter basis).[a]

Nutrient	Ground horsemeat	Nebraska Feline ®
Crude protein, %	51.7	50.0
Fat, %	19.7	31.6
Vitamin A, IU/g	-	97.1
Vitamin D, IU/g	-	1.2
Vitamin E, mg/kg	-	42.4
Ca, %	-	1.6
P, %	-	1.3
Mg, %	-	0.05

[a] Values from manufacturers' guaranteed analysis and analyses performed at or for Brookfield Zoo.

Nutrient	Toronto Zoo Feline Diet*	Toronto Carnivore Diet	Dallas Crown Diet
Protein % (min.)	50	50	30, 56 expected(exp)
Fat % (min.)	20	25	10, 20% exp.,40 max.
Crude Fibre % (max.)	6	4	3
Calcium % (min.)	0.8	0.7	1.3
Phosphorus % (min.)	0.7	0.6	1.2
Magnesium %	0.09	0.07	0.09
Iron ppm.	160	190	80, exp. 183
Zinc ppm.	120	170	110
Taurine %	0.2		0.3
Vit. A IU/kg	11,000	18,000	14,000
Vit. E IU/kg	200	170	470
Vit. D IU/kg	2,160	3,540	2400

***** Manufactured by: Milliken Meat Products, Ltd. of Ontario, Canada. ^ Manufactured by Dallas Crown, Inc., Kaufman, Texas.

Nutrient content of several commercially available fish species and marine products (dry matter basis).[a] (Values vary with each shipment and provider.)

Nutrient	Capelin	Herring	Oyster shell	Shrimp[17,19]	Smelt	Trout
Dry matter, %	18.8	27.7	100	24.1	22.7	23.7
GE, kcal/g	5.5	6.3	-	2.3	7.0	6.5
Crude protein, %	59.8	45.3	0	20.5-44.2	70.4	55.8
Fat, %	14.8	34.0	0	1.8-4.3	16.6	34.5
Vitamin A, IU/g	44	56	0	-	-	58
Vitamin E, IU/g	0.024	0.034	0	-	-	0.32
Ca, %	1.7	1.7	38.0	10.8	1.4	2.1
P, %	1.7(1.2-1.4)[b]	1.3	0.07	2.1	1.6	1.5

[a]Analyses from Bernard and Ullrey[3], Minnesota Zoo, and Brookfield Zoo, unless otherwise noted.
[b]Unpublished data from the Brookfield Zoo and Minnesota Zoo.

Sample Diets

The one best diet for any of the otters of *ex-situ* populations has not been found and requires further research. However, current recommendations are that a variety of fish species should be offered 3-4 times a week, preferably daily (AZA 2010). 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 (Hazard Analysis and Critical Control Point) 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 diets of several institutions are listed below. This is just a sampling of diets being fed *L. canadensis*. Where available, comments on palatability to the otters, any associated dietary problems and observed physical condition of the animals are noted.

DIETS FROM THE LITERATURE

Liers' Diet (1951)
His mixture was: "*74% ground horse meat, ½% ground raw liver, 2% bone meal, 8% bran, ½% grated carrots, tomatoes, or lemon or orange juice, 10% raw rolled oats, and 5% commercial mink meal. To this I add one teaspoonful of cod liver oil per day per otter, one ounce of brewer's yeast per day per ten otters, one egg a day for each two otters. The entire concoction is mixed thoroughly with enough whole liquid milk to make a soft mash.*" When fresh food was available, such as frogs, crayfish or fish, they were fed as well.

Duplaix-Hall (1975)
Due to their high metabolic rate and rapid digestion, otters *"…eat up to 20 per cent of their own weight daily."* Because an adult will normally eat no more than 500 g (17.5 oz.) of food at a time, otters should be fed at least twice, preferably three times a day. She suggests feeding day-old chicks, and some fish to supply the necessary roughage. The following amounts are given by her as the food requirements per otter per day:

Chopped raw beef or horse meat	1500g.	Dog meal	200g.
Osteo-calcium	1 tablet	Halibut liver oil	4 drops
Soluble multi-vitamins	3 drops	Bone meal	50g.
Bran	50g.	Raw carrot	50g.
Vegetable oil or margarine	50g.	Day-old chicks	4
Fish (6 – 10") or eels	4		

ZOO AND AQUARIUM DIETS (1990'S AND 2000'S)

Example 1(Reed-Smith 1997)
Fed at least three times a day, sometimes four or five times per day. These additional feedings are fish, rib bones, hard-boiled eggs, mice, or live fish as enrichment items offered on a limited basis above and beyond their normal diet. The amount of feline diet is increased only when the female is lactating (for her, fish also is increased), or rarely, as an enrichment treat hidden around the exhibit or frozen in small meat balls. Anecdotally, it has been found

that the addition of extra fish (the diet was fish 2x/week) has improved their coats' water repellency but not caused them to add weight.

142 g (5 oz.) Nebraska Feline Diet – 2x/day, 7x/week.
½ medium carrot – 1x/day scattered on exhibit.
2 rib bones – 1 to 2x/week.
1 medium sized trout – 4x/week.

Example 2 (Reed-Smith 1997)

Daily: 3 – 3 ½ lbs. capelin or smelt, Vitamins B, E, and Pet Tabs added.
The respondent stated after switching to this diet, from a diet of meat and pellets, their animals no longer had any dental problems.

Example 3 (Reed-Smith 1997)
Their otters are fed a diet of fish (approximately 75%), and Nebraska Brand Feline Diet (approximately 25%). No supplements are added. The coats of both animals show good water repellency, the otter that generally refuses most of the feline has consistently had better dental hygiene.

Example 4 (Reed-Smith 1997)
Amounts vary with the animals' weight.
Toronto Diet: 200 to 300g (7 – 10.5 oz.) per feeding (400 - 600g (14 – 21 oz.) per day; mostly in the bottom half of that range (400 - 500g (14 – 17.5 oz.) per day.
Mackeral: One, every-other-day.
Derm-Caps

Example 5 (Reed-Smith 1997)
60% Toronto Feline Diet
40% fish (butterfish, capelin, herring or spot)
Daily they receive one or more of the following: mice, chicks, live fish, carrot, sweet potato, apple, hard-boiled egg.

Amounts vary with the condition and weight of the animal. Generally an adult diet is: *"... about 1 1/4 cup meat daily along with 4 capelin and 2 butterfish or herring. The otters are weighed weekly and amounts adjusted if needed. Usually one chick, one mouse, one egg each for the additional items. We give them 100iu Vitamin E and 25 mg Vit B-1 every third day."*

Example 6 (Reed-Smith 1997)
"I consider fish to be critical. I have seen many facilities offer otters only feline meat product with some vitamin supplements. We feed our otters whole smelt as their primary food source. They each also get two chicken breasts a day, and two whole mice. This provides a variety of items that mimics their natural diet. On top of that we add vitamins (Mazuri Marine Mammal vitamins). This combination has provided our river otters with excellent coats. During random times of the day, other food items are occasionally given to the otters to promote behavioral enrichment. This could be in the form of live minnows, fiddler crabs from our salt marsh, some fruits or veggies frozen in ice, etc...."

Example 7 (Reed-Smith 1997)
Amount fed differs for each animal, based on weight and age. Amounts given are the total for a 24 hour period, animals are fed four times a day. Two to three feedings per day are training sessions, at least one is a "free feed".

1.0 adult, weight: approximately 22 lb.	0.1 7 ½ months, weight: approximately 14 lbs.
7 oz. (198.5 g) Hill's Science Diet-Feline Light	4 oz. (113 g) Hill's Science Diet – Feline Light
½ lb. (227 g) Capelin	¾ lb. (340 g) Capelin
¾ lb. (340 g) Lake smelt	¾ lb. (340 g) Lake smelt
4 baby carrots & ½ medium carrot	4 baby carrots
1 *Nature Made* Antioxidant capsule*	
(this is given once daily)	1 *Derm Cap* every other day^

* *Nature Made* Antioxidant formula capsules: Information available @ 1-800-276-2878 or www.naturemade.com

^ *Derm Cap*: DVM Pharmaceuticals, Inc., Miami, Florida

645mg. per capsule:	crude protein	not less than 7%
	crude fat	not less than 90%
	crude fiber	not less than 1%
	moisture	not less than 2%
	Vit. E	75 IU
	Linoleic Acid	71%
	Gamma Linolenic Acid	2%
	Eicosapentaenoic Acid (EPA)	4%
	Docosahexaenoic Acid (DHE)	3%

Example 8 (Reed-Smith 1997)
Amounts vary with the weight of the animal which varied from 8.7 to 9.7kg for the diets listed below. Listed here are the total amounts which are divided each day into three to six feedings and/or training sessions. Enrichment items are not given here.

Smelt	.7kg.	.8kg.	.7kg.
Clam	.3kg.	.4kg.	.3kg.
Shrimp	.3kg.	.3kg.	.2kg.

2 Mazuri Bird vitamin tablets per day.

Example 9 (AZA 2010)
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

Example 10 (AZA 2010)

- 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

Example 11 (Montgomery 2012 personal communication)

- 825g smelt ± 25 – 50g (dependent on target weight for each otter, can go as high as 950g right before breeding season due to increase in activity; even though this is an all-male group)
- 75 – 200g peeled shrimp (dependent on individual; shrimp peeled due to a problem with possible choking on exoskeleton)
- 4 – 6 whole capelin
- 5g carnivore diet (just switched to horsemeat-based, will increase up to 10g only due to high fat and caloric content of the horse-based product. Fed up to 25g daily of beef-based carnivore diet)
- Live crayfish and fish several times per week, as enrichment

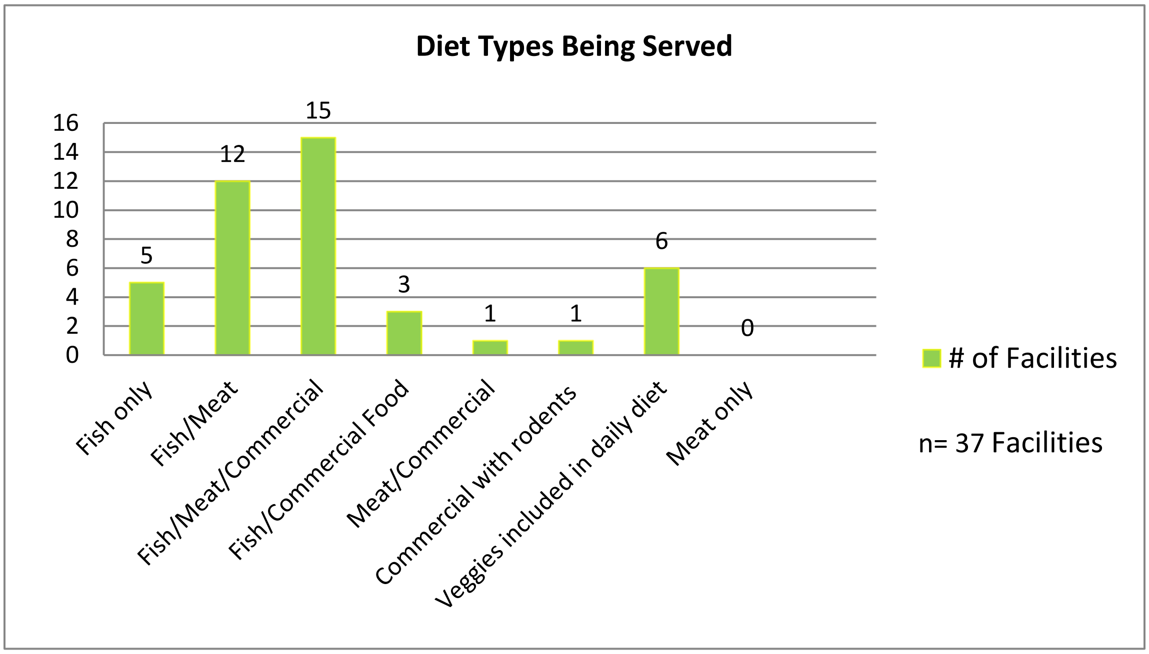
- Five days per week fed a "avian fish-eating vitamin tab" which will soon be switched to a marine mammal tab due to more appropriate Vitamin E levels.

Diet is offered spread out over 4 meals, roughly 2 to 2 ½ hours apart. If the otters refuse one meal this amount is not added to the total (they used to add it but once they allowed them to self-regulate by refusing a meal they found the animals' weights remained more consistent). (Editor Note: The otters fed this diet are 2 years, 14 years, and 14 years old. Their coats are in excellent condition, weights are good, and their teeth are in very good shape.)

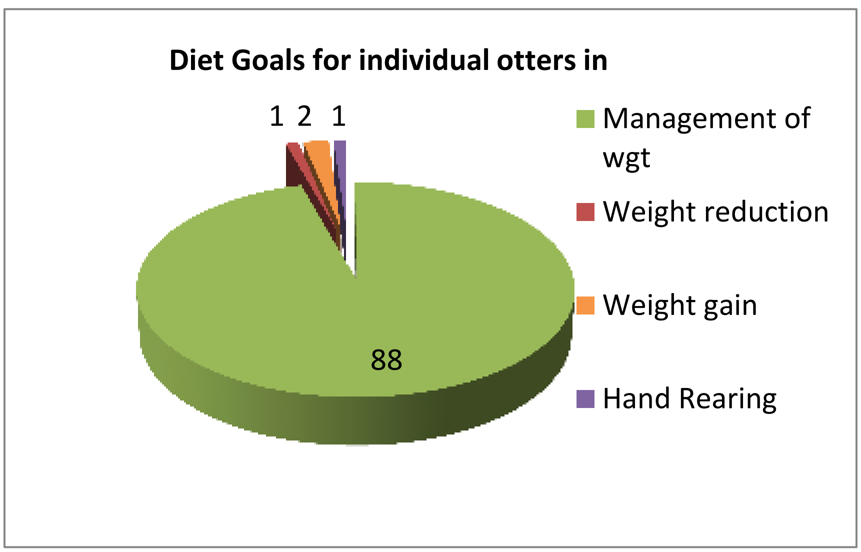
FOTI DIET SURVEY (2010)

Thirty-seven facilities participated in a diet survey, conducted by J. Foti and presented at 2010 AZA Otter Keeper Workshop, by contributing diet information for 93 N. A. river otters (age range 1 – 20 years; weight range 4 – 15 kg). A summary of the survey is presented here in chart form.

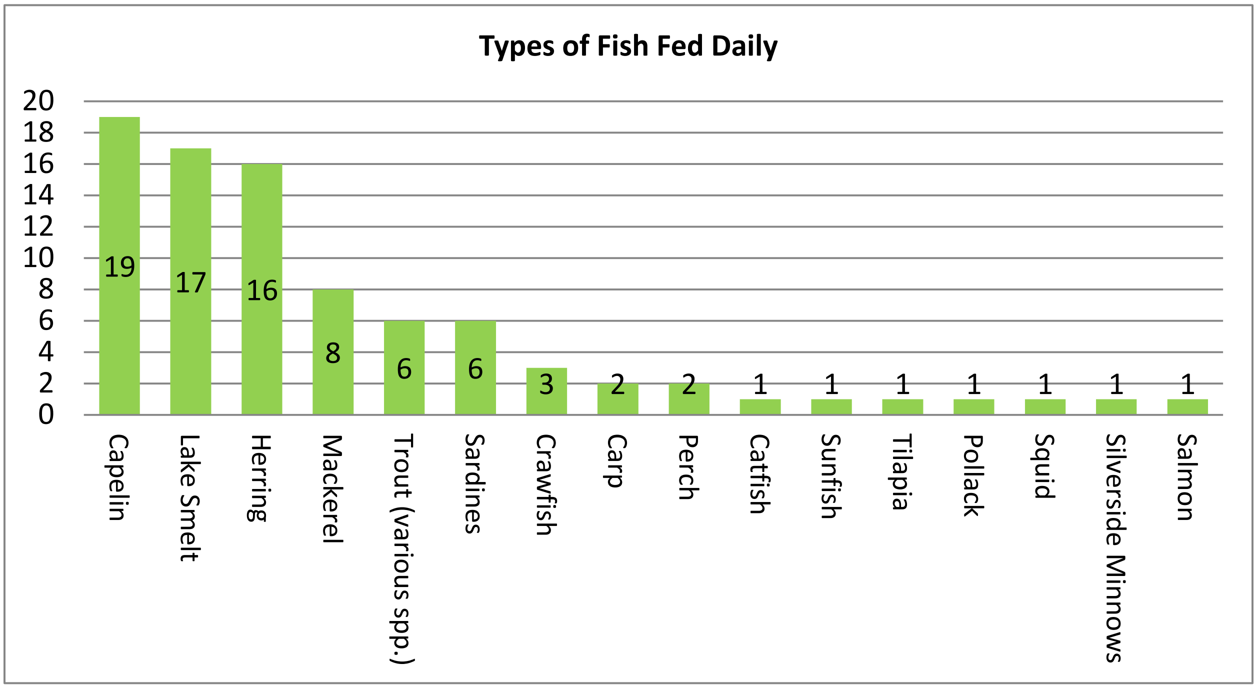
Diet Components (Fish: live and frozen; Meat: Nebraska and chicken; Commercially prepared food ranged from dog food to zoo prepared diets).



