"To the dolphin alone, nature has given that which the best philosophers seek: friendship for no advantage. Though it has no need of help from any man, it is a genial friend to all and has helped mankind."

–Plutarch

To my sons *Lauris* and *Raphaël*, who I hope will become true philosophers,
and to *Kai*, the dolphin friend who has inspired this project

Aquatic Mammals 2005, *31*(3), 279-280, DOI 10.1578/AM.31.3.2005.279

Foreword

One freezing day during the winter of 1990 on the Pacific coast of Japan, I was kneeling at the edge of a dolphin pool, playing with my favourite dolphin, Kai, a shy and slender female Pacific bottlenose dolphin. I was pleased to see how she seemed to enjoy our moments together. She always displayed her pleasure to see me with leaps, splashes, and loud vocalisations, pushing her way to me, displacing other animals despite her low rank in the hierarchy of the pool. We had great moments together during our daily evening tacit appointment. Unfortunately, I could not see her under water because the pool was not equipped with windows. Upon arrival near the pool, I almost fell in the cold water because I had just slipped on ice accumulated on the corners of the square pool. To be able to cuddle and play with her, I was bent in two, kneeling on the pool side, with puddles of water seeping into my plastic boots. Like every day, my back was sore, my knees were bruised, every part of my body hurt. I felt miserable in such an uncomfortable position. My body was strained from the efforts of carrying hundreds of kg of fish every day, four buckets at a time, through narrow staircases, arms full of frozen packs between the truck and the freezer, and between the freezer and the kitchen. Thirty-three dolphins; three belugas; four killer whales; and innumerable walruses, seals, sea lions, and sea otters had to be fed every day by the small pinniped and cetacean teams, mainly composed of young light-framed Japanese women! To this very day, I remember trying for the first time to picture in my mind the ideal pool for Kai, along with a more comfortable ledge on which to handle her, a fish kitchen at the same level as the stage and closer to the pools, and every pool accessible by trolleys. From that day on, I started putting in writing some of my ideas about the design and husbandry problems which had been making the animals' lives unnecessarily stressful and my life, as an apprentice dolphin trainer, needlessly tiring. I drafted a whole list of items that could be modified and improved—from the shape of the pool, the design of the kitchen, to husbandry protocols. At the end of my stay at Kamogawa Sea World, I had several notebooks filled with sketches, plans, photos, and notes scribbled in French, English, and Japanese.

I had a degree in architectural design and a passion for cetaceans. Thanks to a scholarship from the Japanese Ministry of Education, I could reconcile both interests in Japan by studying marine architecture at the Nihon University in Tokyo. I had toured many of the numerous dolphinaria in Japan and had been accepted for a seven-month training at Kamogawa Sea World, which at that time was the largest and most famous institution in Japan. It is located on the Pacific coast of the Chiba Peninsula that forms Tokyo Bay, in a small remote tourist resort that is 2 hours from Tokyo. At Sea World, I was completely immersed into the life of the dolphinarium, sharing the same accommodations as my young Japanese colleagues—their meals, their parties, emergency situations, joys and sorrows with the animals, and 16-h days. The time spent there still remains for me an incredible experience in my life, as well as an extraordinarily rich professional training.

It was later, after I left Japan and stayed in France for several years, that the opportunity was offered to me by Dr. Elizabeth Taylor, head of the Dolphin Study Group of the Tropical Marine Science Institute (TMSI) at the National University of Singapore (NUS), to do a full-time research project on cetacean environments in human care. I could finally combine my education in architectural design and my short experience in dolphin training and husbandry to try to enhance the environment of captive dolphins and whales, as well as the working conditions of their trainers. I dug up my notebooks and left for a new part of Asia, a place so dear to my heart.

I was first employed by the Faculty of Architecture of NUS and then by TMSI when the institute became an official entity within the university. The research grant allowed me to travel in Asia, Australia, and the USA. During visits to numerous dolphinaria, I saw the same mistakes repeated over and over again, mainly by architects for whom the expression of their art conflicted with the remarks and suggestions of trainers, curators, veterinarians, and research scientists, or by uninventive replication of the same designs. Sometimes these mistakes were made by managers too eager to please the public to the detriment of the animals. Fortunately, there also were visits when the environments included some ingenious husbandry features that would greatly facilitate medical training; a stunning new design concept for pools, a great support facility layout, or a reliable husbandry protocol to promote preventive medicine.

A survey of international institutions displaying cetaceans was part of the research project. The report presenting the results of this project was entitled *Dolphins and Whales Captive Environment Project*. One hundred and fifty copies were distributed or sold internationally, mainly to facilities displaying cetaceans, to trainers, and to some architects and planners.

After the completion of the project, I left Singapore and returned to France, my home country, where I joined the French company, Grévin & Cie, owner of attraction parks, of two dolphinaria in France and the Netherlands, and of several aquaria and animal parks. In the department of Grévin & Cie which creates and renovates attractions and animal exhibits, as well as new visual identity and pedagogical projects, I was hired as Animal Project Coordinator. It gave me an incredible chance to apply some of the fruits of my research, get deeper into various aspects like the life support system, and explore new domains such as pinniped exhibits and "edutainment."

This is how I became a more active member of the European Association for Aquatic Mammals (EAAM). I had been familiar with the association and a reader of the journal, *Aquatic Mammals*, since my research days in Singapore. In 2004, the editorial board of the EAAM approved the idea of republishing an updated version of the guidebook as a special issue of *Aquatic Mammals*. Grévin & Cie would sponsor the publication of the special issue as a contribution to the improvement of cetacean housing. I updated every chapter, which were then separately peer-reviewed and corrected by specialists from Europe, the USA, and South Africa. The present issue is the result of this collaborative work.

Happy and thriving animals require more than clever environmental design, clean water, and a healthy diet. They also require careful attention, frequent and regular training; a well-balanced community of poolmates; dedicated trainers; and environmental enrichment through play, foraging, and socialisation. Apart from a brief overview on husbandry, I did not cover these aspects because I felt that better qualified people have done it in other books and articles. I tried in this work to mainly cover the physical and technical aspects of the cetacean environment. Guidelines and recommendations are presented in a simple and straightforward way so that every professional, from trainers and architects to curators and life support system specialists who were involved in the planning, design, maintenance, renovation, or management of a dolphinarium, could understand the elements involved. I seek the forgiveness of the people more experienced than me in domains such as water chemistry and veterinary medicine who might find the related chapters superficial. I tried to make them as comprehensive as possible.

Finally, I would like to make clear that this book is not about the pros and cons of captivity. It is not about judging bad facilities either. It is about how to contribute to make the environment of cetaceans in human care as suitable and as good as possible, so the animals can thrive, live long and healthy lives, and so that the work of their caretakers can be as comfortable and as safe as possible. Thriving animals make the visitor's experience more enjoyable and the resulting awareness and education in regards to cetacean conservation more durable and effective.

Throughout this endeavour, I had Kai and her poolmates always in mind.

Laurence Couquiaud, Senlis, August 2005

If the reader has questions or comments, please send them to the author at lcouquiaud@yahoo.com.

Aquatic Mammals 2005, *31*(3), 281, DOI 10.1578/AM.31.3.2005.281

Acknowledgments

I express my gratitude to the late Dr. Tobayama and the staff of Kamogawa Sea World for their support and friendship. Without them, I would not have known Kai and her companions who have inspired this research and this book.

I am deeply grateful to Dr. Elizabeth Taylor, Deputy Director of the Tropical Marine Science Institute, who initiated the interdisciplinary research project on cetacean environment in human care at the National University of Singapore and who enabled the resulting guidebook. With her support and guidance, I was able to fulfil a project very dear to me. I am also indebted to Pr. Lam Khee Poh, former Dean of the Faculty of Architecture, Building and Real Estate of the National University of Singapore, now Director of Graduate Programmes at the Faculty of Architecture of Carnegie Mellon University, who was the principal investigator of the initial research project, for his generous help, trust, and patience. National University of Singapore provided the financial support for the research project (RP 950050). I am also grateful to the Lee Foundation for the generous donation supporting the Education Fund at the Tropical Marine Science Institute; and to Dr. Peter Kew, who allowed the transfer of copyright from the NUS to the *Aquatic Mammals* journal.

I thank Ms. Karen Pryor for her support of the project and her kind encouragements, Mr. Etienne Douaze for his assistance with the research proposal and the statistical analysis of the survey, Ms. Grace Tan for her help in the data collection, and Ms. Bethel Borgeson for proofreading the first manuscript. I am grateful to the many friends and colleagues of TMSI for their encouragements and kind support, in particular to Ms. Gercende Courtois de Viçose and Ms. Tay Hui Cheng.

I thank all the institutions' staff members who took considerable time and effort to participate in the international survey of cetacean facilities, and to the patient and welcoming staff of the many dolphinaria I visited over the years. I appreciate the information kindly provided by Mr. Chris Thurtell and Mr. Guy Bedford. I extend my sincere thanks to Mrs. Birgitta Mercera from Parc Astérix, Ms. Ida Smit from Dolfinarium Harderwijk, Mr. Jean-Marie Chollet from Grevin & Cie, Mr. Oz Goffman, and the numerous individuals who generously contributed photographs for the book.

I am very grateful to the reviewers of this updated edition for their time and useful comments: Pr. Paul Nachtigall, Marine Mammal Research Project, University of Hawaii; Mr. Jan Mosterd, curator, Dolfinarium Harderwijk, the Netherlands; Mr. Niels van Elk, DVM, Dolfinarium Harderwijk and EAAM President-Elect; Dr. John Koop, Dolfinarium Harderwijk; Ms. Claudia Gili, DVM, Aquario di Genova; Ms. Gabrielle Harris, curator of uShaka Marine World, South Africa; Mr. Benoit Lamort, France; and Mr. Perrti Keskitalo, Tritonet, Finland. Special thanks go to Dr. Manuel Garcia-Hartman for his support of this re-edition project and to the EAAM editorial board.

I am thankful to Mr. Alexandre Gourevitch, communication director of Grévin & Cie, and Mr. Michel Linet-Frion, director of Grévin Productions, for their support inside the company; and the collaboration of the staff of our two dolphinaria: Parc Astérix and Dolfinarium Harderwijk.

The person without whom this publication would not have been possible is Dr. Jeanette Thomas, from Western Illinois University, Editor of *Aquatic Mammals*, who has encouraged me, guided me, supported the project with determination, and spent a considerable amount of time correcting and editing, grateful thanks to her.

Many thanks to Mrs. Laura Caldwell, Mrs. Sandy Larimer, and the Document and Publication staff for their thorough and patient editing work and their help with coordinating this special issue.

Finally, I express my deepest gratitude and love to my family: to my parents, whose endless devotion helped me more than anything to complete the first book through hard times, as well as Mr. and Mrs. Douaze; to my husband, Alain, for his affectionate support, understanding, patience, and faithful love; and to our sons, Lauris and Raphaël. (Raphaël was born at the same time as this issue was published.)

Aquatic Mammals 2005, *31*(3), 283-287, DOI 10.1578/AM.31.3.2005.283

1. Introduction

A Bit of History

Since antiquity, dolphins and whales have been featured in legends, historical writings, and artistic works from the Mediterranean Basin to the far eastern countries of Asia. The streamlined, graceful body of dolphins and their playfulness, and the majestic or frightful sight of the largest whales have intrigued human imagination for centuries. Aristotle, four centuries BC, gave us the first description of physiological and behavioural characteristics of the dolphin (Collet & Duguy, 1987; Cousteau & Diolé, 1994; Thompson, 1989); however, it was only during the Renaissance in 16th century Europe that scientific observations of dolphins and whales were published and thereby reached a wider public. This was followed in the 18th and 19th centuries by reports from naturalists on various new cetacean species.

Dolphins were displayed in royal menageries in Europe as early as the 15th century. In 1400, the Duc of Burgundy displayed dolphins in the ponds of his palace in Dijon, and shortly after, Charles VI of France offered dolphins to Isabeau de Bavière for the Hôtel St. Paul in Paris (Collet, 1984). Near the end of the 18th century (circa 1790), a collection of Amazon River dolphins (*Inia geoffrensis*) was kept by Ferreira in Portugal (Schreib et al., 1994). During the 19th century, intensive whaling activities yielded physiological information on the gentle giants, but much remained unknown about their behaviour, distribution, and abundance.

In 1861, the first live beluga whale (*Delphinapterus leucas*), collected from the St. Lawrence River, was displayed at the Aquarial and Zoological Gardens in Boston with great public success (Hiatt & Tillis, 1997). Later that year, P., T. Barnum also displayed a beluga whale in his American Museum in New York City. This animal was exhibited for two years, along with a bottlenose dolphin (*Tursiops truncatus*). At about the same time, a harbour porpoise (*Phocoena phocoena*) was kept for several months in the Brighton Aquarium in England (Defran & Pryor, 1980). In the late 1870s, beluga whales from Labrador were shipped across the Atlantic Ocean and displayed at the Royal Aquarium in Westminster and at shows in Manchester and Blackpool (Defran & Pryor, 1980; Hiatt & Tillis, 1997; Joseph et al., 1990; Manton, 1989). Around 1878, a solitary specimen of the Ganges River dolphin, the Susu (*Platanista gangetica*), was briefly displayed in Calcutta. Barnum's beluga whale was probably the first cetacean to be trained in captivity, learning hand feeding, harness wearing, and object towing. The first birth recorded in captivity was a stillborn harbour porpoise calf at Brighton Aquarium in 1916 (Defran & Pryor, 1980). At the time, collection, transportation, and husbandry methods were rudimentary, and the whales did not live long in captivity. In 1938, a group of dolphins was established in the first oceanarium, Marine Studios (now Marineland) in Florida, and in 1947, the first live cetacean birth from a female bottlenose dolphin that was pregnant at the time of her collection was recorded there (Defran & Pryor, 1980). The Marine Studios originally was created for filmmakers who wanted a reliable supply of marine creatures before the invention of underwater filming equipment and SCUBA gear. Eventually, Marine Studios became involved in the display and training of captive cetaceans for public viewing, and the staff pioneered modern techniques of cetacean collection, transport, husbandry, and medicine (Defran & Pryor, 1980; Norris, 1974).

The creation of oceanaria allowed the first observations by McBride in 1940 of dolphins' social behaviours, as well as their mating, parturition, and mother-calf behaviours. Very little was known at that time about the behaviour and physiology of dolphins and whales. Their sound production and echolocation abilities were only suspected, but remained untested and not truly understood. Husbandry and medical care were learned empirically over the years by trainers and veterinarians through attempts at curing illnesses. As husbandry, feeding, and water quality requirements for keeping dolphins and whales became better understood, standards were refined. Surprisingly, the first attempts to display cetaceans in a captive environment were in an artificial setting rather than in a fenced-off area in the sea, which would have been easier and less expensive to manage. The display of animals in a theatre setting allowed the oceanarium to accommodate large crowds and present shows. Until very recently, this remained the only type of display, with small additional features for husbandry and training purposes; it is still the dominant presentation type for shows around the world.

In 1969, Hediger, Director of the Zurich Zoological Garden and pioneer in the study of the psychology of zoo animals, demonstrated that abnormal behaviours could be the result of several factors, such as the lack of environmental stimulation and inadequate space. He stated that in "architecture it is not the simplest and the cheapest type of building which should be given primary consideration, but the type of building which comes nearest to meeting the (animals') biological requirements. The cube, indeed a straight line of any kind, is unbiological" (Hediger, 1968; Doherty & Gibbons, 1994; Mench & Kreger, 1996). Markowitz (1982), in his book *Behavioural Enrichment in the Zoo*, introduced the concept of environmental or behavioural enrichment and its potential use in zoos. aquaria and dolphinaria, following the trend of many zoos, are placing increasing emphasis on environmental enrichment. Marine mammal exhibits are expensive to build and to operate; therefore, facilities make an important commitment when they acquire or breed large aquatic animals (Boness, 1996).

At the beginning of the 1990s, the increasing interest in environmental enrichment concepts in animal displays, the surge of public interest in dolphins and whales, and the heated controversy about captivity led to a new "naturalistic" design trend. The purpose was to display the animals in a more natural environment, which resulted in larger, more free-shaped pools, with irregular bottom topography; rock work; and in some cases, wave generators to simulate natural water movement. The development of educational programmes and the creation of numerous Swim-With-The-Dolphins (SWTD) facilities have accentuated this trend.

Many scientific studies have been conducted in captivity (Ellis, 1995), but few have focused on the effects of captivity to identify design elements or features likely to affect the animals' behaviour and health. A study on the effect of pool dimensions on the behaviour of two bottlenose dolphins has been conducted in one facility (Bassos & Wells, 1996), showing a correlation between certain types of pools and dimensions with certain behaviours. It would be interesting to repeat this study in other facilities housing cetaceans and extend it to other parameters and features. Future research with captive cetaceans could focus more on behaviors, such as resistance to gating in bottlenose dolphins (Myers & Overstrom, 1978), increased aggressiveness in confined environments (Overstrom, 1983), and avoidance of certain architectural features (Howard, 1995), in order to improve facilities design (Östman, 1991; Stoskopf & Gibbons, 1994; Ellis, 1995; Samuels & Gifford, 1997). The influence of the composition of dolphin groups on social interactions and on breeding, as well as the quality of the aquatic environment, are among the elements that are important to carefully establish and monitor to avoid stress, breeding failures, and health problems (Asper, 1982; Cornell et al., 1987; Markowitz, 1990; Reeves et al., 1994; Sweeney & Samansky, 1995).

Duffield & Wells (1990) made interesting comparisons between several wild and captive populations of Atlantic bottlenose dolphins in terms of their average age, age distribution, and population dynamics. Survival rates of several cetacean and pinniped species in captivity have been studied (DeMaster & Drevenak, 1988; Small & DeMaster, 1995a, 1995b). The improvement in the quality of the environment and husbandry methods for cetaceans during the past 20 years has significantly increased their survival in human care, making it comparable with the wild in western countries. Substantial differences remain among countries and parts of the world.