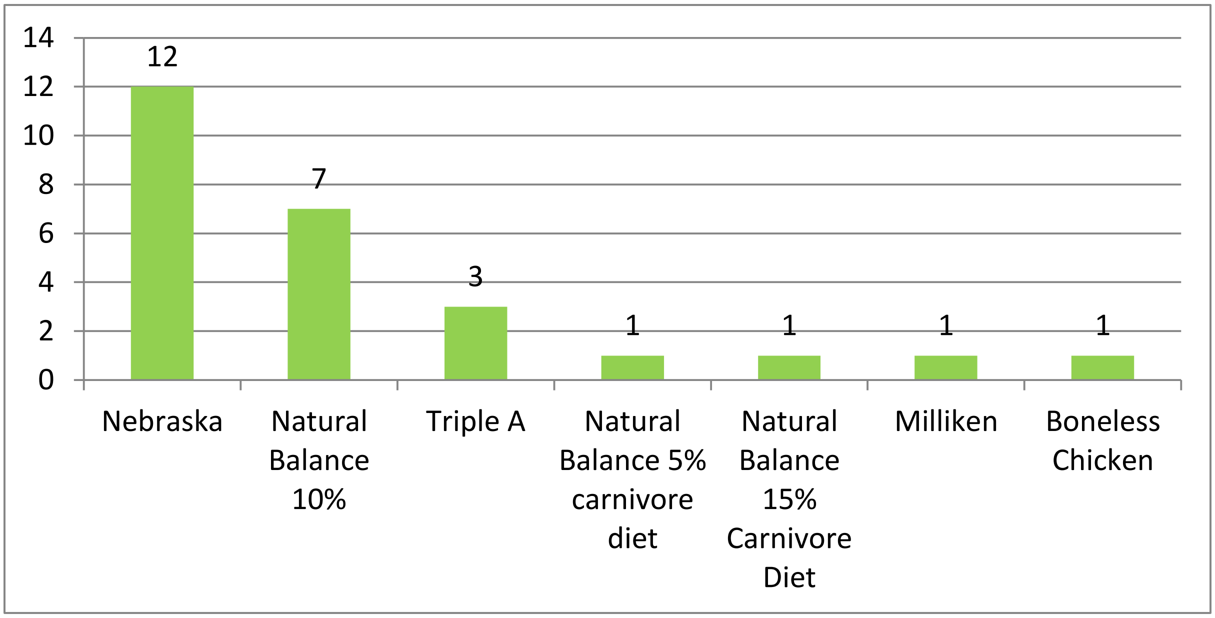
Diet Goals



Types of Fish Fed Regularly (n = 35 feed fish multiple times per week as part of diet)



Meat Products Offered (n = 25 that feed meat products as part of daily diet)



Commercially Prepared Diet Products (n = 20)

Cat Food:
Iams Feline Dry Food (2)
Blue Bonnet
Pro Plan Chicken and Rice Dry Food
Science Diet Hairball Diet
Iams Original Ocean Fish and Rice
Mazuri Exotic Feline Diet
Hills K/D canned cat food

Ferret Chow:
Mazuri Ferret Pellets (2)
National Mink Fur Pellets

Dog Food:
Purina Dog Chow (2)
Sport Mix
Science Diet Canine Maintenance Dry Food
Science Diet Canine Biscuits
EVO Dog Food
Mazuri Large Carnivore Diet

Other Prepared Diets:
Liers' Diet
Brave Meat Eater
Primate Biscuit

CHANGING NUTRIENT REQUIREMENTS

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

Pregnant/Lactating Female: A pregnant female's diet should be monitored closely and increased as necessary to maintain the dam's condition. The pregnant female requires more energy than the non-pregnant female. These requirements may increase up to 17 to 32% (Robbins 1993).

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 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.

In practice, during the last trimester of gestation and through lactation, diet increases of 30% to three times maintenance may be necessary to maintain adequate growth of the pups and body weight of the dam (Reed-Smith 1994). Energy requirements will differ based on numerous variables (exhibit size, environmental temperature, individual activity patterns, stage of gestation/lactation, etc.), thus it is recommended that goal weights be established, animals weighed on a regular basis, if possible, and diet adjustments made based on observed body weight changes.

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

Weight Loss: While otters should carry some body fat and not be kept artificially thin, they are prone to gaining excessive weight in captivity. Tarasoff (1974) reported subcutaneous fat deposits primarily at the base of the tail and caudally on the rear legs, with smaller deposits around the genitalia and in the axillary regions (this is the normal placement of fat stores with some deposits in these areas considered desirable). 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 otter groups around feeding time and the potential for this to increase when less food is offered.

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

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

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

Food Variability

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

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) or used only on an occasional basis. The overall nutrient and caloric intake, body weight of the animal(s), and condition of the animal(s) should be taken into consideration. All otters will benefit from receiving live fish/crayfish (from approved sources), at least as enrichment on a weekly basis. Whole fish should comprise a portion of the daily diet of all other species.

SPECIES-APPROPRIATE FORAGING AND FEEDING:

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

There also are a variety of puzzles and other feeding devices described in the literature that can be adapted for use in river otters. Alternatively, feeding tubes can be built into exhibits that randomly release live prey or food items into the exhibit. See Section 3, Chapter 11 for other enrichment items used, including non-food items.

Feeding Schedule

Due to their naturally nutrient dense diet, reliance on fat as a source of energy, rapid transit time of food through the intestinal tract, feeding style of frequent, small amounts, and generally high activity level – it is recommended that otters be fed at least twice a day and preferably three or more times daily (including enrichment or training feeds). 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. Timing of foraging opportunities and items offered should be varied to prevent habituation. All uneaten food should be removed before it spoils; this may be daily or more frequent in warm climates or seasons. (See below for a discussion of abnormal repetitive behaviors (ARBs) associated with feedings and training. At this time it is still recommended that more frequent feedings are preferable but, staff should make an attempt to 1) record ABRs in all otters and document if feedings are increased or reduced as a result of these, and 2) test the use of a "feeding cue" to distinguish feeding times from other staff visits to the enclosure.

ABNORMAL REPETITIVE BEHAVIORS (ARBS) ASSOCIATED WITH FREQUENT FEEDINGS

Morabito (Morabito & Bashaw 2012) conducted a two-part survey of 106 AZA institutions housing North American river otters; part one consisted of management and exhibit characteristic questions applicable to all otters resident in the enclosure, part two dealt with ARBs in individual otters. Their response rate was 52% representing 61 exhibits in 55 institutions and 129 individual otters (59.70.0). Their results are thought provoking and worthy of further research as they potentially represent a change in how we approach training as well as delivery of food and enrichment to this species. In summary, their findings indicate:

- 46% of the otters at respondent institutions were reported as exhibiting ARBs.
- Neither age nor sex were significant predictors of ARBs in the otters reported on.
- 21 otters were reported to exhibit more ARBs in winter versus 9 exhibiting more in summer (of these it appeared to be associated with increased public attendance).
- 30 otters were reported as exhibiting pre-feeding ARBs; 6 exhibited post-feeding ARBs.
- NARO are prone to developing ARBs, particularly before feeding.
- Most frequently these ARBs take the form of repetitive swimming or pacing.
- Frequent feeding and training were both reliable predictors of ARBs. (this could be because individuals who already exhibit ARBs are targeted for more feedings and/or training sessions.)
- Institutions that utilized feeding cues which served to notify the otters that food/enrichment would be coming reported slightly fewer ARBs in their otters, but this was not statistically significant.
- Their recommendations are:
    - ✓ Further study based on observational data versus self-reporting by individual institutions.
    - ✓ Looking at what types of exhibit designs reduce the effect of visitor attendance on ARBs.
    - ✓ Experimentally manipulating feeding and training frequency to determine if these changes cause, or are a result of changes in ARBs.
    - ✓ DO NOT recommend reducing number of feedings or training sessions at this time.
    - ✓ ADDING reliable cues before feeding/enrichment and training allowing the otters to distinguish between types of keeper visits to the enclosure should be explored further.
    - ✓ ADD or use feeding techniques that require active foraging.
    - ✓ MINIMIZE the amount of time otters are confined in a less preferred environment (e.g. holding), or, provide hiding places to reduce escape-motivated ARBs.

Other Nutritional Issues

USE OF SUPPLEMENTS

For Coat Maintenance

Over one half of the 1997 N. A. River Otter Husbandry Survey respondents indicated use of some type of supplement for maintenance of coat condition in North American river otters (Reed-Smith, 1997). Poor coat condition (i.e. dry, dull appearance, hair loss, etc.) can be a manifestation of multiple disorders (Muller, et al. 1983).

Parasitic or bacterial infections can cause poor coat condition in captive mustelids (Wallach and Boever 1983). Nutritionally, fatty acid deficiency, protein deficiency, vitamin A deficiency or toxicity, vitamin E deficiency, vitamin B complex deficiencies, vitamin C deficiency, or several mineral deficiencies can all be manifested in poor coat condition (Muller, et al. 1983). For this reason, the clinical signs of poor coat quality are crucial in determining the cause and treatment of poor coat quality. The diet should be analyzed. If deficiencies or toxicities are noted, and appear to be the cause of the observed coat condition, adjustments should be made. Acute treatment may be necessary if the insult has occurred for an extended period of time, however, if diet appears the principle cause, adjustment should be paired with that treatment.

For Thiaminase and Vitamin E Loss

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

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

The 1997 N.A. River Otter Husbandry Survey conducted by Central Park Zoo is still one of the most complete. As a matter of institutional memory, retained in this edition is the table listing supplements used at that time. Also provided is diet information collected between 2004 and 2009 and compiled by Jessica Foti (2010).

Dietary Supplements Offered (N = 34) (Foti 2010)

No vitamins offered (8)
Vit.B^1/Vit.E/glucosamine/Chondroitin/Manganese (5)
Thiamine paste/Vitamin E (4)
Pet Tab/Vit. B^1 & E (2)
Mazuri Vita Zu Marine Mammal Tab (3)
Mink Pellets (1)
Clovite (1)
Linatone Plus (1)
Mazuri Vita Zu Bird Tab/ Cod Liver Oil (1)
Sea Tab/Vit. C/Linatone (1)
Linatone Plus/Clovite (1)
Zu Vite (1)
Salmon Oil (1)
Derm Caps (1)
Pet Tab/Cod Liver Oil (1)
Cod Liver Oil (1)
Wheat Bran (1)

Dietary Supplements Offered (N=50) (1997 N.A. River Otter Husbandry Survey)

Supplements are listed as given by the submitting institutions. M = multi-vitamins/minerals, including Mazuri Vit. Blend, Vita Sol, & Chaparral Zoological Vit.; ST = Sea Tabs; P = Pet Tabs; Ca = Calcium; Vi = Vionate; Di = Diaglo S. A.; Cl = Clovite; De = Derm Cap; Os = Osteoform; Li = Lixotonic; Nu = Nutriderm; Lin = Linatone; Ve = Vegetable Oil; Cod = Cod Liver Oil; Br = Bran; Y = Yeast; Wh = Wheat Germ Oil; Ao = Anti-oxidants; D-Ca-Fos split in to D, Ca, Pho; No = None. * Indicates supplements given only sporadically.

Dietary Supplements Table

1997 N. A. River Otter Husbandry Survey

Inst.	B_1	E	B	M	ST	P	Ca	D	Pho	Di	Vi	Os	Li	Nu	Ve	Wh	Br	Cod	Lin	Cl	De	Ao	No
AK																			X*				
AR	X																						
AS					X																		
BA																							X
BG				X																			
BR		X		X																			
BU														X									
BV																							X
BW																							X
CA																				X			
CF	X	X																					
CG																							X
CM																					X	X	
CP	X	X																					
DA																							X
DE																							X
DI																		X					
EL																		X					
HO													X										
HR						X*					X												
JB																							X
KX																							X
LI																							X
LO				X														X					
LP	X																						
LR																							X
MI																							X
MP	X	X	X																				
MT	X	X																					
NC				X																			
NT				X																			
NZ	X															X		X					
OA										X							X						
OH																							X
PB											X	X											
RI																							X
SE																							X
SF																							X
SN	X																						
SP																			X				
ST																							X
SU																X	X						
SZ																							X
TS	X												X										
TT^		X					X	X	X						X					X		X	
TZ	X						X				X												
WO														X									
WP				X																			
ZA																							X
ZO	X																						

DENTAL PROBLEMS

Dental problems are of concern in otters. Merck's Veterinary Manual (1991) and Petrini (1992) suggest offering rib bones one to two times per week for maintenance of dental hygiene. Other options are crunchy dry foods and some whole prey items including fish. Whatever is selected, it is very important that these animals be given something hard to maintain good dental hygiene

ROUGHAGE

As the natural diet of otters may contain some roughage, Duplaix-Hall (1975) and others have stressed the need for roughage in the diet fed to otters. Fish and other whole prey items, and crunchy vegetables are some of the items added to supply this roughage.

NUTRITIONAL RELATED DISEASE

Few reports of calculi are documented, thus this does not appear to be a problem in this species.

Nutrient Overview

This section is not intended to explain in depth the science of nutrition but, to introduce the essential dietary elements. Without these essential nutrients in the required quantities, the animal will become ill, cease to reproduce, and depending on the severity of the deficiency, may die because of the deficiency. Though it is *very important to obtain essential nutrients from the diet, more is not better. There are also maximum* tolerances and safe levels for each nutrient. Some nutrients can be very toxic to certain animals while others may be harmful to a lesser extent (NRC, 1980). Nothing can be considered completely safe and all nutrients must be considered in relationship to interactions with other nutrients. (Robbins, 1993).

ENERGY

Protein, carbohydrate and fat all provide energy to the animal. When any of these is fed in excess to energy needs, the animal will become fat. It is difficult to determine in a practical manner, the energy requirement of any one animal. Energy utilization is a combination of basal metabolic rate (BMR), the energy it takes to digest food, and the energy needed for activity and reproduction (gestation and lactation), and the energy needed for growth (maximum cell division). There are some basic measurements which allow calculations for energy demand (Miller and Koes, 1988). BMR for many mammals equals 70 x Body mass in kg to 0.75 power however, Iversen determined that the BMR of otters can be expressed by the equation: $M = 84.6W^{0.78}(+0.15)$. M = basal metabolic rate in kcal/day and W = body weight in kg. *"This is about 20% higher than expected from the mammalian standard curve described by $M = 70\ W^{0.75}$."* (Iversen 1972; Toweill & Tabor 1982; Kruuk 1995; Estes 1989)

In controlled feeding experiments on captive *L. lutra*, Erlinge (1968) reported that the animals were satiated after consuming about 900 to 1000 grams of live food; Toweill (1982) cites unpublished data of his that, *"...recorded similar volumes of food in moderately distended northern river otter stomachs containing food."* Harris (1968) reports that otters in captivity required about 700 to 900 grams of food.

Klieber outlined energy requirements for a number of species. This is a simplistic approach to a complex issue (Thompson, 1996). Additionally, it is very difficult to determine energy needed for activity, since in many cases, every animal differs in its activity level. So far, the most non-invasive method to measure whether the diet is providing the appropriate amount of energy (measured in kcals) is to weigh the animal periodically and look for changes (Gettys et al., 1988). Additionally, if the diet consumed is accurately measured, one can calculate energy consumed.

PROTEIN AND AMINO ACIDS

Protein is comprised of amino acids (Robbins, 1993). The actual requirement for animals is for amino acids not protein as such. However all amino acids must be present in adequate amounts. Not all of the 20 common amino acids are required in the diet of all animals. Some animals can produce some amino acids to a greater or lesser extent from essential dietary amino acids. These are non-essential amino acids. Simple stomach species generally require ten essential amino acids: arginine, histidine, isoleucine, leucine, threonine, lysine, methionine, phenylalanine, tryptophan, and valine (Robbins, 1993). Cats, being obligate carnivores with simplistic GI-intestinal tracts, require an array of pre-formed amino acids from the diet with additional needs such as taurine (Morris, et al., 1991). The amino acid requirements of otters remains unknown.

Utilization of protein from the diet begins with digestion which depends on sequential cleavage of amino acids from the protein molecule. The cleavage is performed by digestive enzymes. There are many mechanisms for transport of these amino acids through the intestinal cells into the body. Once in the body, some amino acids are metabolized and converted as needed into other amino acids. As needed, proteins are made by the body and comprise muscle, hair, enzymes, hormones, etc. The requirement for protein in the diet is somewhat based on the animal's ability to digest whole protein and utilize protein from sources such as muscle meat, cereals and microorganisms. The animal must receive an adequate compliment of essential amino acids, regardless of the source (Nutrition Reviews, 1985).

Young, growing animals require more protein than adult animals. In general, adult carnivores require a dietary protein level of 18 to 30% whereas a weaned kitten needs approximately 35% and young mink or foxes require 25 to 38%. (Robbins 1993)

Often requirements are expressed as crude protein. This is actually a calculation based on an average nitrogen content of various proteins of (16% nitrogen). To determine crude protein content of a food item the nitrogen content is multiplied by 6.25 (or100/16). There is error in this estimate since not all plant or animal nitrogen is in the form of protein, for example chitin in insects (Robbins, 1993; Bernard and Allen, 1997b). Animal protein requirement estimates are difficult to make because the quality of dietary protein depends on its amino acid composition, the ratio of protein to energy, and the total amount of food consumed. Additionally, amino acids if not needed as body protein, can be broken down and used as an energy source. Thus, protein needs and energy should be considered together (Robbins, 1993).

WATER

Water is sometimes the "forgotten" nutrient but the most important. Water is needed by all animals. Some animals need continuous supplies of drinking water. For safety sake, fresh water should be available at all times. Many otter exhibits have potable water provided via running water and/or pools. Some institutions offer water in separate tubs for drinking (Maslanka and Crissey, 1998). Neonatal mammals have a water concentration of between 71 and 88% of their body weight. Adult animals of normal weight have a concentration of between 50 and 65% (Robbins 1993).

MINERALS

Minerals have been classified into two groups: major (or macro) minerals and trace (micro) minerals. Many minerals, approximately 26, are known to be essential for life. It is not known why certain elements are essential for life while others may not be. It is quite possible that future study will find links and requirements for other minerals. All elements are toxic if ingested or inhaled at sufficiently high levels for long enough periods (NRC, 1980). Sometimes there is a relatively fine line between the biological level of need for a mineral and a toxic level. It should be remembered that the level of a mineral found in any source is, in part, directly related to soil. Plants for example can provide deficient or excess levels of minerals such as selenium depending on the soil in which the plant is grown. Various geographic regions are known for their deficiencies or excesses of minerals (Maynard, et al., 1979).

The major elements are: Carbon, hydrogen, oxygen, nitrogen, sulfur, calcium, phosphorous, potassium, sodium, chlorine, and magnesium. Since carbon, hydrogen, oxygen, nitrogen, and sulfur are major constituents of many molecules, these are not considered with respect to essential nutrient intake. However, calcium, phosphorous, potassium, sodium, chlorine, and magnesium are considered essential dietary nutrients. This category contains minerals present in large quantities in the body (Maynard, et al., 1979; Robbins, 1993).

The trace elements considered in nutrition are: iron, zinc, copper, manganese, nickel, cobalt, molybdenum, selenium, chromium, iodine, fluorine, tin, silicon, vanadium, and arsenic. Each of these is needed in very small amounts. The role of some of these in metabolism is not fully determined. Interactions among minerals are many and each should be considered in light of the other (Miller, et al., 1991).

The form of the mineral in the diet affects its absorption. Some forms are absorbed to a much greater extent than others. For example, it is thought that iron is not absorbed well from the GI-tract, in general. Inorganic sources may be absorbed at only about 5-15%. Heme sources however (associated with animal tissues) are better absorbed. Additionally, many factors may affect absorption of minerals. The example with iron shows that dietary vitamin C and sugars can increase inorganic iron absorption (Smith, 1997). Additionally some animals may have evolved ways to obtain nutrients when evolving either in nutrient toxic or deficient habitats and may not fall in line with the general statements (Kincaid and Stoskopf, 1987).

VITAMINS

Vitamins have been defined as a group of organic substances present in minute quantities in foods which are essential to normal metabolism and lack of which in the diet causes deficiency diseases (Robbins, 1993). Scientists have categorized vitamins as fat soluble and water soluble. Fat soluble vitamins can be stored in the body whereas water soluble vitamins have very limited storage and when fed in excess are primarily excreted (Machlin, 1984). As we learn more about vitamins, we find that animals utilize different forms of the vitamins differently. This can have a profound effect on nutritional status (Papas, et al., 1991).

The fat soluble vitamins are A, D, E, and K. Since these vitamins are stored by the body, toxicities may occur when fed in excess. Vitamin E is considered relatively non-toxic while vitamins A and D are known to cause toxicity symptoms in animals studied (Rucker and Morris, 1997). The toxicity of vitamin K depends on the source of vitamin K utilized.

The water soluble vitamins are vitamin C and "B" vitamins, including Thiamin, Riboflavin, Niacin, Biotin, Pantothenic Acid, Pyridoxin, Folic acid, Vitamin B12, Choline and Carnitine. These vitamins are utilized by the body for many metabolic processes. Each acts in a different way to provide for normal function of metabolism. Since these vitamins are not usually stored in the body for long periods, it is felt that daily intakes are preferred. Excess of these substances are usually excreted in the normal healthy animal and thus are considered somewhat non-toxic (Machlin, 1984; Rucker and Morris, 1997).

Fat-soluble Vitamins – General Functions & Reported Deficiency Symptoms

Vitamin	Major Functions	Deficiency Signs
A (Retinol, retinal, and retinoic acid)	In eye pigment For maintenance, differentiation & proliferation of epithelial tissue..	Reduced fertility or sterility, birth defects, reduced growth or loss of weight, oral & nasal pustules, weakness, night blindness, impaired eyesight because eye problems, bone and teeth deformities, unsteady gait and incoordination, ruffled-droopy appearance.
D (D_2-ergocalciferol; D_3-cholecalciferol	Needed for calcium absorption and metabolism.	Rickets in young, osteomalacia in adults.
E (Tocopherol)	Antioxidant functions	Yellow fat disease (steatitis) sudden death with stress, lumpiness of subcutaneous fat, severe edema, nutritional muscular dystrophy, severe hemolytic anemia, reproductive failure
K (Phylloquinone & menaquinone)	Necessary for blood clotting	hemorrhaging.

(Adapted from: Robbins 1993)

Two vitamins are especially important when animals are fed a large proportion of fish in the diet. This is because these vitamins degrade relatively quickly in killed fish. Diets that contain high levels of marine products may not only predispose individuals to thiamin deficiency, but also to vitamin E deficiency if not adequately supplemented (Engberg, et al 1993).

Thiamin –fish eating animals including dolphins, polar bears, mink, foxes, sea lions, grebes, and gulls have been reported to have had thiamin deficiencies (Robbins, 1993). Many species of fish and shellfish contain the enzyme group called thiaminases that breaks down thiamin in the killed fish (Robbins 1993; Bernard and Allen, 1997a) Robbins (1993) also reports that thiaminase occurs in newly hatched chicks.

Supplementing otters consuming a large proportion of fish in their diet should be performed to ensure adequate thiamin status. The recommended supplementation regime for fish eating animals is 25-30 mg of thiamin per kg of fish fed (fresh weight basis; Bernard and Allen, 1997a)

Vitamin E –Vitamin E deficiencies are most frequently seen in species fed fish-based diets. Marine products contain high levels of poly- and mono-unsaturated fatty acids. Because vitamin E functions as an antioxidant, the breakdown of these oils during storage causes vitamin E destruction. For this reason, it is recommended that a vitamin E supplement be fed to otters on a fish only diet or a diet comprised of a large proportion of fish. (Crissey & Maslanka contribution IUCN Otter Specialist Group Otter Action Plan, in press) The recommended supplementation regime for fish eating animals is 100 IU of vitamin E per kg of fish fed (fresh weight basis; Bernard and Allen, 1997a). Similarly, the recommended supplementation level is 400 IU vitamin E / kg of *dry* diet (Engelhardt and Geraci 1978).

If a fish-based only diet is offered, supplementation with a multivitamin is recommended. Nutritional deficiencies have been reported in commercially farmed mustelids (hypovitaminoses A and E, thiamin deficiency, calcium deficiency; Wallach and Boever 1983, NRC 1982). Fish composition can change based on species, season of harvest, duration of storage, etc., and addition of multivitamin may provide some consistency in the nutrients contained in the diet. However, the diet should be analyzed to determine nutrient levels prior to supplementation in order to minimize the chance of over-supplementation and toxicity (especially of fat-soluble vitamins).

Water-soluble vitamins – General Functions & Reported Deficiency Signs Table

Vitamins	Major Functions	Deficiency Signs
Thiamin (vitamin B_1)	Needed in carbohydrate metabolism.	Anorexia, weight loss, weakness, lethargy, unsteady gait, diarrhea, seizures, and other neurological disorders, “star-gazing”.
Riboflavin (vitamin B_2)	Needed in carbohydrate metabolism	Anorexia, weight loss, poor hair coat, atrophy of hair follicles, diarrhea, leg paralysis, reduced fertility.
Niacin (nicotinic acid & nicotinamide)	Functions in metabolism (in NAD and NADP)	Retarded growth, , anorexia, diarrhea, dermatitis, drooling & tongue discoloration,
Vitamin B_6 (pyridoxine, pyridoxamine, & pyridoxal)	Needed in protein metabolism	Testicular atrophy, sterility, anorexia, retarded growth, poor hair coat, muscular incoordination, neurological disorders.
Pantothenic acid	Needed for fat, carbohydrate, & amino acid metabolism	Skin lesions, crusty scabs about the eyes, emaciation, degeneration of the liver, kidney problems, reproductive failure, death.

Water-soluble vitamins – General Functions & Reported Deficiency Signs Table		
Vitamins	**Major Functions**	**Deficiency Signs**
Biotin	Needed in metabolism	Fur discoloration, hair loss, degenerative changes in the hair follicles, thickened & scaly skin, conjunctivitis,
Folicin (folic acid)	Needed in metabolism	Anorexia, retarded growth, diarrhea, profuse salivation, convulsions, adrenal hemorrhages, , anemia
Vitamin B_{12} (cyanocobalamin)	Needed in carbon & carbohydrate metabolism	Anorexia, weight loss, pernicious anemia neurological & locomotion disorders.
Choline	Nerve functioning	Liver damage, reduced growth of the leg bones, awkward gait, growth retardation, weakness, lowered hematocrit.
Vitamin C (ascorbic acid)	Not required by many species. Needed for bone & collagen formation	Scurvy, severe necrotic stomatitis, anorexia, weight loss, gingivitis, glossitis, pharyngitis, hemorrhages

(Adapted from: Robbins 1993)

FAT AND ESSENTIAL FATTY ACIDS (EFAS)

Fats and oils (lipids) are utilized by animals as energy. Lipids are comprised mainly of glycerol and fatty acids. The energy value of fat is considered to be at least double that of protein or carbohydrates. Dietary fat is also the source of essential fatty acids which are required by animals. Thus the dietary requirement for fat in the diet is actually an energy requirement and the requirement for essential fatty acids. In general, the essential fatty acids include linoleic, linolenic and arachidonic acids or their metabolic derivatives. Some animals may have specific needs different from others (Rouvinen and Niemela, 1992; Stanton, et al., 1989). Essential fatty acids can be converted in the body to other important fatty acids. Some animals can convert these essential dietary fatty acids better than other animals. Obligate carnivores such as cats require not only linoleic acid but also, pre-formed arachidonic acid as they cannot convert the other essential fatty acids to this nutrient.(Burger, 1993). This may apply to otters too.

Much research is being conducted with respect to fatty acids. Of special interest is the work being done with omega-3 fatty acids in both humans and domestic animals. This work could prove very important to fish eating animals in particular (March, 1993).

Deficiencies of essential fatty acids include poor reproduction, kidney problems, poor wound healing, poor coat condition, and possibly dehydration, liver degeneration, and immune system failure.

Arachidonic acid is present in animal matter but not in plants. Good sources of arachidonic acid are meat, liver, and some seafoods. Linolenic acid is found in linseed, soybean, rapeseed oils, and marine fish oils. Omega-3 fatty acids are found in marine fish as well as trout. Oily fish, such as sardines, anchovies, and herring, are much better sources of EFAs than non-oily fish (Robbins, 1993)

CARBOHYDRATES

Sugars, starches and fiber are carbohydrates. Carbohydrate is broken down to simple sugars before it is absorbed from the GI-tract in to the body. It is utilized by animals for energy. Some animals possess the ability to utilize different sources of carbohydrate to a greater or lesser extent (Yokota, et al., 1992; Kienzle, 1993). Carbohydrates play a minor role in the nutrition of otters (Wallach & Boever 1983). Animals that harbor large quantities of microorganisms in their GI-tract, like ruminants, can digest fiber to a greater extent than those with a more simple tract like most carnivores (Bonhomme-Florentin, 1990; Graham and Aman, 1991).

There is, in general, no definitive requirement for carbohydrate, including fiber, in the diets of animals. However, it is well know that fiber allows proper functioning of the GI-tract for many animals and should be included in the diet based on the species and its GI-tract morphology (Shaver, et al., 1988; Milton and Demment, 1988).

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CHAPTER 10 Health Care

Introduction

This section is intended as an overview of health care, not as a guide to diagnosis or treatment. The intent is to help the keeper, curator or veterinarian with no otter experience understand the basic health needs of captive river otters and some of the diseases these animals are susceptible to. (For the reader's convenience, there is a limited glossary of medical terms used at the end of this chapter.)

As is true for many small mammals, otters may be hit with serious disease with little or no warning. The only way to mitigate a potentially life threatening illness is through keeper awareness and familiarity with the animals in their charge. Always take the time to observe, and record, how the animals are behaving; eating; performing bodily functions; using their exhibit; interacting with exhibit mates, and their general physical appearance. A good daily record keeping system can be vital if an animal becomes ill; or, the attention to detail can alert you to a problem before it becomes too severe. If an animal is exhibiting signs of illness call the veterinarian, do not treat it without professional assistance.

Some of this material is generalized to all mustelids and its application to river otters is assumed. For additional information the bibliography contains a number of excellent references.

Weight Ranges

4.5kg – 11.3kg. (10 lbs. – 25 lbs.) Harris 1968
5kg. – 15kg. (11 lbs. – 33 lbs.) Hall 1981

Melquist & Hornocker (1983) found that adult males, on average, were 17% heavier than adult females. They cite an average weight of 7.9 kg. (17.4 lbs.) for females. Every animal will have its own "good weight". Diets should be adjusted as needed to maintain a healthy weight and normal activity pattern. (Not every female should weigh the average and not all males are larger than females.) See Chapter 7, Animal Management, Weight; for weight range photos.

Weights of Captive N. A. river otters (*Lontra canadensis*) (ISIS, 1999)

Weight	Units	Mean	St. Dev.	Minimum Value	Maximum Value	Sample Size[a]	Animals[b]
Weight: 0-1 days age	Kg	.1242	.0147	.0950	.1450	12	10
Weight: 0.9-1.1 months age	Kg	.8669	.1488	.6890	1.160	8	8
Weight: 5.4-6.6 months age	Kg	6.038	1.315	3.900	8.020	10	10
Weight: 1.8-2.2 years age	Kg	8.701	1.629	4.540	10.75	21	11
Weight: 2.7-3.3 years age	Kg	9.033	2.115	5.780	12.50	32	15
Weight: 4.5-5.5 years age	Kg	10.68	1.61	6.818	13.18	21	8

[a] Number of samples used to calculate the reference range.
[b] Number of different individuals contributing to the reference values.

Life Span

In the wild otters live a maximum of about 10 to 13 years. Mortality rates for wild otters increase at three to five years, the reasons for this are unknown (Polechla 1989). Historically, longevity in captivity is given as 25 years (Melquist & Dronkert 1987), and 23 years (Park 1971, Nowak 1991). While these figures are supported by an entry of a 25 year old animal in the N. A. river otter studbook, the median age is 12.3 years with lifespans of 16 to 20 years fairly common (D. Hamilton personal communication).

Physiological Norms

Heart Rate

137 – 170 beats/minute (Grassmere Wildlife Park now Nashville Zoo @ Grassmere)
130 – 178 beats/minute; baseline = 152 beats/minute (Spelman 1999)
160 – 180 beats/minute; baseline = 174 ± 9 beats/minute (Hoover 1986)

EKG

From: Hoover 1986

Rhythm: Normal sinus rhythm
Mean Electrical Axis (frontal): 54 ± 13°

Lead II	Mean ± SD	Range
P (sec)	0.051 ± 0.006	0.040 – 0.060
PR (sec)	0.086 ± 0.008	0.075 – 0.095
P (millivolt)	0.24 ± 0.6	0.15 – 0.30
QRS (sec)	1.82 ± 0.48	1.30 – 2.60
QT (sec)	0.184 ± 0.011	0.170 – 0.200
T (millivolt)	0.45 ± 0.11	0.30 – 0.60

Respiration Rate

When under anesthesia, the important factor is that the mucous membranes and mouth stay a pink color.
Grassmere (Nashville Zoo) records show a respiratory rate range of 30 – 60 while under anesthesia.

Range: 10 – 60 breaths/minute; baseline = 31 breaths/minute (Spelman 1999)
Range: 20 - ~34 breaths/minute (Hoover & Jones 1986) These figures were obtained from otters during chemical immobilization and inhalation anesthesia.

Body Temperature

Body temperature range of 37.5° to 40°C (99.5° to 104°F) for otters involved in a translocation project (Serfass (1994). (The upper end of this spectrum should be considered pathologic if it continues very long. An animal's normal temperature may reach this height after the exertion and stress associated with being caught.)

Body temperature range: 38.1 – 38.7°C (100.6 to 101.7°F); baseline = 38.4°C (101.1°F) (Spelman 1999)

Body Temperature of captive N. A. river otters (*Lontra canadensis*) during immobilization (ISIS, 1999)

Test	Units	Mean	St. Dev.	Minimum Value	Maximum Value	Sample Size[a]	Animals[b]
Body Temperature:	°F	102.0	1.8	96.8	105.8	161	96

[a] Number of samples used to calculate the reference range.
[b] Number of different individuals contributing to the reference values.

ARTERIAL BLOOD PRESSURE

Mean arterial blood pressure range: 31 – 77 mm Hg; baseline = 63 mm Hg (Spelman 1999)

BLOOD VALUES

Captive Animals

Hematology Values for Captive North American River Otters (*Lontra canadensis*) (ISIS, 1999)

Test	Units	Mean	St. Dev.	Minimum Value	Maximum Value	Sample Size[a]	Animals[b]
WHITE BLOOD CELL COUNT	*10^3/µl	7.935	4.138	1.980	32.30	236	130
RED BLOOD CELL COUNT	*10^6/µl	9.96	1.38	6.62	14.30	175	95
HEMOGLOBIN	g/dl	14.2	1.7	9.2	19.2	208	121
HEMATOCRIT	%	44.8	5.8	30.5	64.2	240	132
MEAN CORPUSCULAR VOLUME	fL	46.6	5.2	30.8	67.6	173	95
MEAN CORPUSCULAR HEMOGLOBIN	pg/cell	14.7	1.5	10.1	23.1	171	95
MEAN CORPUSCULAR HEMOGLOBIN CONCENTRATION	g/dl	31.8	2.3	23.5	41.3	206	121
PLATELET COUNT	*10^3/µl	489	137	190	810	74	45
NUCLEATED RED BLOOD CELLS	/100 WBC	0	0	0	1	25	19
RETICULOCYTES	%	0.3	0.2	0.0	0.9	19	16
SEGMENTED NEUTROPHILS	*10^3/µl	5.563	3.206	0.191	19.10	215	121
LYMPHOCYTES	*10^3/µl	1.606	0.935	0.035	5.630	224	126
MONOCYTES	*10^3/µl	0.214	0.227	0.000	1.292	185	111
EOSINOPHILS	*10^3/µl	0.536	0.699	0.000	4.608	194	117
BASOPHILS	*10^3/µl	0.021	0.044	0.000	0.182	36	28
NEUTROPHILIC BANDS	*10^3/µl	0.384	0.743	0.000	3.060	57	44
ERYTHROCYTE SEDIMENTATION RATE	mm/Hr	0	0	0	0	1	1

[a] Number of samples used to calculate the reference range.
[b] Number of different individuals contributing to the reference values.

Physiological reference ranges are calculated from samples submitted by 34 member institutions and are both sexes and all ages combined.