The creation of standards for the keeping of marine mammals in human care in the United States (Anonymous, 1979-1984, 1995; Young & Shapiro, 2001; Corkeron, 2002) was a first step towards a guarantee of minimum comfort for the animals. Other countries, such as Australia, France, Sweden, the United Kingdom, several other European countries, and the European Union, have established rules ensuring minimum requirements for housing marine mammals and regulating their capture, trade, and transportation (Klinowska & Brown, 1986; Anonymous, 1990, 1992, 1995, 2005b). One of the roles of professional associations, such as the European Association for Aquatic Mammals (EAAM), the International Marine Animal Trainers Association (IMATA), and the Alliance of Marine Mammal Parks and Aquariums (AMMPA) is to help spread good practices and environmental improvements (Anonymous, 2005a, 2005b, 2005c; see also Chapter 8 for details). Many countries lack similar legislation that regulates the capture, trade, or housing of marine mammals. With the help of these associations and the creation of governmental regulations, display facilities and research institutions in those countries hopefully will become aware of the importance of providing a suitable environment for marine mammals and a high quality of husbandry and medical care for the welfare of the animals, the protection of wild populations, the education of visitors, and the success of the exhibits. Hopefully, this work will provide an additional tool toward this goal.

The Project

The present work is the outcome of a three-year joint research project between the Tropical Marine Science Institute (TMSI) and the Faculty of Architecture, conducted at the National University of Singapore (NUS) and funded by an NUS research grant. It was part of the work done at the new Dolphin Study Group within the TMSI, which conducted research on cetacean local populations, behaviour, acoustics, cognition, and environment in human care.

The objectives of this research project were to (1) identify the various aspects and important features of the captive environment and (2) assist the professionals involved in the design of facilities, and especially those not directly from the field such as architects, by pointing-out recurrent mistakes and sharing good design ideas and suggestions for improvements of pool design, husbandry features, life support systems, and support facilities. The ultimate goal was to enhance the cetacean welfare in human care, the work comfort of their caretakers, and enrich the visitors' experience for educational benefits. The outcome of this research project was *The Dolphins and Whales Captive Environment Guidebook*, which was published at the end of 1999 as the research project report for the TMSI and the NUS.

The Methods

To propose appropriate suggestions, good architectural design and husbandry features, as well as recurrent deficiencies, had to be identified. The various types of habitat in which animals are kept also had to be categorised, and the influence of the location, climate, and age of the facility, among other elements, had to be determined. The opinion and advice from many professionals from the field had to be collected.

An extensive worldwide survey of existing cetacean facilities was prepared. A computerised database of cetacean facilities worldwide was compiled, and data from 166 dolphin facilities in 42 countries were recorded at that time. A comprehensive questionnaire on all aspects of the maintenance of cetaceans was designed to solicit information from institutional directors, curators, veterinarians, trainers, and scientists with experience in the care of cetaceans, such as the following:

- Institutions displaying cetaceans around the world
- Animal species, habitat, location, climate, behaviour
- Various types of environment and pool functions
- Architectural design of pools and enclosures, materials and construction, husbandry features, indoor and outdoor facilities, natural and artificial environments, support facilities
- Water quality and life support system
- Diet, food storage, preparation
- Husbandry and population management

The questionnaire was sent to 157 facilities, and 44 responded from 22 countries. Results of this international survey are presented in Chapter 3.

Existing regulations and guidelines from various countries for the housing of cetaceans were collected and analysed. The project team visited 26 facilities in Asia, North America, Europe, and the Middle East, supplementing additional data and enabling the compilation of valuable suggestions from many professionals. The original guidebook was compiled from all the valuable information extracted from the survey, from many articles from specialised journals, from personal experience, and from interviews with professionals during many visits to institutions and international symposia.

At a time when so many new facilities are being created in the Asia-Pacific region, the Middle East, and Central and South America, and when the general public is growing more concerned with animal welfare, I wanted this work to become a useful tool to improve this process. The aim also was to bring the various professionals involved to a better understanding of each other's language and requirements, to achieve a high-quality environment for the comfort and welfare of the animals and their caretakers, and to ensure the enjoyment and education of the public.

Literature Cited

Anonymous. (1979-1984, 1995). *Specifications for the humane handling, care, treatment and transportation of marine mammals* (9 CFR, Subpart E, §3.100-118). Washington, DC: U.S. Department of Agriculture, the Animal and Plant Health Inspection Service.

Anonymous. (1990). *Supplement to the Secretary of State standards of modern zoo practice: Additional standards for UK cetacean keeping.* United Kingdom Secretary of State.

Anonymous. (1992). *Guidelines for the care, holding and transport of marine mammals in Queensland* (Draft). Queensland, Australia: Queensland Department of Primary Industries. 39 pp.

Anonymous. (2003). *Alliance of Marine Mammal Parks and Aquariums standards and guidelines.* Alexandria, VA: AMMPA. Available online: www.ammpa.org. Accessed: 6 July 2005.

Anonymous. (2005a). European Association for Aquatic Mammals (EAAM) website. Available online: www.eaam.org. Accessed: 6 July 2005.

Anonymous. (2005b). European Community and Wildlife Trade website. Available online: http://europe.eu.int/comm/environment/cites/home_en.htm. Accessed: 19 July 2005.

Anonymous. (2005c). International Marine Animal Trainers Association (IMATA) website. Available online: www.imata.org. Accessed: July 6, 2005.

Asper, E. D. (1982). Design features of marine mammal exhibits. In K. Sausman (Ed.), *Zoological parks and aquarium fundamentals* (pp. 99-104). Wheeling, WV: American Association of Zoological Parks and Aquariums.

Bassos, M. K., & Wells, R. (1996). Effect of pool features on the behavior of two bottlenose dolphins. *Marine Mammal Science*, *12*(2), 321-324.

Boness, D. J. (1996). Water quality management in aquatic mammal exhibits. In D. G. Kleiman, M. E. Allen, K. V. Thompson, S. Lumpkin, & H. Harris (Eds.), *Wild mammals in captivity: Principles and techniques* (pp. 231-242). Chicago: The University of Chicago Press.

Collet, A. (1984). Live-capture of cetaceans for European institutions. *Reports of the International Whaling Commission*, *34*, 603-607.

Collet, A., & Duguy, R. (1987). *Les dauphins, historique et biologie.* Le Rocher, Paris: Science et Découvertes.

Corkeron, P. (2002). Captivity. In W. F. Perrin, B. Würsig, & J. G. M. Thewissen (Eds.), *Encyclopedia of marine mammals* (pp. 192-197). London: Academic Press.

Cornell, L. H., Asper, E. D., Antrim, J. E., Searles, S. S., Young, W. G., & Goff, T. (1987). Progress report: Results of a long-range captive breeding program for the bottlenose dolphin, *Tursiops truncatus* and *Tursiops truncatus gilli*. *Zoo Biology*, *6*, 41-53.

Cousteau, J., & Diole, P. (1994). *Les dauphins.* Paris: Arthaud. 239 pp.

Defran, R. H., & Pryor, K. (1980). The behavior and training of cetaceans in captivity. In L. M. Herman (Ed.), *Cetacean behavior: Mechanisms and functions* (pp. 319-358). New York: John Wiley & Sons. 463 pp.

DeMaster, D. P., & Drevenak, J. K. (1988). Survivorship patterns in three species of captive cetaceans. *Marine Mammal Science*, *4*(4), 297-311.

Doherty, J. G., & Gibbons, E. F. (1994). Managing naturalistic animal environments in captivity. In E. F. Gibbons, E. J. Wyers, E. Waters, & E. W. Menzel (Eds.), *Naturalistic environments in captivity for animal behavior research* (pp. 126-139). Albany: State University of New York Press.

Duffield, D. A., & Shell, D. (1994). The past, present and future of marine mammal management. *Proceedings of the 22th Annual IMATA Conference*, Tacoma, Washington.

Duffield, D. A., & Wells, R. (1990). Bottlenose dolphins: Comparison of census data from dolphins in captivity with a wild population. *Proceedings of the 18th Annual IMATA Conference*, Chicago, Illinois.

Ellis, S. (1995). Marine mammal behavior: Conservation through research. In E. F. Gibbons, Jr., B. S. Durrant, & J. Demarest (Eds.), *Conservation of endangered species in captivity: An interdisciplinary approach* (pp. 441-463). Albany: State University of New York Press.

Hediger, H. (1968). *The psychology and behaviour of animals in zoos and circuses.* New York: Dover Publications.

Hiatt, M., & Tillis, K. (1997). The beluga whale (*Delphinapterus leucas*). *Soundings*, *22*(1), 22-31.

Howard, C. J. (1995). *Dolphin chronicles.* New York: Bantam Books. 304 pp.

Joseph, B. E., Asper, E. D., & Antrim, J. E. (1990). Marine mammal transport. In L. Dierauf (Ed.), *CRC handbook of marine mammal medicine: Health, disease, and rehabilitation* (pp. 543-551). Boston: CRC Press.

Klinowska, M., & Brown, S. (1986). *A review of dolphinaria*. London: Department of the Environment. 247 pp.

Manton, V. J. A. (1989). Les cétacés en captivité. In R. J. Harrison & M. M. Bryden (Eds.), *Baleines, dauphins et marsouins* (pp. 196-203). Paris: Bordas.

Markowitz, H. (1982). *Behavioural enrichment in the zoo*. New York: Van Nostrand Reinhold.

Markowitz, H. (1990). Environmental opportunities and health care for marine mammals. In L. Dierauf (Ed.), *CRC handbook of marine mammal medicine: Health, disease, and rehabilitation* (pp. 483-488). Boston: CRC Press.

Mench, J. A., & Kreger, M. D. (1996). Ethical and welfare issues associated with keeping wild mammals in captivity. In D. G. Kleiman, M. E. Allen, K. V. Thompson, S. Lumpkin, & H. Harris (Eds.), *Wild mammals in captivity: Principles and techniques* (pp. 5-15). Chicago: The University of Chicago Press.

Myers, W. A., & Overstrom, N. (1978). The role of daily observation in the husbandry of captive dolphins (*Tursiops truncatus*). *Cetology*, *29*.

Norris, K. S. (1974). *The porpoise watcher.* New York: Norton & Co. 250 pp.

Östman, J. (1991). Changes in aggressive and sexual behavior between two male bottlenose dolphins (*Tursiops truncatus*) in a captive colony. In K. J. Pryor & K. S. Norris (Eds.), *Dolphin societies* (pp. 305-316). Berkeley: University of California Press.

Overstrom, N. A. (1983). Association between burst-pulse sounds and aggressive behavior in captive Atlantic bottlenosed dolphins (*Tursiops truncatus*). *Zoo Biology*, *2*, 93-103.

Reeves, R. R., DeMaster, D. P., Hill, C. L., & Leatherwood, S. (1994). Survivorship of odontocete cetaceans at Ocean Park, Hong Kong, 1974-1994. *Asian Marine Biology*, *11*, 107-124.

Samuels, A., & Gifford, T. (1997). A quantitative assessment of dominance relations among bottlenose dolphins. *Marine Mammal Science*, *13*(1), 70-99.

Schreib, S., Burrows, A., & Smith, T. (1994). The Amazon river dolphin. *Soundings*, *19*(2), 5-29.

Small, R. J., & DeMaster, D. P. (1995a). Survival of five species of captive marine mammals. *Marine Mammal Science*, *11*(2), 209-226.

Small, R. J., & DeMaster, D. P. (1995b). Acclimation to captivity: A quantitative estimate based on survival of bottlenose dolphins and California sea lions. *Marine Mammal Science*, *11*(4), 510-519.

Stoskopf, M. K., & Gibbons, E. F. (1994). Quantitative evaluation of the effects of environmental parameters on the physiology, behavior, and health of animals in naturalistic captive environments. In E. F. Gibbons, E. J. Wyers, E. Waters, & E. W. Menzel (Eds.), *Naturalistic environments in captivity for animal behavior research* (pp. 140-160). Albany: State University of New York Press.

Sweeney, J. C., & Samansky, T. (1995). Elements of successful facility design: Marine mammals. In E. F. Gibbons, Jr., B. S. Durrant, & J. Demarest (Eds.), *Conservation of endangered species in captivity: An interdisciplinary approach* (pp. 465-477). Albany: State University of New York Press.

Thompson, R. (1989). Les cétacés dans l'art et dans la littérature. In R. J. Harrison & M. M. Bryden (Eds.), *Baleines, dauphins et marsouins* (pp. 168-181). Paris: Bordas.

Young, N. M., & Shapiro, S. L. (2001). U.S. federal legislation governing marine mammals. In L. A. Dierauf & F. M. D. Gulland (Eds.), *CRC handbook of marine mammal medicine* (2nd Ed.) (pp. 741-766). Boston: CRC Press.

Aquatic Mammals 2005, *31*(3), 288-310, DOI 10.1578/AM.31.3.2005.288

2. Whales, Dolphins, and Porpoises: Presentation of the Cetaceans

Whales, dolphins, and porpoises live their entire lives in the water, and in the process of adapting to this environment, became the most specialised mammals. From the small vaquita (*Phocoena sinus*) to the giant blue whale (*Balaenoptera musculus*), cetaceans have adapted to the aquatic environment in very different ways yet have retained many common anatomical and behavioural characteristics.

This overview of the taxonomy, biology, and behaviour of cetaceans may help the reader to become familiar with aspects of the animals that are relevant to the design of their habitat.

Introduction

Whales, dolphins, and porpoises are marine mammals belonging to the order Cetacea. Other marine mammals belong to two other orders: the Sirenia, which includes the manatees (*Trichechus* sp.) and the dugong (*Dugong dugon*), and the very large mammalian order Carnivora, which includes the suborder Pinnipedia—seals (Phocidae family), sea lions (Otariidae family), and walrus (Odobenidae family)—and the suborder Fissipedia, or land-based carnivores, with the two families Mustelidae and Ursidae, which include the sea otter (*Enhydra lutris*) and the polar bear (*Ursus maritimus*). The sirenians are the only other marine mammals, besides cetaceans, to live their entire life in the water.

The order Cetacea is subdivided into two suborders: (1) Mysticeti or baleen whales and (2) Odontoceti or toothed whales. There are presently 14 known species of mysticetes and 72 known species of odontocetes (Rice, 1998; Anonymous, 2001; Beasley et al., 2005).

Despite their streamlined, fusiform body shape, cetaceans retain all the key characteristics of mammals: they are warm-blooded, breathe air with lungs, bear live young, nurse their young, and some even have traces of hair.

Most species kept in captivity are from the suborder Odontoceti. Apart from two stranded gray whale calves (Stewart, 2001), no baleen whale has been kept in captivity for a long period of time due to its large size.

The ancestor of all cetaceans is a small land mammal, *Mesonyx*, a quadrupedal animal that gave rise to the modern ungulates. Around 50 million years ago, this animal started colonising coastal fringes, swamps, and possibly the warm waters of the ancient Tethys Sea, later evolving into the cetacean ancestors, the suborder Archaeoceti. Between 38 and 25 million years ago, they diverged into the ancestors of the odontocetes and the mysticetes (Evans, 1987). The Delphinidae family is a relatively modern group, the largest and the most diverse among cetaceans, comprising the small Commerson's dolphin (*Cephalorhynchus commersonii*), the bottlenose dolphin (*Tursiops truncatus*), the large pilot whales (*Globicephala* sp.) and the killer whale (*Orcinus orca*). It is the cetacean family most widely represented in oceanaria.

Taxonomy

Tables 2.1 and 2.2 present the taxonomy of the two cetacean suborders. The classification and names follow Rice (1998) and Anonymous (2001).

Cetacean Biology and Behaviour

Anatomy, Physiology, and Life History

Cetaceans developed some unique adaptations to live a completely aquatic life. Their body is streamlined and hydrodynamic to minimise resistance in the water. Nostrils migrated to the top of the head to allow rapid air exchange while briefly surfacing. They exit the head as one external blowhole in odontocetes and two blowholes in mysticetes. Forelimbs have been reduced to paddle-like "flippers" that are used mainly for steering. The hind limbs evolved into boneless horizontal tail flukes, which provide powerful propulsion. Unlike the fish tail, which has a side-to-side movement, the tail flukes of a cetacean have an up-down motion. Most species have a dorsal fin that serves as a stabilizer and/or plays a social role (Geraci, 1986a; Evans, 1987).