Biochemistry Values for Captive North American River Otters (*Lontra canadensis*) (ISIS, 1999)

Test	Units	Mean	St. Dev.	Minimum	Maximum	Sample Size[a]	Animals[b]
CALCIUM	mg/dl	8.8	0.7	7.4	11.2	212	122
PHOSPHORUS	mg/dl	5.6	1.6	2.2	12.3	202	118
SODIUM	mEq/L	150	4	139	164	204	116
POTASSIUM	mEq/L	4.3	0.5	3.0	5.9	207	117
CHLORIDE	mEq/L	114	4	97	128	200	116
BICARBONATE	mEq/L	23.5	2.4	19.0	28.0	28	25
CARBON DIOXIDE	mEq/L	23.7	3.5	15.0	30.0	75	42
OSMOLARITY	mOsmol/L	303	14	287	328	14	11
IRON	µg/dl	175	62	77	359	48	32
MAGNESIUM	mg/dl	1.59	0.55	0.65	2.50	23	16
BLOOD UREA NITROGEN	mg/dl	28	8	13	56	220	126
CREATININE	mg/dl	0.6	0.2	0.3	1.8	205	115
URIC ACID	mg/dl	1.9	0.7	0.0	4.3	123	81
TOTAL BILIRUBIN	mg/dl	0.3	0.2	0.0	1.0	198	116
DIRECT BILIRUBIN	mg/dl	0.1	0.1	0.0	0.3	52	32
INDIRECT BILIRUBIN	mg/dl	0.2	0.1	0.0	0.4	51	32
GLUCOSE	mg/dl	96	33	35	247	218	123
CHOLESTEROL	mg/dl	233	67	99	421	213	123
TRIGLYCERIDE	mg/dl	47	37	4	201	136	81
CREATINE PHOSPHOKINASE	IU/L	509	446	99	2613	81	55
LACTATE DEHYDROGENASE	IU/L	524	1051	24	6200	129	83
ALKALINE PHOSPHATASE	IU/L	83	43	17	279	215	122
ALANINE AMINOTRANSFERASE	IU/L	106	67	32	449	196	112
ASPARTATE AMINOTRANSFERASE	IU/L	93	49	20	334	202	117
GAMMA GLUTAMYLTRANSFERASE	IU/L	12	11	3	83	102	65
AMYLASE	U/L	14	22	0	105	66	44
LIPASE	U/L	27	25	0	98	29	21
TOTAL PROTEIN (COLORIMETRY)	g/dl	6.6	0.6	5.2	8.3	205	119
GLOBULIN (COLORIMETRY)	g/dl	3.6	0.6	2.2	6.0	171	105
ALBUMIN (COLORIMETRY)	g/dl	2.9	0.3	2.0	3.8	171	105
ALBUMIN (ELECTROPHORESIS)	g/dl	2.8	0.1	2.7	2.8	2	2
TOTAL THYROXINE	µg/dl	2.1	0.1	2.0	2.2	3	3

[a] Number of samples used to calculate the reference range.
[b] Number of different individuals contributing to the reference values.

Physiological reference ranges are calculated from samples submitted by 34 member institutions and are both sexes and all ages combined.

Wild animals

Hematology values for adult live-trapped river otters (*Lutra canadensis*) (Tocidlowski, 1997)					
Test	Units	Median	Minimum Value	Maximum Value	Sample Size
WHITE BLOOD CELL COUNT	*10^3/µl	11.3	4.7	33.2	132
RED BLOOD CELL COUNT	*10^6/µl	10.99	6.10	14.50	132
HEMOGLOBIN	g/dl	15.1	10.4	19.0	132
HEMATOCRIT	%	47.6	32.2	60.8	132
MEAN CORPUSCULAR VOLUME	fL	43.3	38.3	49.0	132
MEAN CORPUSCULAR HEMOGLOBIN	pg/cell	13.7	11.3	15.8	132
MEAN CORPUSCULAR HEMOGLOBIN CONCENTRATION	g/dl	31.4	27.8	39.2	132
PLATELET COUNT	*10^3/µl	565	298	931	132
SEGMENTED NEUTROPHILS	*10^3/µl	8878.5	3003.0	28220.0	132
LYMPHOCYTES	*10^3/µl	1254.0	123.0	4950.0	132
MONOCYTES	*10^3/µl	452.3	52.0	2380.0	132
EOSINOPHILS	*10^3/µl	312.0	0.0	1833.0	132
BASOPHILS	*10^3/µl	88.0	0.0	219.0	132
NEUTROPHILIC BANDS	*10^3/µl	94.0	0.0	486.0	132

Biochemistry values for adult live-trapped river otters (*Lutra canadensis*) (Tocidlowski, 1997)					
Test	Units	Median	Minimum Value	Maximum Value	Sample Size
CALCIUM	mg/dl	8.4	6.8	10.0	50
PHOSPHORUS	mg/dl	5.8	3.2	8.3	50
SODIUM	mEq/L	152	136	158	50
POTASSIUM	mEq/L	4.4	3.5	5.3	50
CHLORIDE	mEq/L	113	94	121	50
CARBON DIOXIDE	mEq/L	24	19	28	21
BLOOD UREA NITROGEN	mg/dl	31	17	56	50
CREATININE	mg/dl	0.5	0.4	0.8	50
TOTAL BILIRUBIN	mg/dl	0.2	0.1	0.5	50
GLUCOSE	mg/dl	130	56	225	50
CHOLESTEROL	mg/dl	152	63	279	29
TRIGLYCERIDE	mg/dl	31	9	72	29
CREATINE PHOSPHOKINASE	IU/L	219	67	1300	50
LACTATE DEHYDROGENASE	IU/L	149	36	10820	21
ALKALINE PHOSPHATASE	IU/L	85	29	282	50
ALANINE AMINOTRANSFERASE	IU/L	194	46	990	50
ASPARTATE AMINOTRANSFERASE	IU/L	85	34	1260	50
GAMMA GLUTAMYLTRANSFERASE	IU/L	19	8	38	29
AMYLASE	U/L	12	2	22	21
TOTAL PROTEIN (COLORIMETRY)	g/dl	7.3	5.7	9.0	50
GLOBULIN (COLORIMETRY)	g/dl	4.0	2.9	5.8	50
ALBUMIN (COLORIMETRY)	g/dl	3.3	2.4	4.1	50

Hematology/Blood Chemistry – Serfass, 1994 – Pennsylvania River Otter Reintroduction

Range	
Hematocrit gm/dl	34 – 59%
Red Blood Cell Count	9.4 – 11.9/mm
Segmented Neutrophils	61 – 87%
Basophils	0
Mean Corpuscular Hemoglobin (MCH)	14.2 – 16.9pg
Blood Urea Nitrogen (BUN)	26. 45 mg./dl
BUN/Creatinine mg/dl ratio	41 – 130
Triglyceride meg/l	20 – 128 mg/dl
Total Protein gm/dl	6.3 – 7.2 gm/dl
Globulin	3.4 – 4.3 gm/dl

Range	
Hemoglobin	13.0 – 18.5
WBC	7.0 – 16.1/mm
Band Neutrophils	0 – 5%
Lymphocytes	5 – 20%
Glucose	74 – 148 mg/dl
Creatinine	0.3 – 0.6 mg/dl
Cholesterol	108 – 242
Sodium	148 – 153
Albumin	2.7 – 3.0
Alkaline Phosphatase	81 – 193 I.U./L

Range	
Alanine Aminotransferase (ALT)	77 – 321 I.U./L
Aspartate Aminotransferase (SGOT)	0 – 1053 I.U./L
Monocytes	0 – 8%
Mean Corpuscular Volume (MCV)	38.6 – 52.3 u3
Potassium	3.9 – 4.9 meg/L
Bicarbonate	21 – 25 meg/L
Total Bilirubin	0.1 – 0.2 mg/dl
Ionized Calcium	3.9 – 4.1 mg/dl

Range	
Lactate Dehydrogenase (LDH)	73 – 390 I. U./L
Uric Acid	1.3 – 3.7 ug/ml
Eosinophils	0 – 14%
Mean Corpuscular Hemoglobin (MCHC)	29.3 – 40.2%
Chloride	108 – 114 meg/L
Iron	71 – 192 meg/L
Calcium	8.1 – 9.1 mg/dl
Phosphorus	4.5 – 7.5 mg/dl

Physical Norms

TEETH

3/3 Incisors; 1/1 Canines; 4/3 Premolars; 1/2 Molars x 2 = 36 Total

VERTEBRAE

14 rib bearing; 7 cervical; 14 thoracic; 6 lumbar; 3 sacral; 22 caudal. The normal total is 52. (Toweill & Tabor 1982)

MAMMAE
Four, inguinal.

FEET
Webbing between all digits but, slightly more extensive on the hind feet. The claws are sharp and probably aid in gripping. The hind feet are generally larger than the fore and the hind legs are longer leading to the typical hump-backed gait when traveling across land. The soles of the feet have tufts of hair under the toes (in some subspecies). Plantar pads are found on the soles of the hind feet. Pentadactyl and plantigrade. Also see skeletal adaptations under Descriptions.

Captive Care

MEDICAL RECORDS
Thorough and accurate medical records are essential to learn and understand more about the medical problems of any of our 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 can be easily transmitted from one institution to the next is extremely beneficial. The 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.

IDENTIFICATION

Transponder Chips:
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 (Photo: G. Myers), SQ/IM in the intrascapular area at the base of the ears, and many institutions have placed them between the scapulae. Placement in all of these areas has been met with success and failure (migration, loss, unable to read them as planned). At this time, the AZA Otter SSP recommends the forehead area as the preferred area of placement; 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.

Tattoos:
River otters should have their studbook number tattooed on the medial thigh as soon as they reach adult size. This should be applied to the left medial thigh for females and to the right medial thigh for males.

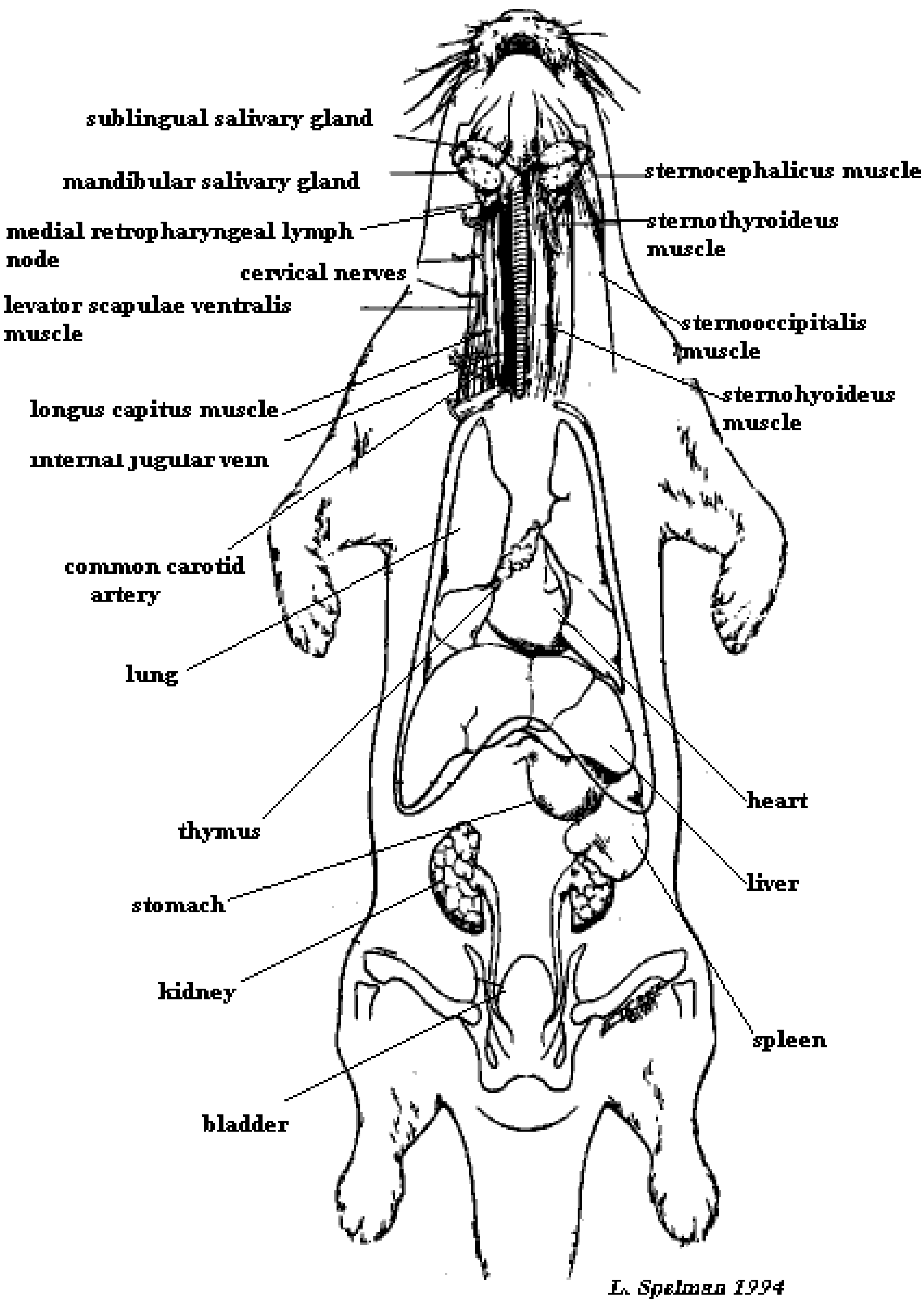
PREVENTATIVE HEALTH CARE

Annual physical examinations

It is recommended that all animals have at least a biannual examination and, if possible, an annual examination 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. 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.
- An annual fecal examination should be performed to check for internal parasites, and anthelmintics administered if necessary (see 'parasite control' section).
- Vaccines updated if necessary (see 'vaccination' section).

General Otter Anatomy



IMMUNIZATIONS

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.

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 Health Corporation, P.O. Box 3113, Omaha, NE 68103), a modified-live canine distemper vaccine of primate kidney tissue cell origin, Onderstepoort type.

Safety and efficacy of canine distemper vaccinations in exotic species of carnivores have been problematic. Vaccine-induced distemper has occurred in a variety of mustelids using modified-live vaccine, and killed vaccines have not provided long-lived protection and are not commercially available. However, to date there have been no cases of vaccine induced distemper in otters given the Galaxy product, and excellent seroconversion following vaccination using this product has been documented in young N.A. river otters (K.Petrini, unpublished data, Petrini et al. 2001). The use of any modified-live canine distemper vaccine in exotic species should be done with care, especially with *P. brasiliensis*, young animals, and those that have not been vaccinated previously. The use of PureVax™ Ferret Distemper Vaccine is recommended where possible.

Parvovirus

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

Rabies

The efficacy of rabies vaccines has not been proven in *Lontra canadensis* or other exotic mustelids. Vaccinated otters that bite humans should not be considered protected from rabies. **Only killed** rabies products should be used in otters. One commonly used product is Imrab® 3 (Merial Ltd., 3239 Satellite Blvd., Duluth, GA 30096), which is a killed rabies vaccine that has been used extensively in small carnivores without apparent adverse effects. Otters should be given 1ml of vaccine IM once at 16 weeks of age followed by a yearly booster. Although new rabies glycoprotein, canarypox vector vaccinations have recently been introduced to the market, these vaccines have not been used widely in exotic carnivores and their safety has not been determined at this time.

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.

Vaccination schedule

AGE	Canine Distemper	Feline Panleukopenia	Rabies	Leptospirosis
8 weeks	X	X		
12 weeks	X	X	X	(X)
16 weeks	X	X		(X)
Adult	X	X	X	(X)

- Sub adults should be vaccinated with killed parvovirus at 8, 12, and 16 weeks. Vaccination should begin earlier in pups from unvaccinated dams**.** (Photo: G. Myers)
- Veterinarians should **consider** vaccinating sub adults at 8, 12, and 16 weeks for distemper. **See discussion of distemper vaccines above.**
- Rabies vaccine should be given once at 8 or 16 weeks (product dependent) for animals at risk of contracting rabies.
- Adults should be vaccinated annually for all of the above.

PARASITE CONTROL

Otters should have fecal examinations performed regularly. The frequency of these examinations depends on the incidence of parasitism in the geographic region and the animals' likelihood of exposure. Animals should also be screened for parasites before shipment and during quarantine. Fecal testing should include both a direct smear examination as well as fecal flotation and sedimentation techniques. 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. External parasites such as ear mites, fleas, ticks, etc. can be detected during a physical examination. A list of some of the parasites that have been identified in river otters and other mustelids is included in the disease section of this chapter for reference. See also Appendix A for parasites (endo- and ecto-) found in otters.

Recommendations for Parasite Testing

Otter parasite testing protocols

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

Parasite Treatment

Recommended anthelmintic treatments for otters (G. Myers, DVM; Otter SSP Veterinary Advisor)

Treatment	Dose
Fenbendazole	50 mg/kg PO sid X 3-5 days
Pyrantel pamoate	10 mg/kg PO sid; repeat in 2 weeks
Ivermectin	For heartworm prevention 0.024 mg/kg PO q 30 days For GI nematodes 0.2-0.4 mg/kg PO or SQ
Praziquantel	5mg/kg SQ or orally

Upon completion of treatment, fecal exams should be repeated to assure that the therapy was successful; two to three weeks after cessation of treatment, and repeated in two weeks.

Heartworm

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).

Heartworm Positive Test

According to the AZA Otter SSP Veterinary Advisor, Dr. Gwen Myers, several institutions now have had one or more positive heartworm tests in 3 otter species (NARO, ASCO, Spot-necked). None of these animals were symptomatic and all were on annual preventative (monthly ivermectin). The positives showed up during routine screening using heartworm ELISA antigen tests. She recommends that while positive tests warrant further work-ups these animals should NOT be treated for heartworm until it is confirmed 100% that they have heartworm.

PRE-SHIPMENT EXAMINATION RECOMMENDATIONS

All otters should receive a thorough pre-shipment physical examination as outlined above in the preventative health care section. 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 shipping and receiving institutions' veterinarians **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 the medical records on a floppy disc or send them via E-mail.

QUARANTINE

> *"…every animal is capable of carrying infectious diseases in a quiescent state (cub-clinical infection), which can readily be reactivated by the stress of moving and change in regime. Such an animal may not necessarily become ill itself, but may begin shedding infectious agents which pose a risk to other animals in the collection. Freedom from disease or latent disease must never be assumed."* (Lewis 1995)

Basic guidelines for quarantine have been established by the Veterinary Standards Committee of the AAZV which are designed to prevent the introduction of infectious disease into an animal collection. While the individual zoo has ultimate control over in-house quarantines it is recommended that all animals should undergo a 30-day quarantine stay at the receiving institution before being added to 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. The diet should be slowly adjusted over several weeks if there is to be a diet change.

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, then quarantine animals.

Balance between the necessity of keeping the quarantine pen clean and the needs of the animal can be tricky. There is some indication that many mustelids do *"…better isolated in enclosures as opposed to hospital-type quarantine pens"* (Lewis 1995). If this is not practical, or possible, a privacy box, climbing furniture, substrate suitable for rubbing/drying-off on, and a pool or water tub suitable for swimming should be provided. Whatever type holding facility is used, be sure it is otter-proof, they will climb, dig and chew.

There is much to be learned from a close physical examination (see below). In the case of otters, this requires putting the animal under a general anesthetic. The facility's policy on non-vital anesthesia should be followed, however, because otters are particularly adept at masking signs of illness, it is advised that a thorough physical exam be conducted before releasing the animal into the resident population.

Quarantine examinations

Two quarantine exams are recommended for otters; one performed at the beginning of the quarantine period (even if one has been conducted at the shipping institution) 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.

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

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

Final exam:
During the last week of quarantine, a thorough physical examination should be conducted as outlined in the preventative health care section. **It is extremely important to take radiographs of the animal during this time even if they were done at the previous institution.** 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.

CONTROL OF REPRODUCTION

Note: This information has been updated as of 2012 AZA Wildlife Contraception Center recommendations for contraception in otter species. Their website should always be checked for more current information.

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 or email Contraception@stlzoo.org (Sally Boutelle).

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.

Gonadotropin releasing hormone (GnRH) agonists [Suprelorin® implants, or Lupron Depot®]: GnRH agonists 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 should be 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. The depot vehicle (Lupron®) cannot be removed to shorten the duration of efficacy to time reversals. The implant, Suprelorin®, may be placed strategically to allow for removal though this technique has not been fully tested. Contact the WCC for more information and tips on placement to facilitate removal which may hasten reversal. The most widely used implant formulations are designed to be effective either 6 or 12 months, but those are to be considered 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. Suprelorin may also be used to mitigate aggression, however the suppression of testosterone does not always stop aggression if they are learned behaviors.

Progestins [Melengestrol acetate (MGA) implants, Depo-Provera® injections, Ovaban® pills] If progestins 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.

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.

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Anesthesia

ANESTHESIA ADMINISTRATION, MONITORING AND RECOVERY

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. A variety of agents have successfully been used in otter species for immobilization. These include Ketamine alone (not recommended), Ketamine with midazolam (preferred), 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. However, due to the stress experienced by the animal as a result of the amount of time required using this method it is considered less desirable. Training otters to receive voluntary hand injections of anesthesia agents is the preferred method. Despite the method of induction, anesthesia can be maintained by intubating the animal and maintaining it on Isofluorane (Ohmeda Pharmaceutical Products Division Inc., P.O. Box 804, 110 Allen Rd., Liberty Corner, NJ 07938) or Halothane (Fort Dodge, 9401 Indian Creek Parkway, Ste. 1500, Overland Park, KS 66210) anesthesia. Otters are relatively easy to intubate, and this method is preferred when it is necessary for an animal to be immobilized for procedures longer than 15 minutes.

Careful monitoring of anesthetic depth and vital signs is important in any immobilization. Body temperature, respiratory rate and depth, heart rate and rhythm, and mucous membrane color and refill time should be assessed frequently. Pulse oximetry sites include the tongue, the lip at the commissure of the mouth, or in the rectum; however, it can be difficult to find a suitable probe site for pulse oximetry. Spelman found the following methods successful: a Nellcor D-25 probe (Nellcor, Inc.) *"..folded over the tongue or digits and secured with a paperclip, or a Nellcor RS-10 reflectance probe modified as an esophageal or rectal probe (or any comparable probe), indirect blood pressure can be readily measured with a small or neonatal cuff on the base of the tail"* (Spelman 1999). Oxygen supplementation should be available and administered when indicated.

The AZA Otter SSP veterinary advisor (G. Myers) suggests the following to improve anesthetic events:

- Accurate weights to establish appropriate drug doses,
- Reduction of stress leading up to anesthetic event, (training, conditioning),
- Intubation of all otters on procedures lasting longer than 15 minutes,
- Oxygen supplementation, intermittent positive pressure ventilation,
- Monitoring equipment, (SpO2, EKG, TPR),
- Well trained support staff (keepers, technicians)

Intubating an Otter

Photos: G. Myers

River Otter Short-term Anesthesia (Max 25-30 mins.) - Table

Anesthetic combination (mg/kg)*	Comments
Ketamine (10), midazolam (0.25)	Highly recommended
Ketamine (2.5), medetomidine (0.025) (atipamezole 0.125)	May need higher dosages (Ket 3.5, Med 0.035) but respiratory depression more likely
Tiletamine-zolazepam (4) ** (flumazenil 0.08)	Recovery may be prolonged without flumazenil
Ketamine (10)	Expect muscle rigidity and variable duration
Ketamine (10), diazepam (0.5 – 1)	Prolonged recovery compared to ketamine, midazolam
Ketamine (5-10), xylazine (1-2) (yohimbine 0.125)	Variable effects from heavy sedation only to respiratory depression. Alternative dosages Ket (3-4), xyl (3-4)
Azaperone (0.1), fentanyl (0.1-0.2) (naloxone 0.04)	Not recommended

***Dosages given are based upon intramuscular administration.**
From: Spelman, Lucy DVM: Table 2 from EAZWV Proceedings May 1998, Recommended anesthetic dosages (including reversal agents, in brackets, where appropriate) for short term anesthesia (25-30 min) in North American river otters (*Lutra canadensis)*.

** Petrini uses 8mg/kg Tiletamine-zolazepam (Telazol®) in *L. canadensis*

Physiologic Measures and the Effect of Anesthetic-Related Complications - Table

Physiologic Measure	Baseline (Range)	Increased	Decreased
Heart rate(beats/minute)	152 (130 – 178)	>180 (tachycardia)	<100 (bradycardia)
Respiratory rate (breaths/minute)	31 (10 – 60)	>40 (tachypnea)	<8 (bradypnea)
Relative oxyhemoglobin saturation (%)	97 (92 – 100)	NA	<80% (hypoxemia)
Mean arterial blood pressure (mm Hg)	63 (31 – 77)	<50 (hypotension)	>100 (hypertension)
Rectal temperature (°C)	38.4° (38.1 – 38.7°)	>40.1° (hyperthermia)	<36.7° (hypothermia)

From: L. Spelman 1999

Representative Arterial Blood Gas Results from 6 River Otters Anesthetized with Different Protocols - Table

Anesthesia Combination	PO_2 (mm Hg)	PCO_2 (mm Hg)	pH	HCO_3- (mEq/L)	SPO_2 (%)	SaO_2 (%)
Ketamine-midazolam	81.6	56.6	7.29	27.4	93	94.2
Tiletamine-zolazepam	69.3	51.9	7.32	27.2	93	91.8
Medetomidine-ketamine	68.8	54.2	7.29	26.4	93	90.9
Ketamine-diazepam	60.3	54.1	7.38	30.0	93	88.7
Fentanyl-midazolam	34.4	90.7	7.11	29.2	<50	44.6
Fentanyl-midazolam-azaperone	29.4	76.2	7.24	32.7	<50	42.0

From: L. Spelman 1999

Supplemental oxygen should always be available for administration, if necessary. For procedures lasting 30 minutes or longer, animals should be maintained on Isofluorane.

Signs of Illness

◄ It has been frequently repeated that an otter that skips one meal is worthy of concern; an otter that skips two meals is definitely ill. (An exception to this rule is a female that has just given birth; she may skip meals just prior to, and just after parturition. If this persists a veterinarian should be consulted.)

◄ Another excellent indicator of the state of an otter's health is the condition of its coat. It should be smooth, soft and shiny looking when dry. When wet, the coat should form spikes which allow the moisture to bead and run off upon emerging from the water. If it stays matted down, the coat is becoming saturated which is an indication there is something wrong. (This could be a sign of illness, poor nutrition or, an environmental problem such as caustic substances in the water, i.e. chlorine or an ozone system set too high.)

◄ If an animal is spending an excessive amount of time rubbing and rolling in an attempt to dry its coat.

◄ A reluctance to go in the water can mean an animal's coat is becoming drenched. Not only does this cause the animal to feel the cold but, also is an indication of a potential health problem.

◄ Loose or excessively soft stool; a healthy otter, receiving an appropriate diet, should have formed, soft stools.

◄ All of the non-species specific signals also should be watched for, i.e. swelling, lethargy, unusual behaviors, abnormal fluid drainage, etc.

Common Injuries or Ailments

Vaginal Discharge and Urogenital Disorders

Captive otters are often noted to have a slightly bloody or even a reddish-brown vaginal discharge. The significance of this is as yet unknown. One author describes a slightly blood-tinged vaginal fluid as being normal during estrus in the river otter (Seager 1978); blood spotting had been reported by some institutions. Other researchers suggest that bloody mucopurulent vaginal discharge is indicative of genitourinary disease (Hoover 1984, Hoover 1985) which appeared to be common in wild-caught captive females (4 of 10 females; *Proteus mirabilis* was the most important pathogen found in all four). There have been reports of females with intermittent red to reddish-brown vaginal discharge that have appeared healthy, but other animals have had serious, even life-threatening urinary or uterine disease. Some otter caretakers believe that bloody vaginal discharge is seasonal and related to the estrous cycle. However, one captive otter with a persistent history of bloody vaginal discharge had an ovariohysterectomy performed, and although histopathology of the uterus supported mild endometrial disease, the bloody vaginal discharge returned after the surgery. This case suggests that the etiology of bloody vaginal discharge in this species is not simple and is probably multifactorial. Numerous organisms have been isolated from the vagina of otters with bloody discharge, including *Pseudomonas aeruginosa* and *Proteus mirabilis*. Since pyometras and other significant urinary and uterine infections do occur in otters, it is important to investigate any symptom that may indicate genitourinary disease. These symptoms may include a distended abdomen, increased water consumption, frequent urination, and genital rubbing, as well as vaginal discharge.

POOR COAT CONDITION

Hair coat problems are frequently reported in captive North American river otters. A healthy otter coat has guard hairs which form spikes when wet rather than becoming clumped or smooth. Water on the surface of a well waterproofed otter will form small droplets that do not penetrate the undercoat and are quickly removed with one good body shake. Indeed, the lighter colored undercoat should not be visible (Duplaix-Hall, 1975). If the coat is in poor condition the otter may refuse to go in the water because it is becoming waterlogged. One of the causes of a poor coat is poor water quality either due to excessive organic debris or to chemicals used in pool filtration systems. High levels of chlorine pose a threat to the water repellent ability of an otter's coat. Chlorine levels should be monitored closely and not be allowed to go above 0.5 ppm for long periods of time. (See Captive Management section for chlorine discussion.) Another potential cause of poor coat quality is lack of appropriate dry surfaces for grooming. Sufficient land area, bedding material, exhibit furniture, and a variety of substrates are very important for the otter to maintain a healthy coat. Over-grooming is another common cause of coat problems. Over-grooming is often associated with stress or overcrowding. It can either be self-induced, or caused from an overzealous exhibit-mate. Dietary imbalances and dermatological disease from fungi, bacteria, parasites, or allergies can also cause hair/coat problems. A methodical approach to investigating the cause of poor coat condition is necessary to identify and correct the problem.

Coat Photos

Poor coat quality (Photo, right: Gary Woodburn; Photo below: OKWS 2010)

Good coat quality (Photo: Wikipedia)

ALOPECIA

Seasonal coat changes and even a partial alopecia may occasionally occur in both males and females during the breeding season. The alopecia most commonly involves the tail and ventral abdomen and usually is bilaterally symmetrical. It resolves spontaneously at the end of the breeding season and is a normal hormonal phenomenon. In addition, pregnant females may occasionally pull hair from their abdominal region when parturition draws near. Because Alopecia is a commonly reported problem in river otters the AZA Otter SSP Veterinary Advisor (G. Myers) has evaluated records submitted to her regarding this issue.

Her findings to date are as follows:

- Most report a focal area of hair loss
- Reported in both sexes, but more often in females
- Most often involves the tail and/or the abdomen
- Most cases resolve with, or without intervention or treatment, if the resolve
- Agrees, it is possibly associated with breeding season and hormonal influences

Her diagnostic recommendations are:

- Skin scrapes; superficial and deep to detect parasites
- Fungal and bacterial cultures
- Biopsy if it does not resolve in a timely fashion, but these are often unrewarding
- Multiple treatments have been tried with varying success:
    - Antibiotics
    - Antihistamines
    - Environmental changes
    - Eliminating pain or neuropathy as causal agents

BITE WOUNDS

Bite wounds caused by exhibit mates can become infected and form abscesses. These should be surgically drained and the wound flushed with antibacterial agents such as dilute Nolvasan® or Betadine®. Systemic antibiotic therapy is often indicated. Bacteria commonly found infecting bite wounds include *Pasturella sp., Streptococcus sp*., and *Staphylococcus sp.*

FOOT PAD ABRASIONS

Abrasions, erosions, and ulcers of the feet are generally caused by the lack of a suitable substrate for the otter to adequately dry itself on, or continued pacing. If an exhibit consists primarily of concrete, or gunite, bedding of some type should be provided. When feet are chronically wet, the pads become soft and can be easily rubbed raw by rough surfaces.

LOOSE STOOL/DIARRHEA

Otter feces are normally softly formed. Loose stools or diarrhea can result from a variety of diseases, but often is simply the result of poor diet, overfeeding, abrupt dietary change, or consuming food that is spoiled or has become rancid. Many experienced otter rehabilitators report that diarrhea can be a problem when hand raising pups. Frequently this is the result of overfeeding or feeding an inappropriate diet. (See Hand Rearing and Nutrition sections for more information on these topics.) Clostridial enteritis is one of the most commonly reported diseases in both wild and captive North American river otters and can result in diarrhea, often with blood and mucous. See Bacterial Diseases for more details on *Clostridium*. Many other bacteria, viruses, and parasites can also cause loose stools or diarrhea.

PNEUMONIA

Pneumonia is relatively common in both captive and free-living otters (Madsen 1999, Chanin 1985, Duplaix-Hall 1975, Hoover 1984, Hoover 1985). Pneumonia and other respiratory disease can result from parasitic, bacterial, fungal, or viral disease. Often it is secondary to underlying problems such as stress, poor coat condition, extreme environmental conditions, or concurrent illnesses that have reduced the animal's immune capability. Treatment usually includes appropriate antimicrobial or antiparasitic agents, supportive care, and eliminating any underlying environmental or health problems. Symptoms of pneumonia include coughing, dyspnea, and rapid breathing. Otter pups are particularly susceptible to pneumonia and respiratory disease.

STRESS

Foster (1986) defines stress as, "*…a cumulative response, the result of an animal's interaction with its environment through receptors. This is an adaptive phenomenon. All responses are primarily directed at coping with environmental change, and behavioral repertoires may be dependent upon the stressful interaction of an animal with its environment.*"

Stressors can be somatic, psychological, behavioral or miscellaneous; all of which can lead to poor health. Signs of stress may include poor coat condition, lack of appetite, unusual behavior, and/or frequent screaming. Examples of stressors given by Foster (1986) are listed below:

- Somatic stressors include: strange sounds, sights, and odors; unexpected touches; changes in position, heat, cold, or pressure; abnormal stretching of muscles and tendons; or effects of chemicals or drugs.
- Behavioral stressors include: unfamiliar surroundings, overcrowding, territorial or hierarchical upsets; changes in biological rhythms; lack of social contact; lack of isolation; the lack of habitual or imprinted foods.

Many other sources of stress are also possible. The vigilant otter keeper should always be on the alert for potential sources of stress to animals in their care.

DENTAL DISEASE

Periodontal disease**,** fractured teeth, and apical abscesses are common problems in captive otters. Facial swelling is a common symptom of an abscessed tooth, but it is not uncommon for animals to have severe dental disease and show no clinical signs. Regular examinations can help identify problems before they become too severe. Prompt treatment of fractures and abscesses is extremely important. Endodontic procedures such as root canals and pulpotomies can be performed on diseased teeth following appropriate antibiotic therapy. Alternatively, diseased teeth can be extracted. Animals with periodontal disease should have their teeth regularly cleaned and polished. Antibiotic therapy initiated several days before a dental cleaning and extended for 1-2 weeks following a procedure can help minimize bacterial embolism. Providing bones, such as knucklebones or neck bones from sheep or other large animals **twice** weekly, along with regular cleaning and polishing will help reduce periodontal problems.

Common Causes of Death

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

Common Causes of Death in L. Canadensis
(Unpublished research, G. Myers, DVM – Otter SSP Veterinary Advisor)
2007

Cause of death	Causal factor
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	-

*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 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 vs. 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).

General Mustelid Disease

VIRAL DISEASE

See also Appendix A.

Canine Distemper

Canine distemper has been confirmed in multiple species of mustelids, including the Eurasian otter, *Lutra lutra* (Madsen 1999, Geisel 1979, Loupal In Press). Antibody titers have been noted in *L. canadensis* (Kimber 2000), and recently a suspected case of distemper occurred in *L. canadensis* in Canada. Distemper could not be confirmed but clinical signs and distemper serology were supportive of the diagnosis. The animal survived without treatment (Sandra Black 2000, personal communication). In general, however, otters may be relatively resistant to canine distemper virus, compared to other mustelid species such as weasels and ferrets. In North America there have been many well-described epizootics of canine distemper in wild foxes and raccoons in areas where otters are common and there has been no corresponding mortality in otters. The clinical presentation of distemper in mustelids is similar to that in dogs. In addition to mucopurulent oculonasal discharge, respiratory disease, diarrhea, hyperkeratosis of the footpads, and C.N.S. signs; ferrets and mink also frequently get a rash under the chin and in the inguinal area. In the black-footed ferret intense pruritis and cutaneous hyperemia is common. Vaccine-induced distemper may have a slightly different clinical presentation, but has never been reported in the otter despite the use of a variety of modified-live products in this species.

Mink Enteritis Virus, Feline Panleukopenia, Canine Parvovirus

These closely related viruses have been shown to affect mink and the skunk. The domestic ferret is not susceptible to these viruses under natural circumstances. The disease in mustelids is similar to that in felids and includes diarrhea, vomiting, fever, and leukopenia. Several studies have reported positive antibody titers in wild river otters (Hoover 1985, Kimber 2000) and there has been at least one suspected case of parvovirus in a small-clawed otter housed in a United States zoo. However, viral particles were not found on histopathology and viral cultures and serology were not performed. “Feline enteritis/panleukopenia” was diagnosed in 18 out of 88 post mortems from a variety of zoos that were reviewed by one author (Duplaix-Hall), although the method of diagnosis was not described in these cases.