Being warm-blooded, cetaceans use a lot of energy to maintain a stable body temperature in water where the heat exchange is 25 times faster than in the air. Instead of an insulating coat of fur or hair, which would impede swimming in the water, they developed an insulating layer of fat called blubber. Not all cetaceans lack hair, but most are vestigial and scarce. Hair is present at birth in some species, but quickly disappears,

Table 2.1. Taxonomy of the order Cetacea, suborder Mysticeti: baleen whales (Rice, 1998; Anonymous, 2001)

Family	Family common name	Species scientific name	Species common name
Balaenidae	Right & bowhead whales	*Balaena mysticetus*	Bowhead whale
		Eubalaena australis	Southern right whale
		Eubalaena glacialis	North Atlantic right whale
		Eubalaena japonica	North Pacific right whale
Neobalaenidae	Pygmy right whale	*Caperea marginata*	Pygmy right whale
Eschrichtiidae	Gray whale	*Eschrichtius robustus*	Gray whale
Balaenopteridae	Rorquals	*Balaenoptera acutorostrata*	Common minke whale
		Balaenoptera bonaerensis	Antarctic (southern) minke whale
		Balaenoptera borealis	Sei whale
		Balaenoptera edeni	Bryde's (Eden) whale
		Balaenoptera musculus	Blue whale
		Balaenoptera omurai	Discovered in November 2003
		Balaenoptera physalus	Fin whale
		Megaptera novaeangliae	Humpback whale

Table 2.2. Taxonomy of the order Cetacea, suborder Odontoceti: toothed whales (Rice, 1998; Anonymous, 2001)

Family	Family common name	Species scientific name	Species common name
Physeteridae	Sperm whales	*Physeter macrocephalus*	Sperm whale
Kogiidae		*Kogia breviceps*	Pygmy sperm whale
		Kogia sima	Dwarf sperm whale
Ziphidae	Beaked whales	*Berardius arnuxii*	Arnoux's beaked whale
		Berardius bairdii	Baird's beaked whale (North Pacific bottlenose whale)
		Hyperoodon ampullatus	Northern bottlenose whale
		Hyperoodon planifrons	Southern bottlenose whale
		Indopacetus pacificus	Indo-Pacific beaked whale (Longman's beaked whale)
		Mesoplodon hectori	Hector's beaked whale
		Mesoplodon mirus	True's beaked whale
		Mesoplodon europaeus	Gervais' beaked whale
		Mesoplodon bidens	Sowerby's beaked whale
		Mesoplodon grayi	Gray's beaked whale
		Mesoplodon peruvianus	Pygmy beaked whale
		Mesoplodon bowdoini	Andrew's beaked whale
		Mesoplodon bahamondi	Bahamonde's beaked whale
		Mesoplodon carlhubbsi	Hubb's beaked whale
		Mesoplodon ginkgodens	Ginko-toothed beaked whale
		Mesoplodon stejnegeri	Stejneger's beaked whale
		Mesoplodon layardii	Layard's beaked whale
		Mesoplodon densirostris	Blainville's beaked whale
		Tasmacetus shepherdi	Tasman (Shepherd's) beaked whale
		Ziphius cavirostris	Cuvier's beaked whale
Platanistidae	River dolphins	*Platanista gangetica*	Ganges and Indus (South Asian, or Indian) River dolphins
Iniidae		*Inia geoffrensis*	Boto (Amazon River dolphin)
Lipotidae		*Lipotes vexillifer*	Baiji (Chinese River dolphin)
Pontoporiidae		*Pontoporia blainvillei*	Franciscana (La Plata dolphin)

Monodontidae		*Delphinapterus leucas*	Beluga (white whale)
		Monodon monoceros	Narwhal
Delphinidae		*Cephalorynchus commersonii*	Commerson's dolphin
		Cephalorhyncus eutropia	Chilean dolphin
		Cephalorhyncus heavisidii	Heaviside's dolphin
		Cephalorhyncus hectori	Hector's dolphin
		Delphinus delphis	Short-beaked common dolphin
		Delphinus capensis	Long-beaked common dolphin
		Delphinus tropicalis	Arabian common dolphin (controversial), subspecies of *D. capensis**
		Feresa attenuata	Pygmy killer whale
		Globicephala melas	Long-finned pilot whale
		Globicephala macrorhyncus	Short-finned pilot whale
		Grampus griseus	Risso's dolphin
		Lagenodelphis hosei	Fraser's dolphin
		Lagenorhynchus acutus	Atlantic white-sided dolphin
		Lagenorhynchus albirostris	White-beaked dolphin
		Lagenorhynchus australis	Peale's (black-chinned) dolphin
		Lagenorhynchus cruciger	Hourglass dolphin
		Lagenorhynchus obliquidens	Pacific white-sided dolphin
		Lagenorhynchus obscurus	Dusky dolphin
		Lissodelphis borealis	Northern right whale dolphin
		Lissodelphis peronii	Southern right whale dolphin
		Orcaella brevirostris	Irrawaddy dolphin
		Orcaella heinsohni	Australian snubfin dolphin**
		Orcinus orca	Orca (killer whale)
		Peponocephala electra	Melon-headed whale
		Pseudorca crassidens	False killer whale
		Sotalia fluviatilis	Tucuxi
		Sousa chinensis	Pacific humpback dolphin
		Sousa plumbea	Indian humpback dolphin
		Sousa teuszi	Atlantic humpback dolphin
		Stenella attenuata	Pantropical spotted dolphin
		Stenella clymene	Clymene dolphin
		Stenella coeruleoalba	Striped dolphin
		Stenella frontalis	Atlantic spotted dolphin
		Stenella longirostris	Spinner dolphin
		Steno bredanensis	Rough-toothed dolphin
		Tursiops aduncus	Indo-Pacific bottlenose dolphin
		Tursiops truncatus	Bottlenose dolphin
Phocoenidae	Porpoises	*Neophocaena phocaenoides*	Finless porpoise
		Phocoenoides dalli	Dall's porpoise
		Phocoena dioptrica	Spectacled porpoise
		Phocoena phocoena	Harbour porpoise
		Phocoena sinus	Vaquita
		Phocoena spinipinnis	Burmeister's porpoise

* Jefferson & Van Waerebeek (2002)
** Beasley et al. (2005)

except for some species of baleen whales, which retain a few hairs. The thickness of the blubber layer varies among species and according to the water temperature of their habitat. They need to maintain a stable body temperature between 34 and 37º C in water temperatures varying from 0º to 32º C, according to the latitude of their habitat. Cetaceans have small appendages, thereby minimising heat loss by reducing the amount of skin in contact with water. The surface-to-volume ratio is small, which may explain the absence of small species at very high latitudes (Evans, 1987).

Figure 2.1a. Bottlenose dolphin

Figure 2.1b. Beluga whale

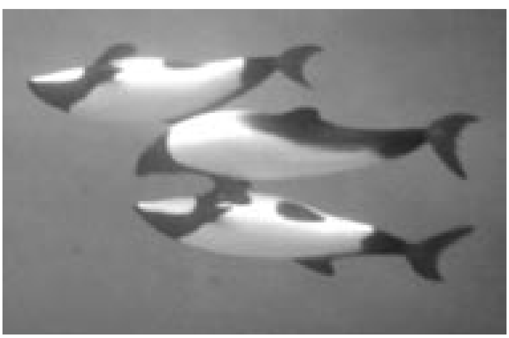

Figure 2.1c. Commerson's dolphins

Figure 2.1d. False killer whale

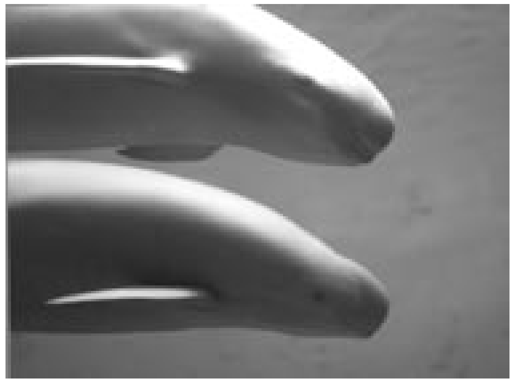

Figure 2.1e. Finless porpoises

Figure 2.1f. Harbour porpoises

Figure 2.1. Photographs showing various species of cetaceans kept in human care

Figure 2.1g. Irrawaddy dolphin

Figure 2.1h. Killer whale

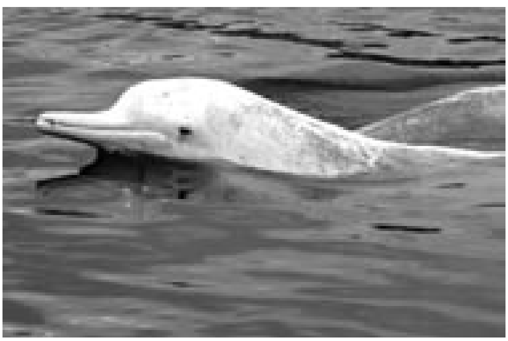

Figure 2.1i. Pacific humpack dolphin

Figure 2.1j. Pacific white-sided dolphin

Figure 2.1k. Risso's dolphin

Figure 2.1l. Rough-tooth dolphin (Photograph from C. Gaspard)

Figure 2.1m. Hybrid false killer whale x bottlenose dolphin

Maintaining the water temperature in captivity at the same level as the original habitat is critical. Although marine mammals have efficient ways of regulating temperature, exposing the animal to excessive heat is as life threatening as excessive cold (Geraci, 1986b). They can tolerate and adapt to some variation, but they should not be exposed to drastic and sudden changes to maintain a balanced energy budget. It is important to keep an animal wet, especially the flippers and flukes, during transportation, to prevent overheating.

Odontocetes have teeth, which they use to grasp prey or for social interactions. In some species, the teeth are barely apparent, or their number is greatly reduced, as in the narwhal or beaked whales. Odontocetes do not chew their food, but some dolphins behead fish by smashing or shaking. Other cetaceans swallow their fish whole, heads first. They eat a wide variety of fish, squid, and crustacean; and some larger whales, such as killer whales in the wild feed on other marine mammals and birds (see Chapter 7 for details). The smallest species of odontocete, the harbour porpoise, requires a daily portion of food estimated to be up to 13% of its body weight in winter (Evans, 1987), whereas the bottlenose dolphin may need between 3 and 6.5% of body mass per day (Barros & Odell, 1990), depending on the type of food, the water temperature, season, activity, and if females are pregnant or nursing (Reddy et al., 1994; Kastelein et al., 2003). In contrast, the baleen whales have plates of keratin, hanging from the palate, instead of teeth. These plates filter zooplankton, fish, and squid from the water into the mouth (Evans, 1987). Because Baleen whales require such a specialized diet and such large quantities of food, they rarely have been kept in captivity (Stewart, 2001).

Pelagic marine species live in a salty environment that is four times more concentrated than body fluids. The anatomy and physiology of cetaceans are aimed at reducing water loss. This can be observed in the small amounts of water lost during respiration and in the production of concentrated milk, faeces, and urine. Salt and water balance is achieved mainly through food, which provides water directly available or derived from the oxidation of fat. The fattier the fish eaten, the more water is available to the cetacean. As a result, a prolonged fast in a sick animal can quickly lead to dehydration (Geraci, 1986a).

Cetaceans are voluntary breathers, meaning that their breathing is under conscious control; the blowhole is normally closed. This is different from humans whose breathing is involuntary and a reflex. During sleep periods, dolphins rest each hemisphere of their brain separately while the other hemisphere is actively keeping the animal in motion and controling breathing. All cetacean species studied have been shown to exhibit unihemispheric slow-wave sleep (Mukhametov et al., 1977; Mukhametov, 1987, 1988; Mukhametov & Lyamin, 1994; Lyamin et al., 2002; Ridgway, 2002) or signs of this sleep state behaviourally (Lyamin et al., 1998, 2000; Sekiguchi & Koshima, 2003). During respiration, a cetacean exchanges about 80% of the air contained in their lungs as opposed to 17% for humans. Exhaling and inhaling usually takes less than one second.

Some cetaceans can hold their breath from several minutes to more than an hour. Bottlenose whales have been recorded under water for as long as 80 min, and sperm whales at depth of 3,195 m (Leatherwood & Reeves, 1983; Evans, 1987; Klinowska, 1991; Hooker & Baird, 1999). Many of the smaller whales and dolphins can dive to several hundred meters for up to 10 min. The necessity to dive at great depths and to hold their breath for long periods of time has brought about anatomical and physiological adaptations. These include the compressible rib cage and lungs, a slower heartbeat while diving, a shunting of the blood away from limbs and toward the heart and brain, and a high content of oxygen-binding protein—the myoglobin—which prevents muscles from oxygen deficiencies (Geraci, 1986a). In captivity, only shallow depths are available compared with great depths in the wild. Yet, it is important to provide animals with habitats as large and deep as possible to encourage diving and rapid swimming.

Adaptations of the epidermis reduce turbulence, and the streamlined shape of their bodies allows dolphins to swim fast. Recent studies have demonstrated that realistic maximum swimming speeds for dolphins are lower than previously reported, which were based on sparse data and imprecise measurement techniques. Bottlenose dolphins, false killer whales, and short-beaked common dolphins have reached maximum speeds between 28.8 and 29.5 km/h, and long-beaked common dolphins can attain the speed of 24.1 km/h (Rohr et al., 2002; Anonymous, 2003). Cruising speed of bottlenose dolphins is on average 6 km/h (Anonymous, 2003).

Cetaceans become sexually mature at different ages, depending on their species, but individuals seem to reach sexual maturity when they obtain a certain size, rather than a certain age. Gestation period varies from 7 to 12 months for dolphins and up to 18 for larger species such as the killer whale (Schroeder, 1995). Calving intervals vary from one to six years. Gestation, calving interval, and life spans are longer for larger species. The harbour porpoise, for example, is expected to live for up to 24 years (Read, 1999), the bottlenose dolphin for 50 years (Wells & Scott, 1999), the false killer whale and the short-finned pilot whale for 63 years (Evans, 1987; Odell & McClune, 1999), the killer whale for 80 to 90 years (Evans, 1987; Dahlheim & Heyning, 1999), and the sperm whale for 70 years (Rice, 1989). Among mysticeti whales, the minke whale can live for 50 years and the fin whale as long as 90 years (Evans, 1987). Bowheads can live up to 100 years (George et al., 1999). These are maximum known longevities, and in general, females live longer than males.

Senses

Smell and Taste—Dolphins most likely have no sense of smell in the form that exists in most mammals because they lack the peripheral olfactory structures; however, a sense of taste is present in dolphins. Little is known about olfaction or taste in baleen whales. Anatomical studies have shown the presence of taste buds at the base of the tongue of dolphins, and experimental studies have shown food discrimination capabilities and preferences. A bottlenose dolphin's ability to taste chemicals perceived by humans as sour and bitter is very good; acidic is nearly as good; and sweet is not as good as humans (Nachtigall, 1986). Little is known about the perception of chemical signals in the water, but dolphins and whales seem to be able to detect blood, sexual pheromones in the faeces and urine, and possibly alarm pheromones (Herman & Tavolga, 1980; Norris & Dohl, 1980; Nachtigall, 1986). Therefore, it is important to maintain a controlled aquatic environment as free of external chemicals, such as chlorine, as possible because their presence can prevent the detection of pheromones or destroy them (van der Toorn, 1987).

Touch—Cetaceans are highly tactile; their skin is well-innervated and extremely sensitive, with friction ridges in the epidermis similar to human fingerprints (Ridgway & Carder, 1990). Dolphins and whales frequently touch each other. They may stroke or pat each other with pectoral fins, flukes, or rostrum; rub bodies; or swim in physical contact during affiliative, play, or precopulatory behaviours (Herman & Tavolga 1980; Evans, 1987). Tactile contacts also may be aggressive such as tooth-raking, striking with the fins or flukes, or ramming with the head or rostrum. Dolphins also may use mechano-reception to coordinate movements with each other, using tactile receptors that respond to a mechanical stimulus, such as a change in pressure (Pryor, 1990). Tactile contacts are essential in cetacean social behaviours, displaying family and friendship bonds, hierarchy, and sexual interest, among others.

Vision—The cetacean's eyes are adapted for underwater vision. Bottlenose dolphins have good visual acuity both under water and in air (Nachtigall, 1986). Short range (1 m) visual acuity is better under water, whereas longer range (2.5 m) acuity is similar in the two media (Herman, 1989). Some dolphins, river dolphins, for example, have very poor vision and mainly rely on their echolocation abilities. Although appropriate receptors are present in the dolphin eye, experiments have shown that dolphins seem unable to discriminate colours. Because the eyes of most dolphins and whales are set far back on the head, they see monocularly laterally, but have binocular or stereoscopic forward vision downward and upward. As individual eye movements are not coordinated, this binocular vision might be only a visual overlap (Ridgway, 1986). Dolphins readily make eye contact, but it is usually brief and the dominant animal normally looks away first (Evans, 1987; Pryor, 1990).

Audition—Audition is the main sense used by cetaceans. The external ear is no more than a tiny hole in the skin just behind the eye. The inner ear, though small, is very well developed. Sound is used in two main ways: (1) echolocation and (2) communication. Echolocation involves an active process of emitting intense, short broadband pulses of sound called clicks between 0.250 to 220 kHz (but mainly in the ultrasonic range above 20 kHz) in a narrow beam, bouncing off objects in their path. From the resulting echoes, the animal is able to build an acoustic picture of its surroundings (Nachtigall, 1986; Evans, 1987). By using this complex system, dolphins and whales can determine size, shape, speed, distance, direction, and even some internal structure of objects in the water. Jones & Sayigh (2002) found that bottlenose dolphin echolocation characteristics vary by geographical region, activity state, and dolphin group size. Some dolphins seem to use this system sparingly in the wild, maybe because it advertises the dolphin's location to potential prey, predators, or competitors. Recent experiments show that bottlenose dolphins extensively use passive listening for long-range detection of soniferous prey during the search phase of the foraging process (Gannon et al., 2005). Other hypotheses have been presented on the ability of dolphins to detect objects and prey without emitting echolocation clicks, by using the echo of ambient noise. This Ambient Noise Imaging (ANI) technique might explain why dolphins can locate and catch prey while remaining silent, even in murky waters or blindfolded during experiments (Potter et al., 1997; Taylor et al., 1997).

Odontocetes have good functional hearing between 200 Hz and 100 kHz, although individual species may have functional ultrasonic hearing to nearly 200 kHz (Ketten, 1998). Bottlenose dolphins are most sensitive to sounds in the range of 500 Hz to 100 kHz, whereas humans hear well from about 200 Hz to 17 kHz. The majority of odontocetes have peak sensitivities in the ultrasonic ranges, although most have moderate sensitivity from 1 kHz to 20 kHz and no acute hearing below 500 Hz (< 80 dB re 1 μPa). Models indicate that the mysticetes' functional hearing range commonly extends down to 20 Hz. Several species are expected to hear well into infrasonic frequencies, from 20 Hz to 30 kHz (Ketten, 1998).

The odontocete larynx does not possess vocal cords. Instead, sounds are produced by nasal sacs in the blowhole region and controlled by organs called monkey lips (Cranford et al., 1997; Degollada et al., 1998). The fatty melon above the skull acts as an acoustical lens focusing and amplifying these sounds into a beam. Received sounds are transmitted to the inner ear by the bones and fatty channels of the lower jaw. Many odontocetes communicate with whistles and burst-pulse sounds. Whistles are continuous, frequency-modulated pure tones, varying in intensity and pattern, with one or more harmonics. The frequency of bottlenose dolphin whistles is generally between 4 and 24 kHz (Herman, 1980). Dolphins can whistle and echolocate at the same time. Burst-pulse sounds are another type of communication sounds. They are broadband signals resembling moans, trills, grunts, squeaks, and creaking doors. Sperm whales, members of the porpoise family, and members of the river dolphin family appear to produce only clicks and bursts of clicks, which may function for both echolocation and communication (Evans, 1987). Acoustical interference can have a dramatic effect on the behaviour and physiology of captive animals. Loud, human-made, underwater noises within their hearing frequencies can adversely affect cetaceans (Richardson et al., 1995). Acoustical interference, which cetaceans cannot escape in captivity, can cause endocrine changes, increased aggression, decreased appetite, and irreversible hearing loss (Stoskopf & Gibbons, 1994). The quality of their acoustical environment in human care is critical and often has been neglected. It is important to suppress human-made background noise, and to design a habitat that reduces sound reverberation (see Chapter 5 for details).

Cognition

Senses are used by animals to understand their environment. The cognitive characteristics of dolphins and whales are reflected in how the information is selected, encoded, stored, analysed, and retrieved by various sensory receptors. Little is known about baleen whales' cognitive abilities, but those of the bottlenose dolphins have been studied for many years. The bottlenose dolphin is a quick learner in most types of auditory tasks, including those having intricate conceptual demands. Its auditory memory is very faithful (Herman, 1980). The dolphin is capable of forming and generalising concept rules. Dolphins also seem proficient at imitative behaviours, and they are able to perform both vocal and motor mimicry, demonstrating that they can learn by observation. Despite the fact that attempts to demonstrate the existence of a natural language have failed, dolphins may be capable of comprehending a simple artificial language. Gestural language comprehension seems to be better if the visual information manipulated is dynamic (Herman, 1980, 1986, 1989). The extensive development of the dolphin's brain and the resulting cognitive skills have evolved from the demands of social living, and according to Herman (1989), they require "the acquisition and use of knowledge to facilitate an exchange of information within a mutually dependant social network." In other words, cetaceans may have developed their complex cognitive abilities because of the need for high flexibility and "communality" in their social organisation and behaviour (Johnson & Norris, 1986).

Social Life and Behaviour

Whales and dolphins are essentially social animals. Group size varies greatly among species, being anywhere from two to several thousand individuals; however, common social patterns and behaviours have been identified. Living in groups has several advantages: it maximises foraging, helps with defence against predators, brings individuals together for reproduction, and increases the efficiency of calf rearing (Evans, 1987). Some species, such as the killer whale, show stronger social cohesion than others. Cetaceans do not form stable long-term male-female bonds, as wolves do for example. The basis of the social unit is the reproductive female; most of the stable and long-lasting social interactions that are ongoing within a group are between adult females and their offspring (Sweeney, 1990). In bottlenose dolphins, multigenerational female bands tend to be composed of females who are related or who share long histories of association within a common home range. Females at the same stage of their reproductive cycle tend to swim together. Female cetaceans care for their offspring until they are weaned. Bottlenose dolphin calves typically remain with their mother for three to six years (Wells & Scott, 1999). A mother and young pair may remain together on an intermittent basis for long periods. Bottlenose dolphins may return to each other in times of stress many years after the offspring have reached adulthood (Norris & Dohl, 1980). Mothers continue to care for their young until the following pregnancy and some even longer. Other females sometimes assist the mother during birth and in supervising calves while the mother is hunting or diving to great depths where calves cannot follow (Johnson & Norris, 1986; Evans, 1987). In captivity, the oldest, parturient female often is observed to be the "focal point on which the social activity of the tank is centred" (Tavolga, 1966, quoted in Johnson & Norris, 1986, p. 338). Sexual segregation occurs commonly in dolphin schools (Norris & Dohl, 1980). Many oceanic cetacean juvenile males form separate groups, and adult males travel alone, in tightly bonded pairs, or in small groups, joining the basic social group for brief periods, primarily for breeding purposes (Sweeney, 1990; Wells & Scott, 1999). In some populations, bonds between males of similar age develop early in life, and pair bonds may be maintained for 20 years or more (Wells & Scott, 1999). Males and females also may form sex-segregated alliances when they encounter resources and cooperate in competition for these resources with other conspecifics (Connor et al., 2000a, 2000b).

Play, courtship, and sexual behaviours have an important role in the social life of whales and dolphins and are probably more developed in captivity since the time dedicated to the feeding is reduced and traveling is suppressed. Play is commonly seen among young animals and may serve as learning and practice progress. Adults also play with youngsters or among themselves. They frequently leap, chase each other, or engage in preludes of courtship. Social and sexual behaviours are carried out throughout life, and they probably function not only in courtship, but as a means to maintain familiarity between group members (Evans, 1987). These behaviours are sometimes directed towards other species and humans. Other social behaviours include altruistic and care-giving behaviours. It may take the form of cooperation such as collective food herding, assistance to a sick animal, "standing by" when a school mate is captured, rescue and defence of a threatened member of the group, and the assistance given by the mother to her calf (Johnson & Norris, 1986). Care-giving behaviour has been observed in captivity among species such as the bottlenose dolphin, the pilot whale, the common dolphin, and the Pacific white-sided dolphin, and between individuals of different species (Defran & Pryor, 1980; Johnson & Norris, 1986).

For other detailed and recent sources on cetacean life, behaviour, and physiology, please refer to Reeves et al. (1999), Reynolds & Rommel (1999), Mann et al. (2000), and Perrin et al. (2002).

Possible Impact of Controlled Environment on Behaviour

Life in a controlled environment may impede certain aspects of normal social dynamics. Aggressive hierarchical dominance may be naturally occurring in the wild, but in captivity, it can disrupt the group and harm subordinates (Geraci, 1986b). The situation in captivity is altered because adult males interact permanently with the social unit. Dominance hierarchies can be established in both sexes. Large adult males can dominate other poolmates, and the largest and oldest females may dominate younger or smaller males and females (Östman, 1991; Samuels & Gifford, 1997; Wells & Scott, 1999). In captivity, this male dominance is often the source of many social and behavioural problems, especially related to juveniles within the social group (Johnson & Norris, 1986; Sweeney, 1990). The aggression often reported in adult male bottlenose dolphins towards infants and juveniles in captivity may reflect a tendency on the part of such adults to herd the young when threatened in the wild. This behavior may escalate into obsessive hostility in captivity where the young can neither escape nor be buffered from the male by a tightly packed crowd of adults (Johnson & Norris, 1986). Since they are unable to leave the area to avoid the situation, stress, psychological, and physical trauma can occur. Attempts to solve problems caused by dominance or other compatibility problems in the enclosure are challenging. These disruptions also can be caused by the fact that some of the animals may have been removed from their original social structure, separated from family members or a social unit, and now have to adjust to a new social environment. Many surveyed facilities emphasized the importance of recreating an adequate social structure in captivity, especially by avoiding mixing adult males together in the presence of females or adult males with new juvenile males, and by providing ample space and a multiple pool complex to allow separation and escape. In the wild, vertically organised swimming arrangements have been seen among groups of pantropical spotted dolphins, Pacific white-sided dolphins, and bottlenose dolphins, and in captivity, among spinner dolphins. It has been hypothesised that these were dominance arrangements, expressed as priority of access to the surface (Herman & Tavolga, 1980; Norris & Dohl, 1980; Johnson & Norris, 1986; Pryor, 1995). I have made similar observations in a dolphinarium in Japan about bottlenose dolphins and false killer whales living in a restricted space. In a crowded square tank, the dominant animal, a large adult male bottlenose dolphin, was always seen swimming closer to the surface and to the edge of the tank, enjoying the optimal swimming surface available and having to make little effort to surface to breathe. Below him in the hierarchy was an adult female false killer whale that also enjoyed the upper layer and the outer periphery of the tank. The lowest ranking animals, a juvenile female false killer whale and a juvenile male bottlenose dolphin, always swam near the bottom of the tank and nearer to the centre. They were constantly displaced by other animals and heavily marked with tooth rakes. Whenever they came to the surface to breathe, they returned immediately to the bottom of the pool.

Although stereotypy has not clearly been demonstrated for cetaceans in captivity, some behaviours tend to occur when space is limited; the environment does not provide occupational activity; and when animals are kept alone, deprived of stimulus diversity, or are subject to environmental stress. It is generally true that the smaller the habitat, the more likely the animal is to behave stereotypically (Gygax, 1993; Carlstead, 1996); however, the shape also plays a role. Varied shapes and enriched environment where animals have opportunities to forage, play, and socialize with conspecifics and escape aggressive behaviours will greatly reduce chances of stereotypical behaviours.

In captivity, not all cetacean species have been kept routinely or with equal success. Additionally, the ease of maintaining a species in a tank seems to reflect, in part, the ecological characteristics of the natural habitat of the species. It is not surprising that species-specific behaviours will be largely maintained across diverse environments and circumstances. Some populations of bottlenose dolphins, accustomed to shallow, coastal waters, find it easier to adjust to tank conditions, whereas open-ocean pelagic species, such as the Dall's porpoise, that travel long distances in the wild, seem to have much greater difficulty in adapting to the tank environment (Defran & Pryor, 1980).

Although most countries forbid the capture of wild cetaceans, there is an increasing number of new public facilities displaying cetaceans in countries with less stringent regulations. This may lead to the exhibition of species infrequently or never previously displayed. Animals from strandings, which are kept for long-term rehabilitation or that cannot be released for medical reasons, also can belong to species never before kept in human care. Bearing this in mind, the Cetacean Species Information Table (Table 2.3) was compiled to present several environmental, physiological, and behavioural parameters for all species kept in institutions that can be useful in the design of their environment.

Introduction to the Cetacean Species Information Table

The United States, Australia, and some European Union countries such as the United Kingdom, France, and Sweden (Klinowska & Brown, 1986) developed strict regulations regarding marine mammal display, handling, medical care, transportation, and housing (see the "Regulatory Offices and Organisms" section in Chapter 8). Many other countries have rules that are either legally too broad to be applied, or they lack the executive means to enforce them. Therefore, exhibit dimensions and quality, life support system requirements, medical care, and husbandry can tremendously vary among countries. In the United States, the standards for the maintenance of marine mammals in research or display facilities are regulated by the Animal and Plant Health Inspection Service (APHIS) under the U.S. Animal Welfare Act (Anonymous, 1979-1984, 1995; Corkeron, 2002). "Facilities and Operating Standards," "Animal Health and Husbandry Standards," and "Transportation Standards," on which several other countries have based their own regulations, provide minimum requirements for enclosure sizes and volume of water, and recommendations for humane handling of animals and transportation methods. Size recommendations have been calculated on the animals' Average Adult Length (AAL) (rough estimates of AAL in feet that are then multiplied in meters). Although it is not stated in the regulations, it has been suggested that the species differentiation into two categories was made according to their inshore or pelagic habitat.

The fact that calculations are made on AAL is subject to discussion as individual animals may greatly exceed it. In some cases, the discrepancy may be a meter or more. For architects and planners, it would be more logical to base pool and enclosure dimensions on the maximum known adult body length, or Maximum Adult Length (MAL). This would ensure that animals are given ample space to display natural behaviours. I suggest that the MAL of the largest sex of the largest species housed in a pool be used to estimate the appropriate dimensions. Table 2.3 provides MAL by species and sex.

Although the classification of species according to their habitat is justified, it is no longer adequate in regards to recent research findings. For example, species like the Risso's dolphin, false killer whale, and pilot whale are known to be pelagic, yet they are classified with inshore species; while inshore species, such as the finless porpoise, are classified with offshore species. This classification was made at a time when little was known about many cetacean species in the wild. In the following table, species have not been classified according to their habitat, but its description is included. Planners are encouraged to exceed legal minimum size recommendations in surface area, depth, and volume, especially for species described as pelagic and gregarious.

The Cetacean Species Information Table provides information on all species that have been kept in captivity in the past or are presently displayed, as well as all species listed as vulnerable, endangered, or critically endangered by the International Union for Conservation of Nature and Natural Resources (IUCN), also known as the World Conservation Union (Anonymous, 2004). Because the habitat of such species is irreversibly being destroyed, it might prove necessary in the near future to attempt to breed these species in captivity to ensure their survival (Klinowska, 1991; Ridgway, 1995). It does not take into account all species briefly kept for rehabilitation from strandings. At the time of writing, 18 species of odontocetes are kept in human care on a permanent basis (vs. short-term rehabilitation). The table is listed in alphabetic order by scientific names, with an asterisk indicating those species currently kept in human care.

Information on each species is presented as follows:

Scientific name – English common name-*=kept in captivity at present • IUCN status • geographical distribution • habitat range • most frequent group size; rarer group size • behaviour in the wild; associations with other species in the wild • behaviour in captivity • success rate in captivity; breeding.

The MAL for males and females and the maximum known adult body weight or Maximum Adult Weight (MAW) for male and female is also displayed in the table. It can be noted that the MAW does not necessarily correspond to the MAL, as sometimes the weight of the largest measured animals has not been recorded. MAW is just indicative.

Finally, this table displays water temperature range in the wild as an indication of what the water temperature range should be in a controlled environment. It is important to know the initial place of capture, stranding, or previous housing to respect the animal's original environment temperature. For example, a bottlenose dolphin coming from northern Japan and one coming from the Gulf of Mexico might not be comfortable with the same water temperature at first. Therefore, facilities are advised to obtain information on water temperature averages at the place of origin in order to adjust the temperature range in the controlled environment.

Data have been compiled from literature on both wild and captive animal studies, mainly extracted from Klinowska (1991), the reference series *Handbook of Marine Mammals* by Ridgway & Harrison (1989, 1994, 1999), the *IUCN Red List* (Anonymous, 2004), and the survey. Other references are made to Defran & Pryor (1980), Leatherwood & Reeves (1983), Jefferson et al. (1993), Ridgway (1995), and the *Marine Mammal Inventory Report* (Anonymous, 1997). I suggest readers consult the *Aquatic Mammals* and *Marine Mammal Science* journals, among others, for regular updates on species distribution and population dynamics and for new anatomical, physiological, and behavioural findings. The website of the Convention of Migratory Species (CMS) provides a good online review of small cetacean species and is regularly updated (Culik, 2003).

Table 2.3. Cetacean species information table; MAL = Maximum Adult Length in m, Temp. = water temperature in Celsius degrees, MAW = Maximum Adult Weight in kg, * = species currently housed in captivity

Species characteristics	MAL male	MAL female	MAW male	MAW female	Temp.
Suborder mysticeti					
Balaenoptera acutorostrata – Common minke whale					
Cosmopolitan: tropics to ice edge worldwide; some may migrate • inshore / coastal / pelagic • single to herds of hundreds • small number unsuccessfully kept in captivity	9.80	10.70	14,000		0º-30º
Eschrichtius robustus – Gray whale					
Tropics to Arctic, North Pacific • shallow continental shelf • groups ≤ 3; up to 16 and larger aggregations • common aerial behaviour; migrating species • stranded calves have been rescued; two have been successfully rehabilitated and released • adults unsuitable for captivity because of large size	14.60	15.00	35,000		0º-28º
Suborder odontoceti					
Cephalorhynchus commersonii – Commerson's dolphin *					
Cold temperate to temperate; near Argentina and Kerguelen Islands • coastal waters max. 100 m deep • single to 3; up to 100 and more • quick, active animal; fast swimmers, leaping; short attention span in training; reacts quickly and excitedly to changes; cooperative • in Argentina, often associated with Burmeister's	1.67	1.75	Kerguelen 45	66	5º-15º
porpoise, Peale's, and Chilean dolphins • found only in two small and distinct geographical areas, but popular in display facilities in North America, Europe, and Asia • successfully kept and bred in captivity	1.41	1.47	Argentina 78	86	2º-8º
Cephalorhynchus hectori – Hector's dolphin • IUCN status: Endangered					
New Zealand temperate waters • coastal, shallow waters • pairs to 8; up to 50 • active, playful, acrobatic dolphins • small number not successfully kept in captivity so far	1.38	1.53	53	57	6º-22º
Delphinapterus leucas – Beluga whale * • IUCN status: Vulnerable					
Arctic/subarctic to cold temperate, Northern Hemisphere • shallow coastal waters to up river • groups ≤ 15; often several hundreds, up to thousands • highly gregarious, but not showy at surface; swim slowly; deep divers; tolerance to very shallow water; very loquacious • easily trainable for acoustic performances; highly manipulative • successfully kept and recently bred in captivity	5.72	4.74	2,376	1,457	0º-10º
Delphinus delphis, D. capensis – Short-beaked and long-beaked common dolphins *					
Temperate to tropical/subtropical waters around the world • coastal to offshore, mostly pelagic • groups ≤ 30; up to 10,000 • highly gregarious and social, active and energetic, aerial and highly vocal • moderately successfully kept in captivity; requires space; suffers from isolation; poor breeding except with one individual *D. capensis*, which sired several hybrid offsprings; one gave birth to several calves	2.60	2.30	136		15º-28º
Feresa attenuata – Pygmy killer whale					
Tropical to subtropical waters around the world • pelagic • groups ≤ 15; up to several hundreds • look very similar to melon-headed whale, but slower and more lethargic; might hunt dolphins in the wild • unsuccessfully kept in captivity; probably unsuitable because of aggressiveness toward trainers and other cetaceans	2.87	2.45	~225		20º-28º