Aleutian disease (plasmacytosis)

Typically a disease of farm-raised mink, but has been found in feral mink, the domestic ferret, and the striped skunk. Aleutian disease viral antibody has been found in skunks, fishers, and the American marten (*Martes americana*). In one study, a river otter was challenged with Aleutian virus, but did not become clinically ill nor did it develop an antibody titer (Kenyon 1978). However, a disease resembling Aleutian disease was recently described in a wild European Otter *(L. lutra)*, although no virus was actually isolated (Wells 1989). Aleutian Disease is an immune-mediated disease caused by a parvovirus of which there are several strains of varying pathogenicity. In mink, infection can range from unapparent to fatal. Generally, the course of the disease is slowly progressive over months to years. It is characterized by weight loss, hypergammaglobulinemia (greater than 20% of total serum protein), reproductive failure, and an immune-mediated glomerulonephritis. Some animals have hemorrhagic enteritis. Neonates may develop a fatal interstitial pneumonia. Increased numbers of plasmacytes are found in the liver, kidney and other organs, hence the name plasmacytosis. Several methods of detecting the disease ante mortem are used including the rapid iodine agglutination test (IAT) and the counter immuno-electrophoresis (CIEP) test.

Rabies

The Center for Disease Control has records of at least 24 cases of rabid otters in North American. (Serfass 1995). Affected animals may remain calm and asymptomatic .

Coronavirus

Coronavirus has been implicated as the cause of epizootic catarrhal enteritis in both mink and ferrets (Williams 2000). There have been no confirmed cases of coronavirus enteritis in otters. In 1995-1996 thirty-eight river otters were tested for feline and canine coronavirus antibody. All were negative. In 1975, feline infectious peritonitis (FIP), a disease caused by a feline coronavirus, was suspected to have caused the death of 2 small-clawed otters, but the case was never confirmed with serology or viral isolation. (Van de Grift 1976).

Influenza

The domestic ferret is susceptible to certain strains of human influenza and is used as a research animal to study the disease. Symptoms in ferrets include sneezing, conjunctivitis, unilateral otitis, fever, and sometimes photophobia. The disease usually lasts 7-14 days. Avian influenza A virus was responsible for an outbreak of contagious interstitial pneumonia in mink in Sweden in 1984. Although it is not known whether other species of mustelids are susceptible to influenza, it would seem prudent for animal caretakers exhibiting signs of influenza to wear masks and disposable gloves when caring for mustelids. Infected ferrets can also transmit influenza to humans.

Rotavirus

A disease described in domestic ferrets as "ferret kit disease" is caused by a rotavirus. The disease usually affects kits from two to six weeks old causing diarrhea and resulting in high mortality. Histopathologic lesions include villous atrophy and vacuolation of villar epithelial cells in the small intestine. Direct electron microscopy is used to identify the virus in the feces. Serological tests are unreliable. Since secondary bacterial invaders may increase mortality, it has been recommended that affected individuals be treated with oral gentomycin and parenteral ampicillin. A syndrome in farmed mink known as "**3-day disease**", "**Utah disease**", or "**Epizootic Catarrhal Gastroenteritis**" may also be caused by a rotavirus. The disease is characterized by a short course of diarrhea, anorexia, and lethargy. It is rarely fatal.

Transmissible Mink Encephalopathy (TME)

TME is caused by a scrapie-like virus that occasionally causes disease in adult mink. Experimentally, striped skunks are also susceptible. It has a long incubation period lasting five to 12 months. Both morbidity and mortality are high. The main clinical signs can be attributed to lesions in the cerebrum and include behavioral changes, weakness, ataxia, and sometimes paralysis. Reproductive failure including stillbirths (often with anasarca) and congenital defects is also a feature of the disease. Diagnosis is based on histopathologic findings in the cerebrum.

Feline Leukemia Virus

Healthy domestic ferrets have tested positive to FeLV by ELISA, but the significance of this is unknown. It is possible that the test is cross-reacting with another retrovirus or that false positive results are occurring. To date, no immunodeficiency virus has been identified in otters.

Adenovirus (Infectious Canine Hepatitis)

This disease has been reported in the striped skunk, and there has been one river otter that died of symptoms suggestive of canine adenovirus, but the diagnosis was not confirmed by viral isolation (Kimber 2000). Antibody testing of wild, unvaccinated river otters has been negative for canine adenovirus in several studies (Hoover 1985, Kimber 2000).

Feline Rhinotracheitis (Feline Herpesvirus-1) and Feline Calicivirus

Upper respiratory disease suggestive of feline rhinotracheitis or calicivirus has not been reported in North American river otters. Sixty-four wild river otters from New York were serologically tested for antibody to feline herpesvirus-1 and feline calicivirus during a translocation study conducted in 1995-1996. All 64 animals were negative for antibody to both viruses. (Kimber 2000).

Herpesvirus

Herpesvirus-like intranuclear inclusion bodies were found in the oral, esophageal and corneal epithelial cells of a dead adult male sea otter (*Enhydra lutris*) found in Alaska (Harris 1990). Herpesvirus infections have been reported in both wild and captive sea otters. (Reimer 1998). Antibody titers to canine herpesvirus-1 have been reported in wild-caught otters. (Kimber 2000).

Herpes Necrotizing Encephalitis

Herpes necrotizing encephalitis is caused by a herpes simplex virus and has been reported in skunks. The virus causes necrotizing meningoencephalitis with necrosis and hemorrhage in the liver and adrenal gland. Clinical signs include salivation, tremors, and head bobbing. Diagnosis can be made from serology. (Wallach 1983).

Pseudorabies (Mad Itch)

This has been reported in several species of mustelids. Signs are similar to those of other carnivores and may include intense pruritis, ataxia, vomiting, salivation, dyspnea and death. (Wallach 1983).

BACTERIAL DISEASE

See also Appendix A.

Bacterial Pneumonia

Pneumonia has been frequently reported in otters (Duplaix-Hall 1975, Madsen 1999, Hoover 1984, Hoover 1985). No one agent appears to be responsible, although bacterial cultures are not available for most of the cases reported. Signs of pneumonia may include nasal discharge, dyspnea, coughing, anorexia, and lethargy. Treatment involves appropriate antibiotic therapy. Viral infections, poor coat quality, and/or stress can predispose animals to bacterial pneumonia.

Pseudomonas pneumonia

Several serotypes of *Pseudomonas aeruginosa* cause hemorrhagic pneumonia in mink. The disease usually occurs in the autumn and can quickly spread through a ranch. Mortality rates vary from 0.1% to 50%. Animals die quickly, often without showing clinical signs. Occasionally dyspnea, a bloody nasal discharge, or convulsions are seen. The main post mortem lesion is hemorrhagic pneumonia with or without hemorrhagic pleural exudate. There is evidence that bacterial toxins may play a role in the pathogenesis of the disease. Concurrent infection with calicivirus or picornavirus, as well as poor air quality with high ammonia levels has been implicated as predisposing factors in the pathogenesis of the disease. This particular syndrome has not been reported in river otters.

Clostridial Infections

Botulism--Most species of mustelids are susceptible to type C toxin (and to a lesser extent types A, B, and E) produced by *Clostridium botulinum*. Usually animals are found dead but some may exhibit paralysis and dyspnea before dying. There are no postmortem lesions. Eating cooked or uncooked meat contaminated by *Clostridium botulinum* spores found in the soil causes the disease. The prevalence of Clostridial organisms in the soil varies greatly from one geographic area to another. Animals not on a commercially prepared diet may benefit from annual vaccination, however eliminating soil contamination of food can also prevent the disease.

Clostridium perfringens enteritis—River otters appear to be particularly susceptible to overgrowth of *Clostridium perfringens* during periods of stress or dietary conversion. In a recent translocation study, a number of wild river otters became ill shortly after capture (Kollias 1998). These otters died anytime from 6 to 72 hours after capture. Clinical symptoms ranged from sudden death in some animals to mucoid watery diarrhea with or without blood to lethargy, anorexia, and hypothermia. Diagnosis is based on histopathological findings, anaerobic culture and the detection of Type A *Clostridium perfringens* exotoxin. Animals treated early in the course of the disease may respond to oral metronidazole (Kollias 1998) or parenteral trimethoprim-sulfa and *Clostridium perfringens* antitoxin (Kollias 1999), along with supportive care.

Tuberculosis

Many mustelids are susceptible to bovine, avian and human strains of tuberculosis. The disease has been reported in the domestic ferret, mink, otters, and the European badger. It is usually acquired by eating contaminated food, however in the European badger, transmission can occur from mother to cub, by aerosol, or through bite wounds. Clinical signs may include weight loss, enlarged lymph nodes, chronic respiratory disease, and mastitis. Tuberculin skin testing is unreliable. Serological tests used to identify European badgers with *Mycobacterium bovis* have also been unreliable.

Anthrax

Anthrax has been reported in the European badger, the honey badger (*Mellivora capensis*), and mink. Clinical signs include sudden death with blood draining from body cavities. Postmortem findings include subcutaneous and subserosal edema, hepatomegaly, and splenomegaly.

Campylobacteriosis

Diarrhea caused by *Campylobacter jejuni* and *Campylobacter coli* has been reported in domestic ferrets and mink (Petrini 1992). Fever and leucocytosis often accompany infections. Abortion and other reproductive problems occur in both mink and ferrets when they are inoculated with *C. jejuni* during pregnancy. Clinical disease is most common in animals less than six months of age, and asymptomatic carriers are not uncommon especially with adults. Special techniques are required to culture the organism from the feces. Humans are also susceptible to infections with *C. jejuni*. Erythromycin, the drug of choice in humans, does not eliminate the carrier state in ferrets. Raw meat diets appear to predispose mink to *C. jejuni* infection.

Helicobacter mustelae

Helicobacter mustelae is a bacterium that often colonizes the stomach of ferrets and causes a chronic, persistent gastritis, and sometimes leads to gastric or duodenal ulceration, and/or gastric cancer (Fox 1998). The organism is quite common in ferrets but infected animals are often asymptomatic, especially when they are young. Symptoms of disease such as vomiting, dark tarry stools, chronic weight loss, and anemia may occur if gastric ulceration or cancer develops. The diagnosis is made by special culture techniques of gastric biopsy or necropsy samples. Recently a serological assay has been developed (Fox1998).

Proliferative Bowel Disease

This is a syndrome described in young, 4-6 month old domestic ferrets characterized by mucohemorrhagic diarrhea , weight loss, and partial prolapse of the rectum. The disease causes a profound thickening of the mucosa and muscular wall of the colon, which can be palpated per rectum. Pathological lesions are similar to those found in hamsters with "wet tail" and swine with proliferative ileitis, except lesions in the ferret are in the colon not the ileum. A Campylobacter-like organism closely related to *Desulfovibrio spp* causes the disease in ferrets. In swine and hamsters it is named *Lawsonia intracellularis* (Fox 1998). Diagnosis is based on the detection of the organism in histological sections of the colon stained with silver. Recently a PCR test has been developed as a diagnostic test.

E. coli mastitis

Mastitis caused by *E. coli* is a rapidly progressive disease that has been reported in the domestic ferret. Toxemia occurs, and mortality can be quite high. Early, aggressive therapy involving amoxicillin with clavulanate, chloramphenicol, or gentamicin, along with surgical excision of the affected mammae has been successful (Fox 1998). The disease has also been seen in mink (Fox 1998). Other bacteria may also be associated with mastitis in mustelids.

Purulent Pleuritis

Pleuritis is an inflammation of the pleura, often accompanied by an accumulation of pus in the pleural space around the lungs. Purulent pleuritis involving several different bacteria has been commonly reported in mustelids including the European badger, mink, and a North American otter. *Bacteroides melanigenicus* was isolated from the pleural fluid in one case (Griffith 1983). In mink, it is often seen in conjunction with dental disease and severe gingivitis.

Purulent Peritonitis

Peritonitis is an inflammation of the lining of the abdomen (peritoneum). It can be caused by a variety of different bacteria, and is often accompanied by purulent liquid in the abdominal space. Invasion of the peritoneal cavity with bacteria is most often the result of a penetrating foreign object, either through the abdominal wall or through the intestine. However, blood-born septicemias can also result in peritonitis. In one river otter, the peritonitis was due to a pure growth of *Klebsiella pneumonia* with no evidence of underlying disease (Petrini, unpublished).

Brucellosis

Brucellosis is an infectious disease caused by one of several species of *Brucella*. Clinical signs usually included abortion and, sometimes, orchitis and infection of the accessory sex glands in males. An unidentified *Brucella sp*. was recently isolated from the lymph node of a European otter (*Lutra lutra*) which had been killed in a road traffic accident in Scotland (Foster 1996). The significance of this finding is unknown.

Leptospirosis

Wayre considered leptospirosis an important disease in otters (Chanin 1985), and it was suspected to be the cause of death in several otters in a review of postmortem reports from a variety of zoos (Duplaix-Hall 1975). However, Fairley in 1972 found no histologic evidence of leptospirosis in the kidneys of 15 otters (Chanin 1985), and it is rarely mentioned in the literature. Leptospirosis is a contagious disease that causes anorexia, fever, vomiting, lethargy, anemia, hemoglobinuria, icterus, abortion, or death. There are many serotypes of the virus and humans are also susceptible to infection. Transmission occurs via contact with water, moist soil, or vegetation contaminated with infected urine, or by direct contact with the infected animal. Rats are common carriers.

Listeriosis (Circling disease)

Listeriosis is a disease caused by infection with the bacteria *Listeria monocytogenes*. In most species it causes neurological symptoms such as ataxia and circling, and thus must be distinguished from rabies. The disease can also result in abortion, perinatal mortality, and septicemia. The disease has been reported in the ferret, sable and striped skunk. Some animals are asymptomatic carriers of the disease and shed the bacteria in their feces.

Pasteurellosis

Pasteurellosis can be caused by either *Pasteurella multocida* or *Pasteurella pseudotuberculosis*. Although this disease is most commonly seen in rodents and lagomorphs, it has been reported in several mustelid species including mink, marten, badger, otters, and ferrets (Wallach 1983). Clinical signs can vary and may include depression, septicemia, ataxia, anorexia, diarrhea, dyspnea, or acute death.

Salmonellosis
Salmonellosis is caused by one or many species of Salmonella and characterized clinically by one or more of three major syndromes – septicemia, acute enteritis, chronic enteritis. Although it can be treated with antibiotics, it is often difficult to rid the animal of the organism. *Salmonella* spp. have been isolated from the feces of clinically normal otters and does not always cause disease.

Tularemia
Tularemia is a bacterial disease caused by *Francisella tularensis* that results in small granulomas or abscesses throughout the lungs, liver, mesenteric lymph nodes, and spleen. It can infect a variety of species including free-ranging mustelids and ranch mink, as well as humans. Sudden death or acute onset of anorexia is the most common clinical sign in mustelids. Animals are infected when they ingest carcasses of infected animals, particularly fish, rabbits or rodents.

Actinomycosis
Actinomycosis has been reported in the ferret and other mustelids. The infective agent, *Actinomyces sp*., causes a disease known as "lumpy jaw". The organism has a predilection for the cervical area and often results in the abscessation of the submandibular lymph nodes, although lymph nodes throughout the body can also be affected.

MYCOTIC DISEASES

Fungal diseases have not been frequently reported in mustelids. See also Appendix A.

Dermatomycosis
Dermatomycosis is more commonly referred to as "ringworm". The most common causative agents are members of the genera *Microsporum* and *Trichophyton.* Dermatomycosis has been reported in mink, domestic ferrets, and otters. Ringworm is contagious and potentially zoonotic. Although some cases are self-limiting, others require treatment with topical and/or oral anti-fungal agents.

Histoplasmosis
Histoplasmosis is a systemic fungal disease that results in variety of clinical signs including, lymphadenopathy, pneumonia, anorexia, weight loss, splenomegaly, and hepatomegaly. It is caused by the intracellular organism, *Histoplasma capsulatum*. Histoplasmosis has been reported in both domestic ferrets and striped skunks, and is most common in the central part of the United States where the organism can be found in the soil.

Cryptococcosis
Cryptococcosis is an infection caused by *Cryptococcus neoformans* and usually causes neurological signs as a result of meningoencephalitis. There have been several cases in the domestic ferret (Fox 1998).

Blastomycosis
Blastomycosis is a systemic fungal infection caused by the fungus *Blastomyces dermatidis*. It has been reported in the domestic ferret. Blastomycosis can affect both the lungs and skin causing pneumonia and/or cutaneous lesions. The disease occurs most commonly in the central and southeastern United States.

Coccidioidomycosis
Coccidioidomycosis is a fungal disease caused by *Coccidioides immitis* and is most common in the southwestern United States. The disease can cause respiratory disease as well as lymphadenopathy and occasionally osteomyelitis. Several cases of this disease have been reported in captive small-clawed otters from Arizona (Petrini 1992)

Mucormycosis

Mucormycosis is a fungal infection usually caused by *Absidia corymbifera (ramosa).* It generally occurs in conjunction with the ear mite, *Otodectes cyanotes* and causes otitis media and meningoencephalitis. This disease has been reported in farmed ferrets.

Adiaspiromycosis

Adiaspiromycosis is a disease that causes granulomatous lesions in the lungs, and sometimes involves regional lymph nodes. Mustelids appear to be particularly susceptible, and there are several reports of the disease in European otters (Simpson 2000). The disease is caused by the fungus *Emmonsia* crescens (formerly *Chrysosporium crescens).*

PARASITES

Numerous external and internal parasites have been identified in both wild and captive otters. Only a few will be covered here. (See Appendix A).

Microfilaria

Microfilaria are found frequently in the blood of wild North American river otters (Hoover 1984, Hoover 1985, Davis 1992, and Tocidlowski 1997). There are at least 2 species of microfilaria that have been identified in otters. *Dirofilaria lutrae* adults have been found in both the subcutaneous tissues (Hoover 1984 and Hoover 1985) and in the pulmonary arteries (Hoover 1985) of wild otters. A second *Dirofilaria* species has been identified from the pulmonary arteries as well (Hoover 1984.) In addition, there is one report of a North American river otter from Louisiana that had one male *Dirofilaria immitus* in its heart (Snyder 1989). *D. immitus* is the agent that causes heartworm disease in dogs, but in this case no pathology was attributed to the parasite. It is unclear whether *D. lutrae* or any of the other *Dirofilaria* sp. can cause clinical heartworm disease in otters, but since they can occupy the pulmonary arteries, it is possible. Many institutions housing otters outside in heartworm endemic areas keep their animals on heartworm preventative.