Globicephala macrorhynchus – Short-finned pilot whale *					
Warm temperate to tropical waters around the world • deep offshore waters • groups of 10 to 50; up to several hundreds • strong social bond; highly structured female-based pod; deep divers; often associated with other species like bottlenose, Pacific white-sided, Risso's dolphins, and sperm whales • limited play activity in captivity; little fear to novelty, trainable • high mortality rate in captivity and unsuccessful breeding so far	7.20	5.50	~3,500	~2,000	15º-28º
Globicephala melas – Long-finned pilot whale					
Subpolar to temperate; all oceans except North Pacific • coastal to oceanic waters • groups of 10 to 50; up to several hundreds • highly social, stable pods; deep divers • not successfully kept in captivity; no breeding so far	7.62	5.70	~4,000	~2,500	5º-15º
Grampus griseus – Risso's dolphin *					
Temperate to tropical waters around the world • pelagic; deep waters, more than 1,000 m • groups of 5 to 45; up to several thousands • highly gregarious; energetic with aerial behaviour, deep divers; deliberate, non-aggressive; often associated with Pacific white-sided, northern right-whale dolphins, Dall's porpoises, and gray whales • easily trainable, but slower than bottlenose dolphin; successfully kept in captivity, but breeding has produced only hybrids so far	3.83	4.09	~500		10º - 28º
Inia geoffrensis – Boto or Amazon River dolphin * • IUCN status: Vulnerable					
Amazon and Orinoco river systems • fresh water only; not found in estuaries • groups ≤ 3; up to 15 • curious, playful, and enjoy tactile contacts; like chasing and ambushing; aggressive toward trainers and tank mates even of same species when confined in restricted spaces; requires provision of space for separation; very trainable • moderately successful in captivity; high mortality rate during capture and transportation; breeding occurred, but calves did not survive • some individuals lived for very long periods and are still currently housed in some facilities	2.74	2.28	160	96	25º-30º
Kogia breviceps – Pygmy sperm whale					
Temperate to tropical waters around the world • deep, offshore waters - groups ≤ 6 • not gregarious; slow and discrete; deep divers • few stranded individuals unsuccessfully kept in captivity	3.40		408		15º-28º
Kogia sima – Dwarf sperm whale					
Temperate to tropical waters around the world • deep, offshore waters • groups ≤ 5; up to 10 • similar to the pygmy sperm whale; lives in slightly warmer water; shy and undemonstrative • few stranded individuals unsuccessfully kept in captivity	2.70		272		15º-28º
Lagenodelphis hosei – Fraser's dolphin					
Tropical waters around the world • deep, offshore waters and deep coastal waters • groups ≤ 4; up to hundreds or thousands • often associated with melon-headed, false killer whales, and *Stenella* sp. dolphins • shy, nervous, and refuse to eat; fragile in captivity; unsuccessfully kept so far; probably unsuitable for captivity	2.70	2.64	209	164	25º-28º
Lagenorhynchus acutus – Atlantic white-sided dolphin					
Subpolar to cold temperate, North Atlantic • deep oceanic waters • herds of 50 to 100; up to several hundreds • active and acrobatic; highly gregarious • unsuccessfully kept in captivity so far; two rehabilitations from stranding successful	2.82	2.43	234	182	5º-15º

Lagenorhynchus albirostris – White-beaked dolphin					
Subpolar to cold temperate, North Atlantic • deep oceanic waters • groups of 7 to 35; up to several hundred • active, leaping and breaching; often associated with baleen whales and Atlantic white-sided dolphin; travel long distances • unsuccessfully kept in captivity so far; one rehabilitation from stranding successful	3.15	3.05	354	306	5º-15º
Lagenorhynchus obliquidens – Pacific white-sided dolphin *					
Cold temperate to temperate, North Pacific • deep offshore waters; sometimes near shore • herds ≥ 100; up to thousands • highly gregarious, acrobatic, playful, leaping, and somersaulting; often associated with Risso's, Northern right whale dolphins, and many other species • very trainable, especially at aerial behaviours; successfully kept in captivity; recent successful breeding	2.50	2.36	198	145	10º-20º
Lagenorhynchus obscurus – Dusky dolphin					
Cold temperate to temperate, Southern hemisphere • coastal • groups ≤ 20; up to several hundreds or thousands • highly social and gregarious; very acrobatic; high individual to individual fidelity; often associated with common and southern right-whale dolphin and pilot whales • moderately successful in captivity; no breeding so far	2.11	1.93	85	78	10º - 15º
Lipotes vexillifer – Baiji or Yangtze River dolphin • IUCN status: Critically endangered					
Yangtze River system and connected large lakes • shallow water • pairs to 6; up to 13 • shy animal; shallow surfacing • population near extinction; probably only few individuals still alive; creation of semi-natural reserve for breeding and conservation purposes in China without success; probably extinct before the end of the decade • one male remained in captivity for 22 years until 2002; no female survived; no breeding	2.16	2.53	125	167	10º-25º
Lissodelphis borealis – Northern right whale dolphin					
Cold temperate to warm temperate, North Pacific • oceanic waters • herds of 100 to 200; commonly up to 3,000 • highly gregarious; fast swimmers; deep divers; often associated with Pacific white-sided dolphins • very small number unsuccessfully kept in captivity so far	3.10	2.30	113		8º-19º
Lissodelphis peronii – Southern right whale dolphin					
Subantarctic to cold temperate, southern hemisphere • oceanic waters and deep coastal areas • groups of 2 to 30; most commonly herds of 200, sometimes up to 1,000 • highly gregarious; energetic swimmers; common aerial display; possibly deep divers; often associated with hourglass, dusky, and Pacific white-sided dolphins, and pilot whales • no information on suitability for captivity	2.97	2.30	116		1º-20º
Mesoplodon bidens – Sowerby's beaked whale					
Cold temperate to temperate, North Atlantic • deep oceanic waters • single or pairs, maybe up to 15 • rare pelagic species; discrete and shy • one unsuccessful attempt to keep a stranded calf in captivity; unusual swimming patterns; did not adapt to confinement and injured itself by bumping into walls • probably unsuitable for captivity	5.50	5.05	~1,400		5º-20º
Mesoplodon densirostris – Blainville's beaked whale					
Temperate to tropical waters around the world • deep oceanic waters • single or pairs; sometimes groups of 3 to 7 • rare pelagic species; discrete and shy • unsuccessful attempts to keep it in captivity from stranding; probably unsuitable for captivity	6.40	4.71	~1,400		15º-28º

Monodon monoceros – Narwhal (Size not inclusive of tusk length, estimated maximum 3 m)					
Arctic waters • deep inshore to open waters • pairs to 10; up to hundreds or thousands • social; rarely aerial; very vocal; migrating • unsuccessfully kept in captivity; probably unsuitable for captivity because of the presence of the tusk in males, unless in large ocean pens or habitat	4.70	4.15	1,600	1,000	0º-5º
Neophocaena phocaenoides – Finless porpoise *					
Warm temperate and tropical Indo-Pacific seas from Persian Gulf to northern Japan, Yangtze River, and other Asian rivers • fresh/brackish/marine; coastal/estuarine/riverine • singles or pairs; up to 12, sometimes up to 50 • shy and inconspicuous; different colour according to populations, from light gray to black • playful in captivity; easily trainable; successfully kept and bred	2.01	2.00	72		15º - 30º
Orcaella brevirostris – Irrawaddy dolphin *					
Tropical and subtropical Indo-Pacific seas and rivers • fresh/brackish/marine; coastal /estuarine /riverine • groups ≤ 6; up to 15 • quiet, low leaping; sometimes seen in same area as bottlenose and Pacific humpback dolphins • playful, easily trainable in captivity, and moderately successfully kept and bred, but limited to regional oceanaria	2.75	2.32	190		20º-30º
Orcinus orca – Killer whale *					
Cosmopolitan: tropics to ice edge waters around the world • inshore to oceanic; shallow to deep waters • pairs to 20; up to 100 • highly social, very vocal, and inquisitive; common aerial behaviour; stable matriarchal societies • some populations feed on fish; others on marine mammals • playful, easily trainable in captivity; can be aggressive, several attacks on trainers occurred; successfully kept and bred in captivity around the world	9.80	8.50	~10,000	~7,500	0º-30º
Peponocephala electra – Melon-headed whale					
Tropical to subtropical waters around the world • deep oceanic waters • herds of 100 to 500; up to 2,000 • highly social; move at high speed, leaping; associated with other species like Fraser's dolphins • unsuccessfully kept in captivity apart from one individual that displays some aggressiveness toward trainers and tank mates • one young individual successfully rehabilitated from stranding from 1998 to 2003	2.73	2.75	~275		20º-28º
Phocoena phocoena – Harbour porpoise * • IUCN status: Vulnerable					
Subarctic to temperate, Northern Hemisphere • shallow, near shore waters, max. 200 m deep • groups ≤ 8; sometimes 50 to several hundreds • fast moving, inconspicuous animal; complex social behaviour; easily frightened; little aerial behaviour • never aggressive toward humans in captivity; inventive; difficult to maintain in captivity because high mortality rate so far, but successfully kept or rehabilitated in small number of oceanaria	1.43	1.89	50	65	5º-15º
Phocoena sinus – Vaquita • IUCN status: Critically endangered					
Warm waters of the northern Gulf of California • shallow waters max. 36 m • single to 4; up to 8 or 10 • shy and elusive animal, living in an extremely small geographical area; highly endangered with population in the low hundreds • no Vaquita has been kept in captivity, but it might be considered as conservation measure	1.45	1.50	48		20º-28º
Phocoena spinipinnis – Burmeister's porpoise					
Cold temperate to warm temperate around South America • coastal/estuarine shallow waters • groups of ≤ 6; up to 70 • inconspicuous, unobtrusive swimming; little aerial behaviour; only one animal kept in captivity; unsuccessfully rehabilitated from stranding	~2.00	1.91	78	105	5º-20º

Phocoenoides dalli – Dall's porpoise					
Subarctic to temperate, Northern Pacific • deep waters • pairs to 12; sometimes up to several thousands • social animal; very fast swimmers; rare aerial behaviour; often associated with baleen whales • unsuccessful in captivity, throwing itself against walls and bottom; refuses to feed, nervous, irritable, subject to infection and skin slough; reacts badly to space inhibition • probably unsuitable for captivity	2.39	2.10	200		5º-20º
Physeter macrocephalus – Sperm whale • IUCN status: Vulnerable					
Cosmopolitan • deep inshore and offshore waters • groups of 20 to 40; up to 150, rarely up to thousands • probably the deepest diver of all cetaceans; few attempts to rehabilitate stranded young animals • unsuitable for captivity because of large size	18.30	12.50	57,000	24,000	5º-28º
Platanista gangetica (P. g. gangetica and minor) – Ganges River dolphin, Indus River dolphin or Susu • IUCN status: Endangered					
Ganges, Brahmaputra, Karnaphuli-Sangu, Meghna, and Indus river systems • shallow, murky waters • single or pairs; often ≤ 10, sometimes up to 25 • highly threatened habitat; population in low hundreds • active animal; solitary, rarely aerial; echolocates constantly • unsuccessfully kept in captivity, but as highly endangered species, captivity and relocation in semi-natural reserves could be re-envisaged as a conservation measure	2.12	2.52	84		20º-28º
Pontoporia blainvillei – Franciscana *					
Temperate to warm temperate waters from central Argentina to central Brazil • shallow coastal and estuarine waters; sometimes waters of La Plata River; depth max. 36 m • single or pairs, up to 15 • shy and inconspicuous animal, not gregarious, often solitary; population limited to small geographical area; species probably threatened • one individual is successfully kept in captivity in Argentina	1.58	1.74	35	52	10º-25º
Pseudorca crassidens – False killer whale *					
Temperate to tropical waters around the world • deep offshore waters • groups of 10 to 20; sometimes up to several hundreds • gregarious, strong social cohesion; deep divers; lively and fast swimmers • highly adaptive, inquisitive, quick observational learning and malleable; strong bonds occur with other species in captivity • not very successfully kept in captivity because of fragile health • only one calf of the species born in captivity alive so far; several hybrids born between male false killer whale and female bottlenose dolphin; one hybrid still alive and bred several offspring	6.10	5.06	~1,400		10º-28º
Sotalia fluviatilis – Tuxuci *					
Freshwater population in Amazon and Orinoco river systems; marine population along Atlantic coast of northern part of South America • near shore and estuarine • groups of 2 or 3; up to 30 • shy, difficult to approach; aerial behaviour • can go into shock during handling; timid and agitated in captivity; can be aggressive to other species • not very successful in captivity; breeding occurred	Riverine 1.52 Marine 1.49	1.87 2.06	53		25º-30º
Sousa chinensis – Pacific humpback dolphin *					
Warm temperate to tropical waters; coastal rim of Indian Ocean, South-East Asia, and Australia • coastal/estuarine, enters river and mangroves; shallow waters less than 20 m deep • groups of 3 to 5; up to 25 • quiet, moderately acrobatic, slow swimmers; sometimes associated with bottlenose dolphins • riverine and estuarine population may be vulnerable because of habitat destruction • moderately successful in captivity; kept in small numbers in few regional oceanaria	3.20	2.49	284		15º-30º

Stenella attenuata – Pantropical spotted dolphin *							
Tropical to subtropical waters around the world • coastal/pelagic • herds ≤ 100; up to several thousand • gregarious animal; fast swimmers; acrobatic • some individuals kept in captivity for several years, but difficult to train; generally unsuccessful in captivity • members of the *Stenella* sp. are timid, easily frightened, fearful of objects; highly dependent on the presence of other dolphins and react adversely to novelty • only 3 births reported for *Stenella* sp., and none survived • probably unsuitable for captivity • 2 individuals kept from strandings in Florida at present	2.57		2.40		119		25º-28º
Stenella clymene – Clymene dolphin							
Tropical to subtropical, Atlantic Ocean • deep oceanic waters • groups of 8 to 15; up to 50 • quick and agile, acrobatic aerial behaviour; associates with spinner and common dolphins • stranded individuals unsuccessfully rehabilitated in captivity • probably unsuitable for captivity		1.97			79		25º-28º
Stenella coeruleoalba – Striped dolphin							
Temperate to tropical waters around the world • deep oceanic waters, sometimes deep coastal water • groups of 10 to 30; up to 100 to 500, sometimes several thousands • fast swimmers, easily alarmed, nervous; resists handling and training in captivity • unsuccessfully kept so far; probably unsuitable for captivity	2.56		1.90		156		15º-28º
Stenella frontalis – Atlantic spotted dolphin							
Warm temperate to tropical, Atlantic Ocean • shallow coastal to deep oceanic waters • groups ≤ 15; up to 50 • complex social organization; fast swimmers • not successful in captivity; sometime refuses to eat; does not handle isolation; high mortality rate • probably unsuitable for captivity	2.26		2.29		143		20º-28º
Stenella longirostris – Spinner dolphin							
Tropical to subtropical waters around the world • inshore and oceanic waters • groups of 50 to several thousands • highly gregarious; aerial, acrobatic, famous spectacular spin; fast swimmers; often associated with pantropical spotted dolphin • unsuccessfully kept in captivity; many animals died of capture shock • like other pelagic small cetaceans, does not adapt well to enclosed space • probably unsuitable for captivity	2.35		2.04	75		65	25º-28º
Steno bredanensis – Rough-toothed dolphin *							
Warn temperate to tropical waters around the world • deep oceanic waters • groups of 10 to 20; up to 50, sometimes several hundreds • social animal; probably deep divers, often associated with pilot whales, bottlenose, pantropical spotted, and spinner dolphins • easily trainable; innovative and not easily frightened in captivity; bold and investigative; highly manipulative; long attention span for prolonged and complex tasks; but hot-tempered; more volatile and aggressive than bottlenose dolphin • moderately successful in captivity; only one hybrid with male bottlenose dolphin so far • few individuals in rehabilitation at present	2.65		2.55		155		25º-28º

Tursiops truncatus – Bottlenose dolphin *					
Temperate to tropical waters around the world • primarily coastal, but also oceanic; shallow to deep waters, mainly over continental shelf • pairs to 20; sometimes up to several hundreds • highly social animal; complex social behaviour; active and aerial, vocal; opportunistic feeder • highly polymorphic due to cosmopolitan distribution; often associated with pilot whales and spotted, Risso's, and rough-toothed dolphins • the most common species of cetaceans kept in captivity; highly adaptable and easily trainable; manipulative, curious, and acrobatic; forms strong bonds with conspecifics as well as other species • body dimensions should be considered in respect to population's origin; body size seems to vary inversely with water temperature • successfully kept and bred in captivity	3.81	3.67	282	263	10º-30º

Hybrids in captivity:

F. Tursiops truncatus x *M. Pseudorca crassidens* = This female hybrid gave birth to three second-generation hybrids sired by male bottlenose dolphins, one just born and alive at present in Hawaii. Four other hybrids births, including three still-born recorded in Japan; the fourth lived a short time.

F. Tursiops truncatus x *M. Grampus griseus* = 13 hybrids births, including seven still-born, recorded in Japan. They lived from three weeks to several months, and one survived for six years.

F. Tursiops truncatus x *M. Lagenorhyncus obliquidens* = This hybrid recorded in Japan did not survive.

F. Tursiops truncatus x *M. Delphinus capensis* = Four calves born in 11 years in institution in California; two still alive in 2003. One hybrid gave birth to calf sired by *Tursiops truncatus* in 2000.

F. Steno bredanensis x *M. Tursiops truncatus* = Three records of captive-born hybrids; one female lived for four years in Hawaii.