Respiratory Parasites

Lungworms are fairly common in wild otters. Several species affect mustelids including *Crenosoma spp.*, *Perostrongylus spp*., and *Filaroides spp*. Clinical signs vary from cachexia and anemia to coughing, dyspnea, depression, and nasal discharge. Diagnosis is made by finding the first stage larvae in the feces. New York has reported ivermectin-resistant strains of *Crenosoma sp*. in *L. canadensis* and recommends treatment with fenbendazole (Kollias 1999). Baermann and direct fecal smear examination techniques are helpful in diagnosing these parasites.

Capillaria aerophilia is another common nematode that causes respiratory disease in mustelids. The adult worms live in the trachea, bronchi, and bronchioles. Animals with mild infections may be asymptomatic but heavy infestations can result in coughing, respiratory difficulty, bronchopneumonia, nasal discharge and anorexia. *Capillaria* has been identified in the North American river otters (Hoover 1984, Hoover 1985, Kollias 1999). The ova, resembling a whipworm egg, are found in the feces or sputum.

Adult nematodes of the genus *Skrjabingylus* are located in the frontal sinus and cause progressive damage to the skull and nasal turbinates. Clinically, nasal discharge and neurological signs may be seen. *Skrjabingylus spp.* have been reported in mink, ermine, fishers, North American otters, striped skunks, and spotted-necked otters (*Lutra maculicollis*), as well as other mustelids. One study reported that as many as 13 % of wild North American river otters from Ontario were affected by the parasite (Addison 1988).

Lung flukes (*Paragonimus kellicotti*) have been reported in mustelids including mink, marten, badger, weasel, and skunk (Wallach 1983, Davis 1971). Infection results in a moist cough, sometimes with blood, elevated temperature, anemia, and difficulty breathing.

Kidney worm

Dioctophyma renale has been reported in a variety of mustelids including the North American otter. Mink appear to be the primary host in North America. The parasite causes weight loss, abdominal pain, and hematuria. Infection almost always involves the right kidney where a plate of bone often forms which is visible on radiographs. Diagnosis can be made from finding the ova in the urine. Another species of kidney worm, *Gnathostoma miyazakii*, is commonly found in *L. canadensis* in Virginia.

Acanthocephala (Thorny-headed worms)

Acanthocephalus spp. have been found in wild North American river otters (Kollars 1997, Hoberg 1997, Hoover 1984). One species has been identified as *Corynosoma strumosum* (Hoberg 1997). These relatively large worms can cause anemia and enteritis.

"Guinea worm"

Dracunculus insignis is a nematode that has been found in the subcutaneous tissues of several mustelid species including fishers, skunks, and mink. A separate species has been identified in the North American river otter. *Dracunculus* causes a varying degree of pruritis and local erythema. It is most commonly located on the legs. Removal of the adult parasite is curative (Petrini 1992).

Trichinosis

Trichinosis is a disease caused by the nematode *Trichinella spiralis*. The disease has been reported in the European otter and other mustelid species. Mustelids fed raw meat harboring viable trichina larvae are susceptible to the infection. Typical clinical signs include muscular pain, anorexia, and dyspnea. Heavy infestations are fatal.

Cestodes (Tapeworms)

Multiple species of cestodes have been identified in mustelids. Species that have been identified in North American river otters include *Ligula intestinalis* and *Diphyllobothrium* (questionable identification) (Chanin 1985), as well as *Schistocephalus solidus* (Hoberg 1997).

Miscellaneous nematodes

Several species of intestinal nematodes have been identified in North American river otters. One of the most commonly reported is *Strongyloides lutrae* (Kollias 1999, Hoover 1984, Hoover 1985, Hoberg 1997, Kollars 1997). Other species identified include *Ancylostoma sp* (Hoover 1985), and *Eustrongylides sp*. (Hoberg 1997). Some nematodes have been reported but not speciated. Many of the oocytes found in the feces of wild *L. canadensis* are considered incidental, and are actually fish or amphibian parasites whose eggs are being passed in the feces of the otter after consumption of the host.

MISCELLANEOUS TREMATODES (FLUKES)

Several kinds of intestinal flukes have been reported in mustelids. Species that have been reported in North American river otters include: *Euparyphium inerme* (Hoberg 1997), *Baschkirovitrema incrassatum* (Kollars 1997), *Enhydridiplostomum fosteri* (Hoover 1984), *Euparyphium melis* (Chanin 1985), and *Nanophyetus salmincola* (Schlegel 1968).

PROTOZOAN

See also Appendix A.

Coccidia

Heavy infestations of coccidia can cause mortality in young ferret and mink kits. Several species of *Isospora* have been reported in North American river otters (Hoover 1985), but clinical disease due to the parasite was not noted.

Toxoplasmosis

Toxoplasmosis can cause neonatal deaths and stunting in both ferrets and mink. A survey of North American river otters conducted in North Carolina showed that 45 % of the animals had positive titers to toxoplasma (Tocidlowski 1997).

Cryptosporidia

Asymptomatic ferrets can shed C*ryptosporidia* and pose a zoonotic hazard. To date there are no reports of this parasite in the North American river otter.

Giardia

Giardia spp. has been seen in the feces of various mustelids.

Sarcocystis

Sarcocystis spp. have been found in fishers, mink, wolverines, badgers, and recently in a European otter (*Lutra lutra*). In this case, the sarcocysts were found in large numbers in the skeletal muscle, but there was no apparent pathology associated with their presence (Wahlstrom 1999). Ferrets can be infected experimentally but remain asymptomatic (Fox 1998). Sarcocystis eggs can be passed in the feces and are easily mistaken for coccidia.

EXTERNAL PARASITES

External parasites are relatively uncommon in North American river otters (Serfass 1992) compared to other carnivores. However there are occasional reports of ticks, lice, and fleas. See Appendix A.

MISCELLANEOUS DISEASES

Urolithiasis

Urolithiasis has been reported in a variety of otter species. Calcium oxalate stones are common in captive *Aonyx cinerea* (Petrini, 1996). And in one study, 10 out of 14 *Lutra lutra* carcasses submitted for pathological examination were affected with urolithiasis. These uroliths contained calcium phosphate, calcium urate, or ammonium urate (Keymer, 1981). Other pathological reports of *Lutra lutra* have also detected kidney stones (Madsen 1999, Chanin 1985, Simpson 1997). One of these reports identified the calculi in three otters as being composed of ammonium urate (Madsen 1999). Small uroliths are often seen on radiographic examination of *L. canadensis* at the Minnesota Zoological Garden, however clinical disease is rarely noted. Of 21 animals radiographed over the past 20 years, 10 have had uroliths. Both calcium oxalate and magnesium phosphate calculi have been detected at this institution (Petrini, unpublished).

Neonatal mortality

Neonatal mortality in otters is most commonly due to septicemia, but starvation and hypothermia are also common in the first four weeks of life. Trauma and cannibalism can also occur. Neonatal losses can be minimized by providing secluded nest boxes (at least two, offering the dam a choice), sufficient dry bedding, and insuring adequate nutrition for the dam.

Hypocalcemia, Milk Fever, Pregnancy Toxemia

Low serum calcium levels in lactating or late gestation females can cause weakness, rear leg paralysis, convulsions, and death. The disease can affect ferrets approximately 3-4 weeks postpartum but sometimes occurs in the late pregnancy (Petrini 1992). Poor nutrition has been implicated as a cause, but this is not proven. Post mortem findings usually include hepatic lipidosis. Prompt treatment with intravenous calcium can quickly reverse the condition.

Agalactia, "Nursing Sickness"

Both mink and ferrets are afflicted with a syndrome known as agalactia or "nursing sickness". This generally occurs 5-6 weeks postpartum before the kits are completely weaned, but it can occur even after weaning. Clinical signs include lethargy, weight loss, ataxia, weakness, and coma. Occasionally hemolytic anemia can occur. The cause of the disease is unknown, but diets high in polyunsaturated fats may predispose animals to the condition. Some authors believe the condition is due to a sodium chloride deficiency. Hepatic lipidosis is often seen on post mortem examination. Offering food and water to kits beginning at 2-3 weeks of age will help prevent the disease (Petrini 1992).

Gastric ulcers

Gastric ulcers can cause vomiting, halitosis, melena, anemia, and acute death in mink and domestic ferrets. It has also been seen in weaning age otter pups (Joe Davis 1985, unpublished). Although stress has usually been implicated as the etiology, *Helicobacter mustelae* has been shown to cause gastritis and ulcers in domestic ferrets (see Bacterial Diseases for more information). Cimetidine, amoxicillin, metronidazole, and bismuth subsalicylate are suggested treatments (Fox 1998). Gastric ulcers can also occur secondary to renal failure.

Gastric dilatation (Bloat)

Acute gastric dilatation has also been reported in ferrets. It is often associated with dietary changes or overeating, especially after a prolonged fast. *Clostridium welchii* has sometimes been isolated from bloated ferrets. *Clostridium perfringens* enterotoxemia (see Bacterial Diseases for more details) may lead to gastric distention in North American river otters.

Diabetes mellitus

Diabetes mellitus has been described in the ferret and there has been at least one case in an Asian small-clawed otter and one case in a North American river otter (Petrini, unpublished).

Exertional Myopathy (Capture Myopathy)

Capture myopathy has been reported in translocated North American river otters (Hartup 1999). This disease is the result of excessive muscle exertion and stress and involves complex metabolic changes occurring 1-3 days after a stressful event. Animals typically show depression, anorexia, muscle weakness, ataxia, and pain.

Intervertebral disc disease

This is another common entity reported in ferrets, otters, and other mustelids. Exhibit space, housing, handling procedures and activity can predispose animals to vertebral problems and should be carefully evaluated.

Cancers

Numerous **neoplasias** have been reported in mustelids. Lymphosarcoma is the most commonly reported tumor type in the domestic ferret, followed by tumors of the reproductive tract and skin. In the mink, lymphoreticular tumors and anal sac carcinomas are commonly reported. Tumors resembling Hodgkin's Disease in humans have been reported from striped skunks.

Pulmonary Silicosis

Three North American river otters from one zoo died during or shortly after anesthetic procedures and were found to have pulmonary silicosis. Aluminum silicate was found in the lung tissue and also in the insulation material used in the nest box. It is likely that chronic inhalation of silica particles resulted in serious lung pathology and predisposed them to anesthetic complications (Suedmeyer 1999).

Dietary Issues

North American river otters have a high metabolic rate compared to other mammals and will eat approximately 9 % of their body weight per day (wet weight basis) (Davis 1992). One study reported that river otters consume about 177 kcal of gross energy per kg body weight per day (Davis 1992). They also have a very short digestive tract compared to other mammals. It takes only about 3 hours for food to pass through the entire tract once it is consumed (Davis 1992). A diet high in protein and fat and low in carbohydrates fed two or preferably, three times daily is best suited for this type of animal. Frequent feeding of small meals also helps stimulate physical activity and reduces the likelihood that food will spoil and later be consumed.

GENERAL NUTRITION

► **Protein** – Otters are for the most part dependent on animal protein sources.

► **Carbohydrates** – *"Carbohydrates are not as important as an energy source to the mustelids as are fats, and therefore low carbohydrate cat or mink diets are preferable to the high carbohydrate dog foods."* (Wallach 1983)

► **Fats** – *"Fats are an important energy source for the mustelids. Rancid fats cause severe digestive problems and interfere with utilization of several nutrients, including vitamin A, vitamin E-selenium, calcium, and zinc."* (Wallach 1983)

► **Vitamins and Minerals** – The omission of these important elements can lead to serious deficiencies. Although, most commercially prepared diets contain adequate amounts of these essential elements, **all dietary products should be researched and monitored for quality on a regular basis.)**

■ **Thiamine (B_1) –** Thiamine deficiency is also known as Chastek's paralysis. Clinical signs include, anorexia, salivation, ataxia, incoordination, papillary dilatation, sluggish reflexes, and paralysis . Feeding certain types of raw fish that are particularly high in the enzyme thiaminase is the usual cause. Thiaminase destroys the thiamine present in the food and therefore results in a deficiency of this nutrient. Herring, smelt, and carp are only a few of the fish that contain high levels of thiaminase. Otters that are being fed fish should be supplemented with thiamine at the rate of 25-30 mg thiamine per kg of fish fed. (See the Diet/Nutrition Section for more details.)

■ **Vitamin E** – a deficiency of Vitamin E can cause yellow-fat disease, also known as steatitis. Clinical signs may include lethargy, lumpy subcutaneous fat, rear leg weakness, and death. Vitamin E deficiency can also cause fatty degeneration of the liver, hemolytic anemia, and anorexia. Young growing animals are particularly sensitive. Diets that are high in long-chain polyunsaturated fatty acids (common in fish oils), or rancid fats can cause vitamin E deficiency Otters that are being fed fish should be supplemented with vitamin E at the rate of 100 I.U. of vitamin E per kg of fish fed.

■ **Vitamin A deficiency** – clinical signs include, poor growth, dry and dull hair coat, infertility and birth defects in young.

"…Mustelids do not convert b-carotene to vitamin A efficiently, (so), it should not be used as the sole source of vitamin A" (Petrini 1992).

■ **Biotin** – Biotin deficiency can be caused by feeding raw eggs; this is due to the biotinase content of albumin. Biotin deficiency results in pale fur and skin problems.

■ **Vitamin D** – a deficiency of Vitamin D will produce Rickets. Conversely, over supplementation of Vitamin D causes abnormal calcification of soft tissues.

LACTATING FEMALES

"Energy demands during lactation are quite high in mustelids" (Petrini 1992). Although it is not known specifically for *L. canadensis, "…in the American badger (Taxidea taxus), lactation demands 16 times more energy than gestation. This is approximately four times more than required by most mammals"* (Petrini 1992). A gradual increase in diet during lactation to a level 30 to 40% over maintenance level is usually required for successful growth of the pups and maternal health.

Post Mortem Examinations

The value of a thorough postmortem examination for disease surveillance of both wild and captive populations cannot be overemphasized. Animals should be necropsied as soon as possible after death. Bacterial overgrowth begins shortly after death making it difficult to isolate pathogens that may have been involved in the animal's demise. Carcasses should be refrigerated until the examination can be completed. This will retard bacterial growth and help slow down the autolysis of tissues. If it is not possible to complete the necropsy within about 5 days, the carcass should be frozen. This however, greatly changes the microscopic architecture of the tissues, rendering them useless for histological examination. Therefore, a great deal of valuable information will be lost if the carcass is frozen.

The ideal necropsy will include a complete gross examination of the carcass and internal organs, a parasitological exam, and histological examination of the individual tissues. In addition, cultures for pathogens such as bacteria, fungus, and viruses are often indicated.

Below is an example of a standardized necropsy protocol for otters as well as a necropsy report form.

NECROPSY PROTOCOL

Blood Collection

Antemortem blood collection for serum banking is recommended on any animal that is to be euthanized. Collect enough to obtain a minimum of 5 ml of serum. Post mortem blood collection may be possible on specimens that have recently died.

Radiographs

Ventral-dorsal and lateral abdominal radiograph should be taken on all otters that die. Urolithiasis (kidney and bladder stones) is relatively common in several species of otter. Radiographs can help document the degree (or lack thereof) of urolithiasis at the time of death. Although this is not believed to be as prevalent in *Lontra canadensis* radiographs should still be done.

Gross Port Mortem Examination

A veterinarian should perform a thorough post mortem examination as soon as possible after death. The standardized necropsy report included at the end of this section can be used for recording the results.

Handling Pathological Lesions

- Cultures: Cultures (aerobic, anaerobic, and fungal) should be taken of any lesions before they are contaminated.
- Freeze tissues: Samples of lesions should be frozen at -20^{o} or -70^{o} C.
- Histopathology: Make sure all lesions are saved for histopathology.

Formalin Fixed Tissues

Tissues should be collected and placed in 10% buffered formalin. All tissues may be placed together in a single container as long as the volume of formalin is at least 10 times the total volume of the tissues collected. Tissues should be no thicker than 0.5 cm. A checklist of tissues that should be preserved in formalin is attached. In addition, include sections of all lesions in formalin.

Histopathology

Fixed tissues should be sent to a pathologist, preferably one that is familiar with exotic species.

Frozen Tissues

3-5 cm sections of the following tissues should be frozen in plastic bags at –20 to –70 ^{o}C.

- Liver
- Brain
- Kidney
- Serum, (antemortem or postmortem) if possible.
- Sections of any lesions.

Neonates, Stillbirths, Abortions

In addition to the standard adult necropsy protocol, include the following:

- Weight, crown-rump length, and sex.
- Estimate degree of maturity (1^{st}, 2^{nd}, or 3^{rd} trimester).
- Fix umbilical stump and surrounding tissues. Obtain bacterial cultures before fixing if there is evidence of infection.
- Check carefully for evidence of congenital deformities (cleft palate, deformed limbs, heart defects, anal atresia, etc).
- Assess hydration (tissue moistness) and evidence of nursing (milk in stomach).
- Determine if breathing occurred (do lungs float in formalin).
- Note whether there is meconium in the colon/rectum.
- Fix placenta if available. Culture first if indicated.

Tissues to be placed in 10 % formalin.

□ Skin: full thickness of abdominal skin.
□ Skeletal muscle: medial thigh, with sciatic nerve.
□ Tongue: Cross section near tip including both mucosal surfaces.
□ Trachea
□ Thyroid/parathyroid
□ Thymus: representative section.
□ Lungs: section from several lobes including a major bronchus.
□ Heart: Longitudinal section including atrium, ventricle and valves form both right and left heart. Include large vessels.
□ Aorta
□ Salivary gland
□ Gastrointestinal tract: 2-3 cm long section of esophagus, stomach (cardia, antrum, pylorus), duodenum, jejunum, ileum, colon, omentum. Open carefully along the long axis.
□ Lymph nodes: Cervical, bronchial, and mesenteric with a transverse cut.
□ Liver: Sections from several lobes with capsule and gall bladder.
□ Adrenal: Incise transversely.
□ Reproductive tract: Entire uterus and ovaries with longitudinal cut into lumen. Entire testis with transverse cut, entire prostate with transverse cut.
□ Pancreas: Representative sections from 2 areas.
□ Spleen: Cross sections including capsule.
□ Kidneys: Section from both kidneys (cortex, medulla, and pelvis).
□ Urinary bladder/ureter/urethra: Cross section of bladder, 2-cm sections of ureters, cross section of urethra.
□ Eyes: leave intact.
□ Brain, include cerebrum and cerebellum: Sliced longitudinally along the midline.