F. Globicephala macrorhyncus x *M. Tursiops truncatus* = This near-term foetus was recorded in California.

Literature Cited

Anonymous. (1979-1984, 1995). *Specifications for the humane handling, care, treatment and transportation of marine mammals* (9 CFR, Subpart E, §3.100-118). Washington, DC: U.S. Department of Agriculture, the Animal and Plant Health Inspection Service.

Anonymous. (1997). *Marine mammal inventory report.* Washington, DC: U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 673 pp.

Anonymous (International Whaling Commission [IWC]). (2001). Annex U: Report of the working group on nomenclature. *Journal of Cetacean Research and Management*, *3*, 363-365.

Anonymous. (2003). How fast do dolphins really swim? (SD 523, SPAWAR, U.S. Navy Marine Mammal Program, SSC San Diego). *Proceedings of the 31st International Marine Animal Trainer's Conference*, Long Beach, California.

Anonymous. (2004). *The IUCN (World Conservation Union) red list.* Available online: www.iucnredlist.org/. Accessed 7 July 2005.

Barros, N. B., & Odell, D. K. (1990). Food habits of bottlenose dolphins in the Southern United States. In S. Leatherwood & R. R. Reeves (Eds.), *The bottlenose dolphin* (pp. 309-328). New York: Academic Press.

Beasley, I., Robertson, K. M., & Arnold, P. (2005). Description of a new dolphin, the Australian snubfin dolphin *Orcaella heinsohni* sp. n. (Cetacea, Delphinidae). *Marine Mammal Science*, *21*(3), 365-400.

Carlstead, K. (1996). Effects of captivity on the behavior of wild mammals. In D. G. Kleiman, M. E Allen, K. V. Thompson, S. Lumpkin, & H. Harris (Eds.), *Wild mammals in captivity: Principles and techniques* (pp. 317-333). Chicago: The University of Chicago Press.

Connor, R. C., Read, A. J., & Wrangham, R. W. (2000a). Male reproductive strategies and social alliances. In J. Mann, R. C. Connor, P. L. Tyack, & H. Whitehead (Eds.), *Cetacean societies: Field studies of dolphins and whales* (pp. 247-269). Chicago: The University of Chicago Press.

Connor, R. C., Wells, R., Mann, J., & Read, A. (2000b). The bottlenose dolphin: Social relationships in a fission–fusion society. In J. Mann, R. C. Connor, P. L. Tyack, & H. Whitehead (Eds.), *Cetacean societies: Field studies of dolphins and whales* (pp. 91-126). Chicago: The University of Chicago Press.

Corkeron, P. (2002). In W. F. Perrin, B. Würsig, & J. G. M. Thewissen (Eds.), *Encyclopedia of marine mammals* (pp. 192-197). London: Academic Press.

Cranford, T. W., Van Bonn, W. G., Chaplin, M. S., Carr, J. A., Kamolnick, T. A., Donald, A. C., & Ridgway, S. H. (1997). Visualizing dolphin sonar signal generation using high-speed video endoscopy. *The Journal of the Acoustical Society of America*, *102*(5), 3123.

Culik, B. (2003). *Convention of migratory species. Review on small cetaceans: Distribution, behaviour, migration and threats.* Available online: www.cms.int/reports/small_cetaceans/index.htm. Accessed: 7 July 2005.

Dahlheim, M. E., & Heyning, J. E. (1999). Killer whale, *Orcinus orca* (Linnaeus, 1758). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises* (pp. 281-322). London: Academic Press.

Defran, R. H., & Pryor, K. (1980). The behavior and training of cetaceans in captivity. In L. M. Herman (Ed.), *Cetacean behavior: Mechanisms and functions* (pp. 319-358). Malabar, FL: University of Hawaii, Krieger Publishing Company.

Degollada, E., Garcia Hartmann, M., & Cranford, T. (1998). Histological structure of the sound generation complex in delphinoid cetaceans. *Proceedings of the World Marine Mammal Science Conference*, Monaco.

Evans, P. G. (1987). *The natural history of whales and dolphins.* New York: Facts on File. 343 pp.

Gannon D. P., Barros, N. B., Nowacek, D. P., Read, A. J., Waples, D. M., & Wells, R. S. (2005). Prey detection by bottlenose dolphins, *Tursiops truncatus*: An experimental test of the passive listening hypothesis. *Animal Behaviour*, *69*, 709-720.

George, J. C., Bada, J., Zeh, J., Scott, L., Brown, S. E., O'Hara, T., & Suydam, R. (1999). Age and growth estimates of bowhead whales (*Balaena mysticetus*) via aspartic acid racemization. *Canadian Journal of Zoology/Revue Canadienne de Zoologie*, *77*(4), 571-580.

Geraci, J. R. (1986a). Marine mammals: Introduction and identification. In M. E. Fowler (Ed.), *Zoo & wild animal medicine* (pp. 750-756). Philadelphia: W. B. Saunders Company.

Geraci, J. R. (1986b). Marine mammals: Husbandry. In M. E. Fowler (Ed.), *Zoo & wild animal medicine* (pp. 757-760). Philadelphia: W. B. Saunders Company.

Gygax, L. (1993). Spatial movement patterns and behaviour of two captive bottlenose dolphins (*Tursiops truncatus*): Absence of stereotyped behaviour or lack of definition? *Applied Animal Behaviour Science*, *38*, 337-344.

Herman, L. M. (1980). Cognitive characteristics of dolphins. In L. M. Herman (Ed.), *Cetacean behavior: Mechanisms and functions* (pp. 363-429). Malabar, FL: University of Hawaii, Krieger Publishing Company.

Herman, L. M. (1986). Cognition and language competencies of bottlenose dolphins. In R. J. Schusterman, J. A. Thomas, & F. G. Wood (Eds.), *Dolphin cognition and behaviour: A comparative approach* (pp. 221-252). Hillsdale, NJ: Lawrence Erlbaum Associates.

Herman, L. M. (1989). What the dolphin knows, or might know, in its natural world. In K. Pryor & K. S. Norris (Eds.), *Dolphin societies* (pp. 348-364). Berkeley: University of California Press.

Herman, L. M., & Tavolga, W. N. (1980). The communication systems of cetaceans. In L. M. Herman (Ed.), *Cetacean behavior: Mechanisms and functions* (pp. 149-209). Malabar, FL: University of Hawaii, Krieger Publishing Company.

Hooker, S. K., & Baird, R. W. (1999). Deep-diving behaviour of the northern bottlenose whale, *Hyperoodon* *ampullatus* (Cetacea: Ziiphidae). *Proceedings of the Royal Society of London B*, *266*, 671-676.

Jefferson, T. A., & Van Waerebeek, K. (2002). The taxonomic status of the nominal dolphin species *Delphinus tropicalis*, Van Bree, 1971. *Marine Mammal Science*, *18*(4), 787-818.

Johnson, C. M., & Norris, K. S. (1986). Delphinid social organization and social behavior. In R. J. Schusterman, J. A. Thomas, & F. G. Wood (Eds.), *Dolphin cognition and behaviour: A comparative approach* (pp. 335-346). Hillsdale, NJ: Lawrence Erlbaum Associates.

Jones, G. J., & Sayigh, L. S. (2002). Geographic variation in rates of vocal production of free-ranging bottlenose dolphins. *Marine Mammal Science*, *18*, 374-393.

Kastelein, R. A., Staal, C., & Wiepkema, P. R. (2003). Food consumption, food passage time, and body measurements of captive Atlantic bottlenose dolphins (*Tursiops truncatus*). *Aquatic Mammals*, *29*(1), 53-66.

Ketten, D. R. (1998). *Marine mammal auditory systems: A summary of audiometric and anatomical data and its implications for underwater acoustic impacts* (NOAA-TM-NMFS-SWFSC-256). Washington, DC: U.S. Department of Commerce, National Oceanic and Atmospheric Administration, National Marine Fisheries Services, Southwest Fisheries Science Center. 97 pp.

Klinowska, M. (1991). *Dolphins, porpoises and whales of the world: The IUCN red data book*. Gland, Switzerland and Cambridge, UK: IUCN. 429 pp.

Klinowska, M., & Brown, S. (1986). *A review of dolphinaria.* London: Department of the Environment. 247 pp.

Leatherwood, S., & Reeves, R. R. (1983). *The Sierra Club handbook of whales and dolphins.* San Francisco: Sierra Club Books. 309 pp.

Lyamin, O. I., Shpak, O. V., Nazarenko, E. A., & Mukhametov, L. M. (1998). Behavioral signs of paradoxical sleep in the beluga whale. *Journal of Sleep Research*, *7*, 166.

Lyamin, O. I., Manger, P. R., Mukhametov, L. M., Siegel, J. M., & Shpak, O. V. (2000). Rest and activity states in a gray whale. *Journal of Sleep Research*, *9*, 261-267.

Lyamin, O. I., Mukhametov, L. M., Siegel, J. M., Nazarenko, E. A., Polyakova, I. G., & Shpak, O. V. (2002). Unihemispheric slow wave sleep and the state of the eyes in a white whale. *Behavioral Brain Research*, *129*, 125-129.

Mann, J., Connor, R. C., Tyack, P. L., & Whitehead, H. (Eds.). (2000). *Cetacean societies: Field studies of dolphins and whales.* Chicago: The University of Chicago Press. 448 pp.

Mukhametov, L. M. (1987). Unihemispheric slow-wave sleep in the Amazonian dolphin, *Inia geoffrensis. Neuroscience Letters*, *79*, 128-132.

Mukhametov, L. M (1988). The absence of paradoxical sleep in dolphins. In W. P. Koella, F. Obal, H. Schulz, & P. Visser (Eds.), *Sleep 1986* (pp. 154-156). New York: Gustav Fischer Verlag.

Mukhametov, L. M (1995). Paradoxical sleep peculiarities in aquatic mammals. *Sleep Research*, *24A*, 202.

Mukhametov, L. M., & Lyamin, O. I. (1994). Rest and active states in bottlenose dolphins (*Tursiops truncatus*). *Journal of Sleep Research*, *3*, 174.

Mukhametov, L. M., Supin, A. Y., & Polyakova, I. G. (1977). Interhemispheric asymmetry of the electroencephalographic sleep patterns in dolphins. *Brain Research*, *134*, 581-584.

Nachtigall, P. E. (1986). Vision, audition and chemoreception in dolphins and other marine mammals. In R. J. Schusterman, J. A. Thomas, & F. G. Wood (Eds.), *Dolphin cognition and behaviour: A comparative approach* (pp. 79-113). Hillsdale, NJ: Lawrence Erlbaum Associates.

Norris, K. S., & Dohl, T. P. (1980). The structure and functions of cetacean schools. In L. M. Herman (Ed.), *Cetacean behavior: Mechanisms and functions* (pp. 211-261). Malabar, FL: University of Hawaii, Krieger Publishing Company.

Odell, D. K., & McClune, K. M. (1999). False killer whale, *Pseudorca crassidens* (Owen, 1846). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises* (pp. 213-244). London: Academic Press.

Östman, J. (1991). Changes in aggressive and sexual behavior between two male bottlenose dolphins (*Tursiops truncatus*) in a captive colony. In K. J. Pryor & K. S. Norris (Eds.), *Dolphin societies* (pp. 305-316). Berkeley: University of California Press.

Perrin, W. F., Würsig, B., & Thewissen, J. G. M. (Eds.). (2002). *Encyclopedia of marine mammals.* London: Academic Press. 1,414 pp.

Potter, J. R., Taylor, E., & Chitre, M. (1997). Could marine mammals use ambient noise imaging techniques? *Proceedings of the 134th Acoustic Society of America (ASA) Meeting*, San Diego.

Pryor, K. J. (1990). Non-acoustic communication in small cetaceans: Glance, touch, position, gesture, and bubbles. In J. A. Thomas & R. A. Kastelein (Eds.), *Sensory abilities of cetaceans: Laboratory and field evidence* (NATO ASI Life Sciences Series, Vol. 191) (pp. 537-544). New York: Plenum Press.

Pryor, K. J. (1995). Behavior and learning in porpoises and whales. In *On behavior* (pp. 5-27). North Bend, WA: Sunshine Books.

Read, A. J. (1999). Harbour porpoise, *Phocoena phocoena* (Linnaeus, 1758). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises* (pp. 323-356). London: Academic Press.

Reddy, M., Kamolnick, T., Curry, C., Skaar, D., & Ridgway, S. (1994). Energy requirements for the bottlenose dolphin (*Tursiops truncatus*) in relation to sex, age and reproductive status. *Marine Mammals: Public Display and Research*, *1*, 26-31.

Reeves, R. R., Twiss, J. R., Jr., Twiss, J. R., & Montgomery, S. (Eds.). (1999). *Conservation and management of marine mammals.* Washington, DC: Smithsonian Books. 471 pp.

Reynolds, J. E., III, & Rommel, S. A. (Eds.). (1999). *Biology of marine mammals.* Washington, DC: Smithsonian Books. 578 pp.

Rice, D. W. (1989). Sperm whale, *Physeter macrocephalus* (Linnaeus, 1758). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 4: River dolphins and the larger toothed whales* (pp. 177-234). London: Academic Press.

Rice, D. W. (1998). *Marine mammals of the world: Systematic and distribution* (Society of Marine Mammalogy Special Publication Number 4). 231 pp.

Richardson, W. J., Greene, C. R., Malme, C. I., & Thomson, D. H. (1995). *Marine mammals and noise.* London: Academic Press. 567 pp.

Ridgway, S. H. (1986). Physiological observations on dolphin brains. In R. J. Schusterman, J. A. Thomas, & F. G. Wood (Eds.), *Dolphin cognition and behaviour: A comparative approach* (pp. 31-59). Hillsdale, NJ: Lawrence Erlbaum Associates.

Ridgway, S. H. (1995). The tides of change: Conservation of marine mammals. In E. F. Gibbons, Jr., B. S. Durrant, & J. Demarest (Eds.), *Conservation of endangered species in captivity: An interdisciplinary approach* (pp. 407-424). Albany: State University of New York Press.

Ridgway, S. H. (2002). Asymmetry and symmetry in brain waves from dolphin left and right hemispheres: Some observations after anesthesia, during quiescent hanging behavior, and during visual obstruction. *Brain, Behavior, and Evolution*, *60*, 265-274.

Ridgway, S. H., & Carder, D. A. (1990). Tactile sensitivity, somatosensory responses, skin vibrations, and the skin surface ridges of the bottlenose dolphin, *Tursiops truncatus*. In J. A. Thomas & R. A. Kastelein (Eds.), *Sensory abilities of cetaceans, laboratory and field evidence* (NATO ASI Life Sciences Series, Vol. 191) (pp. 163-179). New York: Plenum Press.

Ridgway, S. H., & Harrison R. J. (Eds.). (1989). *Handbook of marine mammals. Vol. 4: River dolphins and the larger toothed whales.* London: Academic Press. 442 pp.

Ridgway, S. H., & Harrison R. J. (Eds.). (1994). *Handbook of marine mammals. Vol. 5: The first book of dolphins and porpoises.* London: Academic Press. 416 pp.

Ridgway, S. H., & Harrison R. J. (Eds.). (1999). *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises.* London: Academic Press. 486 pp.

Rohr, J. J., Fish, F. E., & Gilpatrick, J. W., Jr. (2002). Maximum swim speeds of captive and free-ranging delphinids: Critical analysis of extraordinary performance. *Marine Mammal Science*, *18*(1), 1-19.

Samuels, A., & Gifford, T. (1997). A quantitative assessment of dominance relations among bottlenose dolphins. *Marine Mammal Science*, *13*(1), 70-99.

Schroeder, J. P. (1995). Marine mammals reproductive physiology. In E. F. Gibbons, Jr., B. S. Durrant, & J. Demarest (Eds.), *Conservation of endangered species in captivity: An interdisciplinary approach* (pp. 425-440). Albany: State University of New York Press.

Sekiguchi, Y., & Koshima, S. (2003). Resting behaviors of captive bottlenose dolphins (*Tursiops truncatus*). *Physiological Behavior*, *79*, 643-653.

Stewart, B. A. (Ed.). (2001). Rescue, rehabilitation, and scientific studies of a California gray whale calf. *Aquatic Mammals*, *27*(3).

Stoskopf, M. K., & Gibbons, E. F. (1994). Quantitative evaluation of the effects of environmental parameters on the physiology, behavior, and health of animals in naturalistic captive environments. In E. F. Gibbons, E. J. Wyers, E. Waters, & E. W. Menzel (Eds.), *Naturalistic environments in captivity for animal behavior research* (pp. 140-160). Albany: State University of New York Press.

Sweeney J. C. (1990). Marine mammal behavioral diagnostics. In L. Dierauf (Ed.), *CRC handbook of marine mammal medicine: Health, disease, and rehabilitation* (pp. 53-72). Boston: CRC Press.

Tavolga, M. C. (1966). Behavior of the bottlenose dolphin (*Tursiops truncatus*): Social interactions in a captive colony. In K. S. Norris (Ed.), *Whales, dolphins, and porpoises* (pp. 718-730). Berkeley: University of California Press.

Taylor, E., Potter, J. R., & Chitre, M. (1997). Ambient noise imaging potential of marine mammals. *Proceedings of the Underwater Bio-Sonar and Bioacoustics Symposium*, Loughborough.

van der Toorn, J. D. (1987). A biological approach to dolphinarium water purification: I. Theoretical aspects. *Aquatic Mammals*, *13*(3), 83-92.

Wells, R. S., & Scott, M. D. (1999). Bottlenose dolphin, *Tursiops truncatus* (Montagu, 1821). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises* (pp. 137-182). London: Academic Press.

Literature References for the Cetacean Species Information Table from the *Handbook of Marine Mammals*

Listed by species' scientific names in alphabetic order:

Goodall, R. N. P. (1994). Commerson's dolphin, ***Cephalorhyncus commersonii*** (Lacépède 1804). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 5: The first book of dolphins and porpoises* (pp. 241-268). London: Academic Press.

Slooten, E., & Dawson, S. M. (1994). Hector's dolphin, ***Cephalorhyncus hectori*** (van Beneden, 1881). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 5: The first book of dolphins and porpoises* (pp. 311-334). London: Academic Press.

Brodie, P. F. (1989). The white whale, ***Delphinapterus leucas*** (Pallas, 1776). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 4: River dolphins and the larger toothed whales* (pp. 119-144). London: Academic Press.

Evans, W. E. (1994). Common dolphin, white-bellied porpoise, ***Delphinus delphis*** (Linnaeus, 1758). In S. H.

Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 5: The first book of dolphins and porpoises* (pp. 191-224). London: Academic Press.

Ross, G. J. B., & Leatherwood S. (1994). Pygmy killer whale, ***Feresa attenuata*** (Gray, 1956). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 5: The first book of dolphins and porpoises* (pp. 387-404). London: Academic Press.

Bernard, H. J., & Reily, S. B. (1999). Pilot whales, ***Globicephala*** (Lesson, 1828). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises* (pp. 245-280). London: Academic Press.

Kruse, S., Caldwell, D. K., & Caldwell, M. C. (1999). Risso's dolphin, ***Grampus griseus*** (G. Cuvier, 1812). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises* (pp. 183-212). London: Academic Press.

Best, R. C., & Da Silva, V. M. F. (1989). Amazon river dolphin, boto, ***Inia geoffrensis*** (de Blainville, 1817). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 4: River dolphins and the larger toothed whales* (pp. 1-24). London: Academic Press.

Caldwell, D. K., & Caldwell, M. C. (1989). Pygmy sperm whale, ***Kogia breviceps*** (de Blainville, 1838), dwarf sperm whale, ***Kogia simus*** (Owen, 1866). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 4: River dolphins and the larger toothed whales* (pp. 235-260). London: Academic Press.

Perrin, W. F., Leatherwood, S., & Collet, A. (1994). Fraser's dolphin, ***Lagenodelphis hosei*** (Fraser, 1956). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 5: The first book of dolphins and porpoises* (pp. 225-240). London: Academic Press.

Reeves, R. R., Smeenk, C., Brownwell, R. L., Jr., & Kinze C. C. (1999). Atlantic white-sided dolphin, ***Lagenorhynchus acutus*** (Gray, 1828). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises* (pp. 31-56). London: Academic Press.

Reeves, R. R., Smeenk, C., Brownwell, R. L., Jr., & Kinze C. C. (1999). Whitebeaked dolphin, ***Lagenorhynchus albirostris*** (Gray, 1846). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises* (pp. 1-30). London: Academic Press.

Brownell, R. L., Jr., Walker, W. A., & Forney, K. A. (1999). Pacific white-sided dolphin, ***Lagenorhynchus obliquidens*** (Gill, 1865). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises* (pp. 57-84). London: Academic Press.

Brownell, R. L., Jr., & Cipriano, F. (1999). Dusky dolphin, ***Lagenorhynchus obscurus*** (Gray, 1828). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises* (pp. 85-104). London: Academic Press.

Chen Peixun. (1989). Baiji, ***Lipotes vexillifer*** (Miller, 1918). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 4: River dolphins and the larger toothed whales* (pp. 25-44). London: Academic Press.

Jefferson, T. A., Newcomer, M. W., Leatherwood, S., & Van Waerebeek, K. (1994). Right whale dolphins, ***Lissodelphis borealis*** (Peale, 1848). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 5: The first book of dolphins and porpoises* (pp. 335-362). London: Academic Press.

Mead, J. G. (1989). Beaked whales of the genus **Mesoplodon**. In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 4: River dolphins and the larger toothed whales* (pp. 349-430). London: Academic Press.

Hay, K. A., & Mansfield, A. W. (1989). Narwhal, ***Monodon monoceros*** (Linnaeus, 1758). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 4: River dolphins and the larger toothed whales* (pp. 145-176). London: Academic Press.

Kasuya, K. (1999). Finless porpoise, ***Neophocaena phocaenoides*** (G. Cuvier, 1829). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises* (pp. 411-442). London: Academic Press.

Marsh, H., Lloze, R., Heinsohn, G. E., & Kasuya, T. (1989). Irrawaddy dolphin, ***Orcaella brevirostris*** (Gray, 1866). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 4: River dolphins and the larger toothed whales* (pp. 101-118). London: Academic Press.

Dahlheim, M. E., & Heyning, J. E. (1999). Killer whale, ***Orcinus orca*** (Linnaeus, 1758). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises* (pp. 281-322). London: Academic Press.

Perryman, W. L., Au, D. W. K., Leatherwood, S., & Van Waerebeek, K. (1994). Melon-headed whale, ***Peponocephala electra*** (Gray 1846). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 5: The first book of dolphins and porpoises* (pp. 363-368). London: Academic Press.

Read, A. J. (1999). Harbour porpoise, ***Phocoena phocoena*** (Linnaeus, 1758). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises* (pp. 323-356). London: Academic Press.

Vidal, O., Brownell, R. L., Jr., & Findley, L. T. (1999). Vaquita, ***Phocoena sinus*** (Norris & McFarland, 1958). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises* (pp. 357-378). London: Academic Press.

Brownell, R. L., Jr., & Clapham, P. J. (1999). Burmeister's porpoise, ***Phocoena spinipinnis*** (Burmeister, 1865). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises* (pp. 393-410). London: Academic Press.

Houck, W. J., & Jefferson, T. A. (1999). Dall's porpoise, ***Phocoenoides dalli*** (True, 1885). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises* (pp. 443-472). London: Academic Press.

Reeves, R. R., & Brownell, R. L., Jr. (1989). Susu, ***Platanista gangetica*** (Roxburgh, 1801) and ***Platanista minor*** (Owen, 1853). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 4: River dolphins and the larger toothed whales* (pp. 69-100). London: Academic Press.

Brownell, R. L., Jr. (1989). Franciscana, ***Pontoporia blainvillei*** (Gervais & d'Orbigny, 1844). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 4: River dolphins and the larger toothed whales* (pp. 45-68). London: Academic Press.

Rice, D. W. (1989). Sperm whale, ***Physeter macrocephalus*** (Linnaeus, 1758). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 4: River dolphins and the larger toothed whales* (pp. 177-234). London: Academic Press.

Odell, D. K., & McClune, K. M. (1999). False killer whale, ***Pseudorca crassidens*** (Owen, 1846). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises* (pp. 213-244). London: Academic Press.

Da Silva, V. M. F., & Best, R. C. (1994). Tucuxi, ***Sotalia fluviatilis*** (Gervais, 1853). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 5: The first book of dolphins and porpoises* (pp. 43-70). London: Academic Press.

Ross, G. J. B., Heinsohn, G. E., & Cockroft, V. G. (1994). Humpback dolphins, ***Sousa chinensis*** (Osbeck, 1765), ***Sousa plumbea*** (G. Cuvier, 1829) and ***Sousa teuszii*** (Kükenthal, 1892). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 5: The first book of dolphins and porpoises* (pp. 23-42). London: Academic Press.

Perrin, W. F., & Hohn, A. A. (1994). Pantropical spotted dolphin, ***Stenella attenuata***. In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 5: The first book of dolphins and porpoises* (pp. 71-98). London: Academic Press.

Perrin, W. F., & Mead, J. G. (1994). Clymene dolphin, ***Stenella clymene*** (Gray, 1846). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 5: The first book of dolphins and porpoises* (pp. 161-172). London: Academic Press.

Perrin, W. F., Wilson, C. E., & Archer, F. I., II. (1994). Striped dolphin, ***Stenella coeruleoalba*** (Meyen, 1833). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 5: The first book of dolphins and porpoises* (pp. 129-160). London: Academic Press.

Perrin, W. F., Caldwell, D. K., & Caldwell, M. C. (1994). Atlantic spotted dolphin, ***Stenella frontalis*** (G. Cuvier, 1829). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 5: The first book of dolphins and porpoises* (pp. 173-190). London: Academic Press.

Perrin, W. F., & Gilpatrick, J. W., Jr. (1994). Spinner dolphin, ***Stenella longirostris*** (Gray, 1828). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 5: The first book of dolphins and porpoises* (pp. 99-128). London: Academic Press.

Miyazaki, N., & Perrin, W. F. (1994). Rough-toothed dolphin, ***Steno bredanensis*** (Lesson, 1828). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 5: The first book of dolphins and porpoises* (pp. 1-22). London: Academic Press.

Wells, R. S., & Scott, M. D. (1999). Bottlenose dolphin, ***Tursiops truncatus*** (Montagu, 1821). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises* (pp. 137-182). London: Academic Press.

Literature References for Hybrids

Antrim, J. E., & Cornell, L. H. (1981). *Globicephala-Tursiops* hybrid. *Abstracts of the Fourth Biennial Conference on the Biology of Marine Mammals*, San Francisco.

Dohl, T. P., Norris, K. S., & Kang, I. (1974). A porpoise hybrid, *Tursiops* × *Steno*. *Journal of Mammalogy*, *55*, 217-222.

Nishiwaki, M., & Tobayama, T. (1982). Morphological study on the hybrid between *Tursiops* and *Pseudorca*. *Scientific Reports Whales Research Institute*, *34*, 109-121.

Sylvestre, J. P., & Tasaka, S. (1985). On the intergeneric hybrids in cetaceans. *Aquatic Mammals*, *11*(3), 101-108.

Zornetzer, H. R., & Duffield, D. A. (2003). Captive-born bottlenose dolphin × common dolphin (*Tursiops truncatus* × *Delphinus capensis*) intergeneric hybrids. *Canadian Journal of Zoology/Revue Canadienne de Zoologie*, *81*(10), 1755-1762.

Aquatic Mammals 2005, *31*(3), 311-319, DOI 10.1578/AM.31.3.2005.311

3. Survey of International Cetacean Facilities

In 1996, the first survey of international cetacean facilities was launched to assess the status of existing facilities worldwide and gather information on controlled environments to serve as a base for this guidebook. A 30-page questionnaire was designed to request detailed information on location, function of facility and staff, number of animals kept and species, architectural design of pools and enclosures, life support systems, husbandry practices, diet, and food storage and preparation. The survey attempted to understand successful design features and husbandry practices, as well as recurrent problems. The survey was sent by post with a letter of support by cetacean scientist, Mrs. Karen Pryor, to 157 facilities in 41 countries (9 facilities were not included because of no available contact information). Forty-four questionnaires from 22 countries were received during the following year—early 1997. This chapter presents the survey results. Although the main goal of this survey was to receive information on the above-mentioned topics, the statistical analysis gives a broader picture of cetacean facilities in terms of their similarities and differences, and it allows us to begin to understand the influence of geographical locations, culture, and types of environments on the creation of controlled environments for cetaceans. The survey resulted in the largest database to date on international cetacean facilities. In addition to the survey, the project team visited 26 institutions in Europe, America, and Asia.

The Questionnaire

To prepare the survey, an extensive list of cetacean institutions was compiled based on the directories of the European Association for Aquatic Mammals (EAAM), the International Marine Animal Trainer Association (IMATA), and the International Zoo Yearbook (Olney & Ellis, 1993; Anonymous, 1996, 2005), personal contacts in Japan and the United States, and through the marine mammals Internet mailing list—MarMam (Anonymous, 2005c)—and the World Wide Web.

The Questionnaire Consisted of Five Specific Aspects of a Cetacean Facility

I. General Information—This included questions on the function of the facility (e.g., display, research, rescue, etc.) and its geographical environment, the number and species of animals kept, the number and designation of the staff attached to the cetacean section, the official regulations governing the facility, and research programmes conducted.

II. Facilities for Cetaceans—This section dealt with the design, dimensions, and volumes of the pools and enclosures; construction materials; quarantine areas; gates and channels; underwater windows; drains; lighting; acoustic quality; husbandry and training features; water features; built-in and installed equipment; levels and access; protection of the animals against harassment; cost of construction; and questions pertaining to indoor environments such as ventilation, lighting, temperature, and acoustic quality.

III. Husbandry—This section collected information on husbandry tests and records to assess the influence of the environment on the animals' health and behaviour. It also determined the policies guiding species display and breeding, and the facilities' involvement in the conservation, protection, and rehabilitation of stranded animals and endangered species. Information dealing with animal transportation within the facility and the laboratory equipment were also requested.

IV. Life Support Systems—This included detailed questions on the water system and quality, temperature control, filtration system, cleaning methods, drainage, construction, and running cost of the system.

V. Fish House—This was a section on the animals' diet, food storage and preparation, and the design and construction materials of the fish kitchen.

The questionnaire was designed with multiple-choice tick boxes, and it requested and encouraged facilities to highlight problems and recurrent deficiencies, as well as successful designs and environmental enrichment features.

Geographical Distribution of Facilities

At the time of the 1997 survey, the cetacean facilities database showed 166 research and public display institutions that house dolphins and whales in 42 countries on all inhabited continents. It did not include rescue and rehabilitation facilities, and it counted very few research or military facilities. Today, some firms in India, Vietnam, Turkey, Saudi Arabia, Egypt, United Arab Emirates, Dubai, and Morocco are trying to open new facilities. Many institutions in Central America and the Caribbean Islands have applied for import permits, mainly to develop Swim-With-The-Dolphin (SWTD).

The greatest progression of new facilities was in Mexico. There are possibly many more facilities in China, Russia, and other former Soviet Union republics where military facilities were numerous and, when they closed down, they sold their animals to display facilities. There are also several traveling shows in Chile, Colombia, Indonesia, and probably other South American and Asian countries.

The following map (Figure 3.1) shows the recorded facilities in 1997. The map clearly shows that a large majority of institutions are located in the Northern hemisphere, in warm and temperate climates between latitude 20º N—just below the Tropic of Cancer—and 60º N. They are mainly coastal. The two countries with the longest history of exhibiting cetaceans also have the largest number of facilities: the United States and Japan. Each have 39 display and research institutions.

Results

Forty-four questionnaires were received over a period of 12 months from 22 countries, representing 28% of the questionnaires sent.

Participation from Asia was better than that from North America, despite the language barrier. Participation from the Pacific region was good, and countries in Europe were fairly well represented. This gives a wide geographical representation and, thus, enables comparisons of a variety of facilities across a broad range of geographical locations.

This survey helped to identify and understand various elements essential to the dolphins' welfare in their controlled environments. It enabled the study of existing facilities around the world, which provided new insights into previously unknown facilities in countries where language, communication, development, or economical factors often prevent them from participating in the active information exchange of the international marine mammal professional community.

The main results of the study are given in the following section. They are all based on the statistical analysis of the survey data, except the section on cetacean species in captivity, which includes a comparison between the survey results and the entire database.

Temporal Distribution of Cetacean Facilities

The temporal distribution of sampled facilities was examined to determine if the sample of surveyed facilities was representative in construction date and age and, therefore, would produce accurate information on questions related to design trends, construction techniques, and so on.

Construction dates of sampled facilities (n = 41) ranged from the early 1950s to the late 1990s (Figure 3.2). Construction was present in every decade between 1950 and 2000; however, all decades were not represented equally (Kruskal Wallis, $H = 37.976$, $p < 0.0001$). Many sampled facilities were built in the late 1960s and in the 1990s. The late 1960s period corresponded to the first surge of interest in dolphins—at the time of the *Flipper* TV series in the USA and Europe. The 1990s period witnessed a second surge of

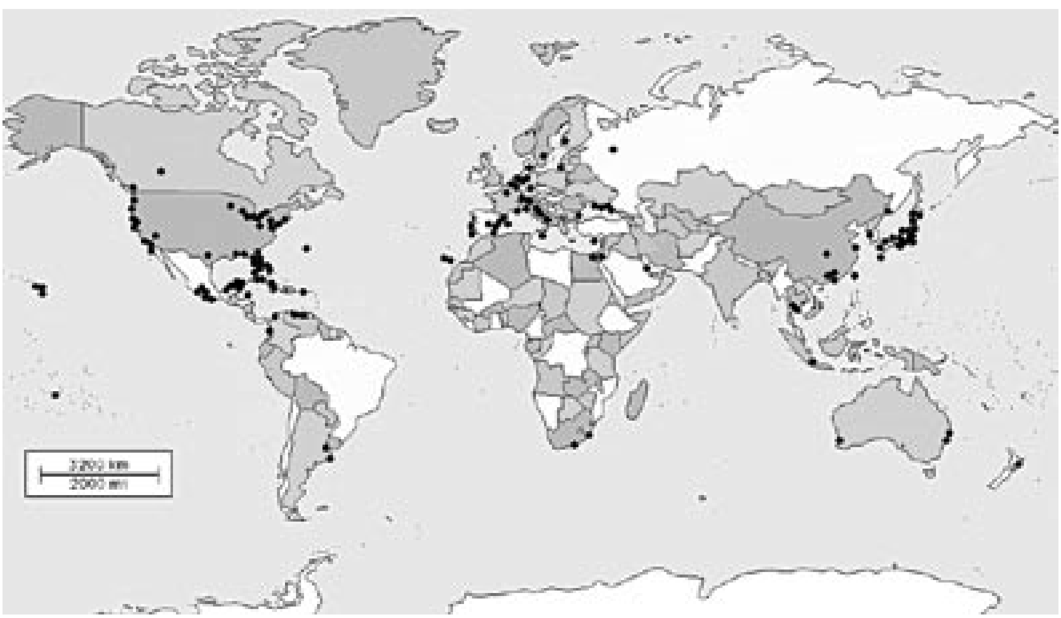

Figure 3.1. World map of cetacean facilities in 1997 (indicated by a black dot)

interest in dolphins and whales due to dolphin therapy, whale watching, and so on. This period saw the creation of numerous SWTD facilities, mainly in Central America, the Caribbean Islands, and Southern Europe, as well as new display facilities in developing countries—especially in Asia.