- □ Pituitary gland: Submit entire gland including dura.
- □ Long bone: Submit ½ of femur.
- □ Mammary gland
- □ Diaphragm
- □ Spinal cord: 1-cm section from cervical cord.

OTTER NECROPSY REPORT

Institution where otter was housed or location where found: ______________________________________

Common name:________________________ Genus / Species:___________________________________

Identification or Accession #: ______________ Necropsy #: ___________ Studbook #:__________

Date of Birth: ______________ Age: _______ Weight:_______ Kg Sex:___________

Length, tip of nose to base of tail:______________ Length, tip of nose to tip of tail: ___________

Date of Death: ________________ Date of Necropsy: _____________________________

Gross exam performed by: ____

Histopathology performed by: ____

Pathology Accession # ____________ Disposition of carcass: ________________________

Tissue saved: Yes 9 No 9

Formalin 9

Frozen 9

Other 9

____

HISTORY (Include clinical signs, treatments, antemortem test results, diet, circumstances of death and quarantine status.

Laboratory Studies: (List bacterial and viral cultures submitted. Attach results of any of the following:)

Hematology	☐	Chemistry	☐	Photography	☐
Cytology	☐	Fluid analysis	☐	Bacteriology	☐
Mycology	☐	Virology	☐	Urinalysis	☐
Parasitology	☐	Toxicology	☐	Urolith analysis	☐
Other	☐				

Radiology:

Urinary calculi Yes 9 No 9

Left kidney: Number _____ Size _____
Right kidney: Number _____ Size _____
Urinary bladder: Number _____ Size _____

Gross Diagnosis: (List each lesion separately. Include organ, lesion type, distribution, severity, etc.)

Histological Diagnosis: See attached report 9 Not done 9

Final Diagnosis:

Summary / Comments:

GROSS EXAMINATION
General Condition: (External appearance, condition of carcass, physical and nutritional condition, pelage, subcutaneous fat stores, body orifices, superficial lymph nodes.)
Musculoskeletal System: (Bone, joints, muscles)
Body Cavities: (Fat stores, abnormal fluids)
Hemolymphatic: (Spleen, lymph nodes, thymus,)
Respiratory System: (Nasal cavity, larynx, trachea, bronchi, lungs, regional lymph nodes)
Cardiovascular System: (Heart, pericardium, great vessels)

Digestive System: (Mouth, teeth, esophagus, stomach, intestines, liver and gall bladder, pancreas, mesenteric lymph nodes)
Urinary System: (Kidneys, ureters, urinary bladder, urethra)
Reproductive System: (Testis, ovary, uterus, oviduct, vagina, penis, prepuce, accessory sex glands, mammary glands, placenta)
Endocrine System: (Adrenals, thyroid, parathyroids, pituitary)
Nervous and Sensory Systems: (Brain, spinal cord, peripheral nerves, eyes, ears)

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Appendix A Ectoparasites, Endoparasites, Viruses, Bacteria, Fungi, and Protozoans known to inhabit river otter (Melquist et al. 2003)

Ectoparasites found in river otter (Melquist et al. 2003)				
Order	**Family**	**Species**	**Geographic location**	**Source**
Acarina	Ixodidae	*Ixodes cookie*	Alabama, Arkansas, Florida, Pennsylvania	Cooney & Hays 1972; Forrester 1992; Polechla 1996
		Ixodes uriae	California	Eley 1977
		Ixodes banksi	Florida, Michigan, Pennsylvania	Forrester 1992
		Amblyoma americanum	Arkansas, Florida	Polechla 1996; Eley 1977
		Dermacentor variabilis	Florida	Eley 1977
	Listrophoridaea	*Lutracarus canadensis*	Alaska	Fain & Yunker 1980
		Lynxacarus mustelae	Alaska	Fain & Yunker 1980
Anoplura	Echinophthiriidae	*Latagopthirus rauschi*	Oregon	Kim & Emerson 1974
Siphonaptera	Certophyllidae	*Oropsylla arctomys*	Pennsylvania	Serfass et al. 1992
Coleoptera	Leptinidae	*Leptinillus validus*	Minnesota	Route & Peterson 1988
		Platypsyllus castoris	Minnesota	Route & Peterson 1988

Helminth Endoparasites Reported in River Otter
(Melquist et al. 2003 citing Kimber & Kollias 2000)

Phylum	Species	Geographic Location	Body Location Parasitized
Pentastomida	*Sebekia mississippiensis*	Florida	GI (intestine)
Platyhelminthe			
Cestoidea	*Spirometra mansonoides*	Florida, Georgia	GI (intestine), subcutaneous
	Diphyllobothrium latum	North Carolina	Subcutaneous
	Ligula intestinalis	Montana	GI (feces)
	Protoecephalus perplexus	Alabama	GI (stomach)
	Schistoecephalus solidus	Oregon, Newfoundland	GI (intestine)
	Braunia sp.	New York	GI (feces)
Trematoda	*Euparyphium melis*	Massachusetts, Michigan, Minnesota	GI (stomach, small intestine)
	Euparyphium inerme	Oregon, Washington	GI (stomach, small intestine)
	Enhydrodiplostomum alaroides	Alabama, Florida, Georgia, Massachusetts, North Carolina	GI (stomach, small intestine)
	Enhydrodiplostomum fosteri	Alabama, Louisiana	GI (small intestine)
	Enhydrodiplostomum sp.	North Carolina	GI (intestine)
	Bashkirovitrema incrassatum	Alabama, Florida, Georgia, Louisiana, Massachusetts, North Carolina, New York, Tennessee, Ontario	GI (stomach, small intestine)
	Nanophyetus salmincola	Pacific Northwest	Unknown
	Paragonius kellicoti	Michigan	Unknown
	Alaria canis	Ontario	Subcutaneous mesenteric fat
	Crepidostomum cooperi	Alabama	Unknown
	Telorchis gracilis	Alabama	GI (small intestine)
	Telorchis spp.	Alabama	Unknown
	Diplostomium alarioides	Ontario	GI (small intestine)
Acanthocephala	*Pilum sp.*	Unknown	Unknown
	Oncicola spp.	Florida	GI (intestine)
	Acanthocephalus spp.	Alabama, Tennessee	GI (large intestine)
	Pomporhynchus spp.	Alabama	GI (large intestine)
	Corynosoma strumosum	Oregon	GI (stomach, intestine)
	Leptorhynchoides spp.	Alabama	Unknown
	Neoechinorhynchus spp.	Alabama	Unknown
	Paracanthocephalus rauschi	Alaska	GI (intestine)
Nematoda	*Physaloptera spp.*	Alabama, Massachusetts	GI (stomach, intestine)
	Skrjabingylus lutrae	Ontario	Frontal sinuses
	Dracunculus lutrae	Arkansas, Nebraska, New York,	Subcutaneous tissues, leg

Helminth Endoparasites Reported in River Otter
(Melquist et al. 2003 citing Kimber & Kollias 2000)

Phylum	Species	Geographic Location	Body Location Parasitized
	Dracunculus lutrae	Ontario	Subcutaneous tissues, leg
Nematoda	*Dracunculus insignis*	Alabama, Florida, Michigan, New York	Facial layers of leg
	Strongyloides lutrae	Alabama, Florida, Louisiana, Oregon, Tennessee, Washington	GI (small intestine), lung
	Crenosoma goblei	Florida, North Carolina	GI (intestine), lung
	Dirofilaria lutrae	Florida, Louisiana, Southeastern USA	Blood, subcutaneous tissue
	Dirofilaria immitis	Louisiana	Heart
	Capillaria plica	Florida, North Carolina	Urinary bladder
	Capillaria aerophilus	Unknown	Lungs
	Capillaria hepatica	Florida	Liver
	Capillaria sp.	Louisiana	Unknown
	Gnathostoma miyazakii	Alabama, Florida, North Carolina, Virginia, Ontario	GI (intestine), kidney
	Dioctophyme renale	Unknown	Kidney
	Filaroides canadensis	Ontario	Lungs
	Contracaecum spp.	Oregon	GI (stomach, large intestine)
	Spinitectus gracilis	Alabama, Oregon	GI (small intestine)
	Spinitectus sp.	Oregon	GI (intestine)
	Eustrongylides spp.	Maryland, Oregon	GI (small and large intestine)
	Anisakis simplex	Oregon, Washington	GI (small and large intestine, stomach)
	Anisakis spp.	Oregon	GI (intestine)
	Metabronema sp.	Oregon	GI (intestine)
	Hedruris siredonis	Oregon	GI (small and large intestine)
	Hedruris spp.	Oregon	GI (intestine)
	Cystidicoloides	Washington	GI (intestine)
	Ancylostoma spp.	Louisiana	GI (intestine)
	Uncinaria stenocephala	Unknown	GI (intestine)
	Strongyle sp.	Minnesota	GI (feces)
	Cruzia sp.	Oregon	GI (small and large intestine)

Viruses, Bacteria, Fungi, and Protozoans Reported From River Otters
(Melquist et al. 2003 citing original sources)

Disease	Causative Agent	Note	Source
VIRAL			
Canine Distemper	Canine distemper virus	Antibody titers, clinical signs, and distemper serology	Kimber et al. 2000
Mink enteritis virus, feline panleukopenia, canine parvovirus		Positive antibody titers	Hoover et al. 1985; Kimber et al. 2000
Aleutian disease (plasmacytosis)	Aleutian virus	Challenged, but not clinically ill, no antibody titer	Kenyon et al. 1978
Rabies	Rabies virus	24 cases reported	Serfass, 1995
Infectious canine hepatitis	Adenovirus	Symptoms but no viral isolation confirmation	Kimber et al. 2000
Feline rhinotracheitis	Feline herpesvirus-1, feline calicivirus	64 animals tested negative for antibody	Kimber et al. 2000
Herpesvirus	Herpesvirus-1	Antibody titers reported	Kimber et al. 2000
BACTERIUM			
Bacterial pneumonia	Unknown	Frequently reported	Hoover 1984; Hoover et al. 1985
Clostridial infection	*Clostridium botulinum*	Susceptible to type C toxin	Reed-Smith 2001
Enteritis	*Clostridium perfringens*	Susceptible during periods of stress or dietary conversion	Kollias 1999
Tuberculosis	*Mycobacterium bovis*	Reported	Reed-Smith 2001
Purulent pleuritis	*Bacteroides melanigenicus* and others	*B. melanigenicus* isolated in one case	Griffith et al. 1983
Purulent peritonitis	*Klebsiella* pneumonia	Presence of bacteria but no underlying disease	Reed-Smith 2001
Brucellosis	*Brucella spp.*	Presence in lymph node of a road-killed *Lutra lutra*	Foster-Turley 1996
Leptospirosis	? bacteria	Believed to be an important disease but no histopathology	Chanin 1985; Reed-Smith 2001
Pasteurellosis	*Pastueurella multocida* or *P. pseudotuberculosis*	Clinical signs vary	Wallach & Boever 1983; Reed-Smith 2001
Salmonellosis	*Salmonella spp.*	Isolated from feces of clinically normal otters, no symptoms	Reed-Smith 2001
FUNGAL			
Dermatomycosis	*Microsporum spp.* and *Trichophyton spp.*	Contagious and potentially zoonotic	Reed-Smith 2001
Coccidiodomycosis	*Coccidioides immitis*	Reported from small-clawed otters	Reed-Smith 2001
Adiaspiromycosis	*Emmonsia crescens*	Reported from Eurasian otters	Simpson & Gavier-Widen 2000

Viruses, Bacteria, Fungi, and Protozoans Reported From River Otters
(Melquist et al. 2003 citing original sources)

Disease	Causative Agent	Note	Source
PROTOZOAN			
Giardiasis	*Isospora spp.*	Light infestation	Hoover et al. 1985
Coccidiosis	*Giardia spp.*	Reported from other mustelids	Reed-Smith 2001

Health Care Glossary

Anasarca: Severe generalized edema. (Taber's Cyclopedic Medical Dictionary)

Anemia: A quantitative deficiency of the hemoglobin, often accompanied by a reduced number of red blood cells causing pallor, weakness, and breathlessness. A lack of vigor, creativity, forcefulness, or the like. (Webster's Unabridged Dictionary)

Apnea: Cessation of breathing.

Ataxia: Defective muscular coordination often manifested when voluntary muscular movements are attempted.

Autolysis: The self-dissolution or self-digestion that occurs in tissues or cells by enzymes in the cells themselves, such as occurs after death and in some pathological conditions. 2) Hemolysis of blood cells occurring as a result of the action of an animal's own serum or plasma. (Taber's Cyclopedic Medical Dictionary)

Bacteria: Any of numerous microscopic, spherical, rod-shaped, or spiral organisms of the class Schizomycetes, various species of which are concerned in fermentation and putrefaction, the production of disease, the fixing of atmospheric nitrogen, etc. (Webster's Unabridged Dictionary)

Bacterin: A vaccine that contains specific bacteria and is injected to increase immunity. (Webster's Unabridged Dictionary)

Cachexia: General ill health, with emaciation, due to a chronic disease, as cancer. (Webster's Unabridged Dictionary)

Catarrh: Term formerly applied to inflammation of mucous membranes, especially of head and throat. (Taber's Cyclopedic Medical Dictionary)

Catarrhal: Of the nature of or pertaining to catarrh.

Conjunctivitis: Inflammation of the mucous membrane that lines the inner surface of the eyelids. (Webster's Unabridged Dictionary)

Cutaneous: Pertaining to the skin. (Taber's Cyclopedic Medical Dictionary)

DHLPP: Canine vaccine. Sometimes called DA_2PL (A=adenovirus)

D = Canine Distemper
H = Hepatitis
L = Leptospira Bacteria
P = Parainfluenza-Adenovirus Type 2
P = Parvovirus vaccine

Dyspnea: Air hunger resulting in labored or difficult breathing, sometimes accompanied by pain. (Taber's Cyclopedic Medical Dictionary).

Enteritis: Inflammation of the intestines, especially the small intestines. (Webster's Unabridged Dictionary)

Erythema: Abnormal redness of the skin due to local congestion, as in inflammation. (Webster's Unabridged Dictionary)

FVRCP: Feline vaccine.

F = Feline distemper
V = Viral
R = Rhinotracheitis
C = Calici Virus
P = Panleukopenia

Hematuria: The presence of blood in the urine. (Webster's Unabridged Dictionary)

Hemorrhagic: Pertaining to or marked by hemorrhage.

Hyperemia: Congestion. An unusual amount of blood in a part. (Taber's Cyclopedic Medical Dictionary)

Hypergammaglobulinemia: Excess amount of gamma globulin in the blood. (Taber's Cyclopedic Medical Dictionary)

Hyperkeratosis: Overgrowth of the cornea. 2) Overgrowth of the horny layer of the epidermis. (Taber's Cyclopedic Medical Dictionary)

Hypostasis: Diminished blood flow or circulation. 2) Deposit of sediment due to decreased flow of body fluid such as blood or urine. (Taber's Cyclopedic Medical Dictionary)

Lacrimation: Tearing.

Leucocytes: Any of the small, colorless cells in the blood, lymph, and tissues, which move like amoebae and destroy organisms that cause disease; white blood corpuscle. (Webster's Unabridged Dictionary)

"Leukocytes, or white blood cells (WBC), in mammalian blood include segmented neutrophils, band (nonsegmented) neutrophils, lymphocytes, monocytes, eosinophils, and basophils. These cells vary in their site of production, duration of peripheral circulation, recirculation, and in the stimuli that affect their release into and migration out of the vascular bed. These factors also vary among species.

***"Leukocytosis** is an increase in the total number of circulating WBC: leukopenia is a decrease."* (Merck 1991)

Leukopenia: A decrease in the number of leucocytes in the blood. (Webster's Unabridged Dictionary)

Mucopurulent: Consisting of mucous and pus. (Taber's Cyclopedic Medical Dictionary)

Mycosis: The growth of parasitic fungi in any part of the body. A disease caused by such fungi.

Mycotic: Pertaining to or caused by mycosis. (Webster's Unabridged Dictionary)

Nematode: Any unsegmented worm of the phylum or class nematoda, having an elongated, cylindrical body; roundworm. (Webster's Unabridged Dictionary)

Orchiditis: Inflammation of the testicles.

Otitis: Inflamed condition of the ear. (Taber's Cyclopedic Medical Dictionary)

Parasite: An animal or plant that lives on or in an organism of another species from whose body it obtains nutriment. (Webster's Unabridged Dictionary)

Plasmacyte: A plasma cell, one of those found in connective tissue, with an eccentrically placed round nucleus and filled with a chromatin mass that stains deeply. (Taber's Cyclopedic Medical Dictionary)

Pruritis: A severe itching. May be a symptom of a disease process such as allergic response, or be due to emotional factors. (Taber's Cyclopedic Medical Dictionary)

Purulent: Full of, containing, forming, or discharging pus; of the nature of or like pus. (Webster's Unabridged Dictionary)

Rhinitis: Inflammation of the nose or its mucous membrane. (Webster's Unabridged Dictionary)

Septicemia: The invasion and persistence of pathogenic bacteria in the blood stream. (Webster's Unabridged Dictionary)

Serology: The scientific study of serum. (Taber's Cyclopedic Medical Dictionary)

Seroconversion: Development of evidence of antibody response to a disease or vaccine.

Silicosis: A form of pneumoconiosis resulting from inhalation of silica (quartz) dust, characterized by formation of small discrete nodules. In advanced cases, a dense fibrosis and emphysema with impairment of respiratory function may develop.

Tachypnea: Rapid breathing.

Titer: Standard of strength per volume of a volumetric test solution. (Taber's Cyclopedic Medical Dictionary)

Vaccine: Any preparation of dead bacteria introduced in the body to produce immunity to a specific disease by causing the formation of antibodies. (Webster's Unabridged Dictionary)

Virus: An infectious agent, especially any of a group of ultramicroscopic, infectious agents that reproduce only in living cells. (Webster's Unabridged Dictionary)