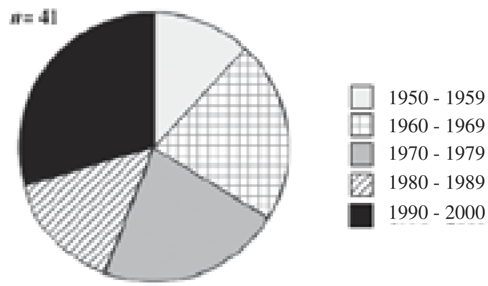

Figure 3.2. Temporal distribution of cetacean facilities' construction dates

The temporal distribution of sampled facilities gives a good representation of old and new facilities, covering a wide variety of types and designs; therefore, the results give insight into problems associated with the age of the facility. It is also possible to trace the evolution of trends in the design of cetacean environments.

Number of Cetaceans per Facilities

Previous censuses did not determine the average number of cetaceans per facility (Cornell & Asper, 1978; Defran & Pryor, 1980; Collet, 1984; Asper et al., 1988, 1990; Duffield & Wells, 1990; Duffield & Shell, 1994, 1996; Nakahara & Takemura, 1997). Determining the average number of animals per facility is relevant to understanding the profile of the institutions, the possible impact on the social composition of the group, and the impact on maintenance and financial issues.

The median number of cetaceans per facility is 7 (maximum = 39). Among sampled facilities (n = 42), 26% had < 5 animals and 45% of all sampled facilities had between 5 and 10 animals (Figure 3.3).

It is likely that the high maintenance and operational costs associated with keeping a large number of animals dictates the extent of the collection. A very large number of animals might not increase the number of visitors substantially. Five facilities have only two animals, which is considered the absolute minimum group size since keeping single animals should be avoided. It should be highlighted that three of these five facilities are research institutions and, therefore, might have either financial or scientific reasons for keeping such a small number of animals.

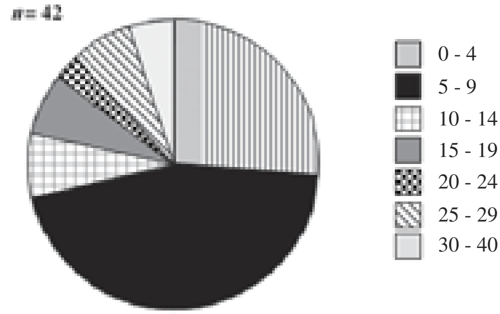

Figure 3.3. Number of cetaceans per facility

Cetacean Species in Captivity

The facilities database enables a census of cetacean species in captivity. At the time of the survey, 21 species—all odontocetes—were kept in human care. This represents approximately 30% of the 72 species of odontocetes. Among the 21 species, 6 species were kept in only one facility. Results show that the bottlenose dolphin, the Pacific white-sided dolphin, the beluga whale, the killer whale, and the false killer whale are the species most commonly kept in captivity, which is in accordance with the results of the regularly updated North American census (Cornell & Asper, 1978; Asper et al., 1988, 1990; Duffield & Shell, 1994, 1996). Table 3.1 shows the number of facilities displaying each species of cetaceans as of 1997. Data displayed in this table are not limited to surveyed facilities but are extracted from the whole database. Information on species was available for 144 of the 166 facilities (87%) in the database.

The bottlenose dolphin is the most commonly displayed species on all continents. This species is displayed in 89% of the facilities, with or without other species.

The species variety was compared between Asian, North American, and European facilities from the data available in the facilities database (Figure 3.4).

In 1997, Europe had 41 facilities in 17 countries, and data on the species were available for 35 of those. European institutions displayed a total of 7 species. The median number of species per facility was 1 (maximum = 4). Results showed that 83% of the facilities only display 1 species, and 94% displayed the bottlenose dolphin as a single species or with other species.

North America had 44 facilities in four countries (The Bahamas and Bermuda were included), and data on the species were available for all of them. Results showed that North American institutions display a total of 12 species, which is in general concordance with the 11 species in the latest update of the North American census, albeit

Table 3.1. Number of facilities displaying cetaceans by species

Cetacean species	Number of facilities
Baiji (*Lipotes vexillifer*) *	1
Beluga whale (*Delphinapterus leucas*)	17
Boto (*Inia geoffrensis*)	3
Bottlenose dolphin (*Tursiops truncatus*)	128
Commerson's dolphin (*Cephalorhynchus commersonii*)	7
Common dolphin (*Delphinus delphis*)	2
False killer whale (*Pseudorca crassidens*)	16
Finless porpoise (*Neophocaena phocaenoides*)	9
Franciscana (*Pontoporia blainvillei*) **	1
Harbour porpoise (*Phocoena phocoena*)	3
Pacific humpback dolphin (*Sousa chinensis*)	3
Irrawaddy dolphin (*Orcaella brevirostris*)	3
Killer whale (*Orcinus orca*)	16
Melon-headed whale (*Peponocephala electra*)***	1
Pacific white-sided dolphin (*Lagenorhynchus obliquidens*)	25
Pantropical spotted dolphin (*Stenella attenuata*)	1
Risso's dolphin (*Grampus griseus*)	9
Rough-toothed dolphin (*Steno bredanensis*)	2
Short-finned pilot whale (*Globicephala macrorhynchus*)	2
Striped dolphin (*Stenella coeruleoalba*)	1
Tucuxi (*Sotalia fluviatilis*)	1

*The only Baiji kept for conservation purposes died in July 2002.

**The only Franciscana died in September 1999.

***This originally rescued animal died in 2003.

with 2 different/additional species (Duffield & Shell, 1996). The median number of species per facility was 1 (maximum = 5). Sixty-one percent of the facilities displayed only 1 species, and 91% displayed the bottlenose dolphin as a single species or with other species.

Asia had 50 facilities in 7 countries, and data on the species were available for 42 of them. Asian institutions displayed a total of 14 species. The median number of species per facility was 2 (maximum = 8). Thirty-two percent of the facilities displayed only 1 species, and 81% displayed the bottlenose dolphin as a single species or with other species.

Sampled Facilities (Data from Survey Only)

In sampled facilities, 15 of the 21 species (71%) kept in human care were represented.

Sampled European facilities (*n* = 17) housed 129 cetaceans, of which 126 (98%) were bottlenose dolphins. Sampled Asian facilities (*n* = 11) house 162 animals, and among them 95 (59%) were bottlenose dolphins. Data from North America and other continents were not analysed since the sample was too small and would not be representative. From the statistical analysis of the database of facilities, North America seems to have results consistent with Europe and Asia. The latest update of the North American marine mammal census showed that out of 454 cetaceans, 341 (75%) were bottlenose dolphins (Duffield & Shell, 1996).

The variety of species displayed was compared between European and Asian facilities (Figure 3.5). All sampled facilities were assigned an identification (ID) number, which corresponded to the order in which the completed questionnaires were received.

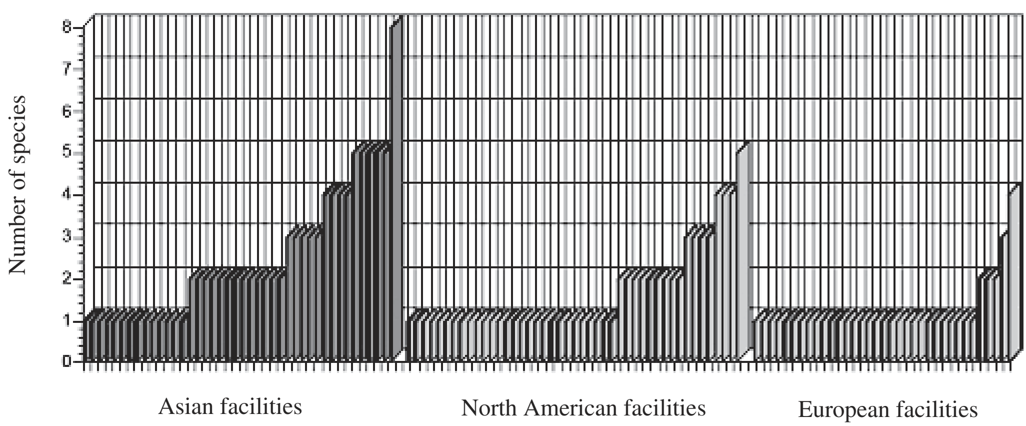

Figure 3.4. Comparison of the captive cetacean species among Asian, North American, and European facilities

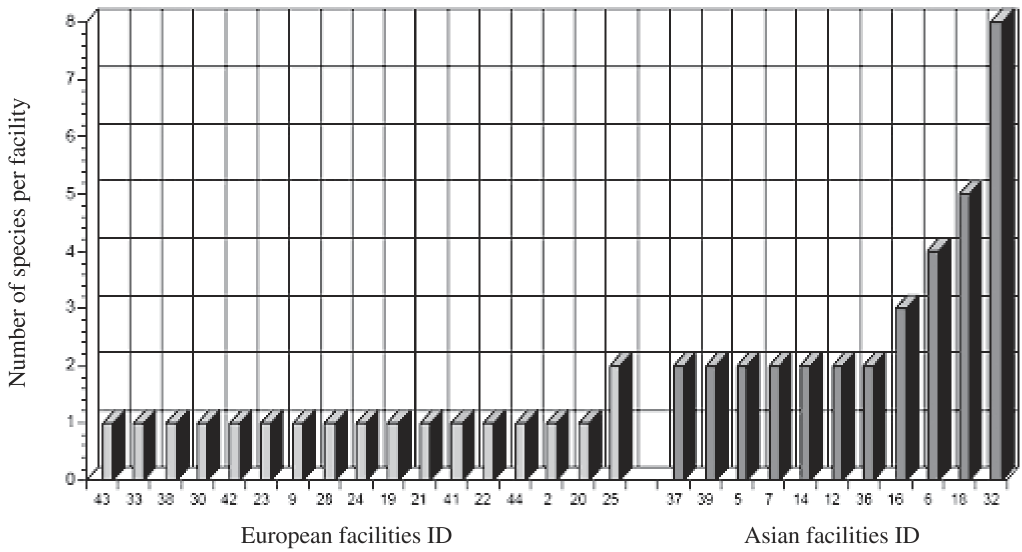

Figure 3.5. Comparison of the variety of displayed cetacean species between Europe and Asia

In sampled European facilities (n = 17), the median number of species was 1 (maximum = 2), in accordance with the statistical analysis of the whole European database. Only 2 of the 7 species displayed in Europe were represented in sampled facilities.

In sampled Asian facilities (n = 11), the median number of species was 2 (maximum = 8). The result was identical to the statistical analysis of the whole Asian database. All 13 species displayed in Asia were represented in sampled facilities.

Sampled European facilities displayed significantly fewer cetacean species than Asian facilities (Man Whitney U test, $Z = -4.234$, $p < 0.0001$).

European facilities display mainly the bottlenose dolphins; the six other species were located in a small number of facilities. In contrast, Asian facilities—mainly Japanese—displayed a wide variety of species. One possible reason for this is that bottlenose dolphins have been more successfully maintained and bred than other species. Europe has a history of importing and trying to maintain and breed as many as 12 species of cetaceans (Collet, 1984); however, the present regulations of many European countries limit or forbid the capture of additional animals from the wild. This makes the replacement of animals difficult and, therefore, promotes breeding and good husbandry. In Asia, capture and trade is not as well-regulated or limited. This makes their acquisition of animals easier. Moreover, Asian exhibits are still greatly oriented toward the display of a collection of several species rather than a socially stable group of any given species. This is a possible explanation for the difference in species variety between Europe and Asia. Many animals from a variety of species that were rescued from strandings are not released. Cetaceans have been part of Asian life, especially in Japan, for a long time—more for whaling purposes than for entertainment and wildlife education. Public perception of cetaceans is slowly changing, as shown by the development of whale-watching and the creation of numerous SWTD programmes in this country (Dudzinski, 1998). At the same time, the integration of husbandry training is progressively orienting facilities toward the sustainable management of stable groups.

Surface Area and Volume of Water per Animal in Artificial and Natural Facilities

The survey data provided information on pool and enclosure dimensions as well as the number of animals. It seemed relevant to determine the average surface area (water surface) and volume of water with which each animal is provided. Although the sample of natural facilities is small, it provided a good means for comparison with artificial facilities. Surface areas and volumes of all pools have been combined, which does not really reflect the reality because the animals are seldom evenly distributed and not all of the pools are available at any one time. Furthermore, social hierarchies might also restrict the space available for individual animals. These theoretical maximum surface areas and volume of water available give a generalised, but still useful, picture of the differences between artificial and natural facilities.

Surface Area per Animal—The surface area is defined as the water surface available to the animals. In sampled artificial facilities (n = 32), the median surface area/animal was 90.5 m^2

(minimum = 14 m², maximum = 195 m²). The surface area/animal ratio in this set of data varied widely among facilities. In sampled natural facilities (n = 7), the median surface area/animal was 400 m² (minimum = 92 m², maximum = 1,633 m²). Surface area/animal ratio varied even more widely from one facility to another.

Although the sample of natural facilities was small, the test showed statistically significant results. The comparison between artificial and natural facilities (see Figure 3.6) showed that facilities keeping cetaceans in natural environments offered a surface area/animal three to four times larger than facilities with artificial environments (Mann Whitney U test, Z = -3.092, $p < 0.025$). It is encouraging to note that many newly created facilities, like those in Mexico, the Caribbean Islands, Palau, and the Philippines, house animals in large natural facilities.

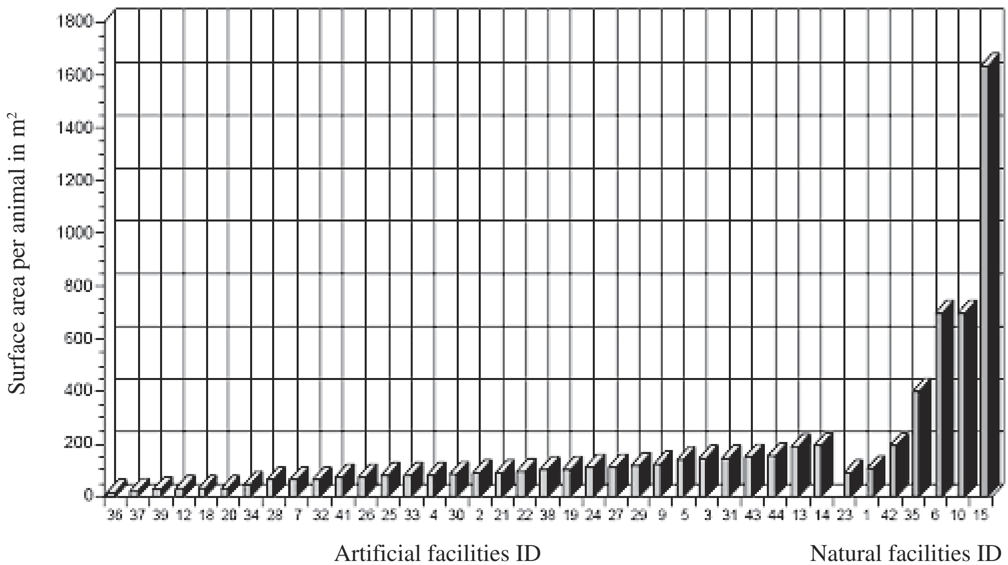

Figure 3.6. Surface area per animal in artificial and natural facilities

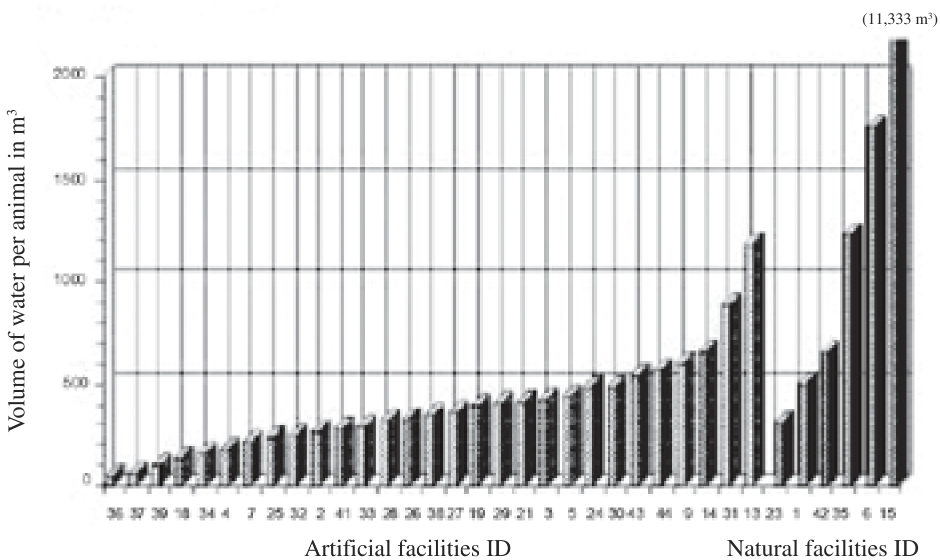

Figure 3.7. Volume of water per animal in artificial and natural facilities

Volume of Water per Animal—In sampled artificial facilities (n = 29), the median volume/animal is 344 m^3 (minimum = 46 m^3, maximum = 1,181 m^3). Volume/animal ratio varies widely from one facility to another. In sampled natural facilities (n = 6), the average volume/animal was 939.5 m^3 (minimum = 306 m^3, maximum = 11,333 m^3). Volume/animal ratio varied even more greatly from one facility to another.

The comparison (Figure 3.7) showed that, in general, facilities keeping cetaceans in natural environments offered a volume/animal ratio that was more than 2½ times larger than artificial facilities (Mann Whitney U test, $Z = -2.67$, $p < 0.01$); however, the graph shows that there is some overlap of values, with several artificial facilities offering a volume/animal ratio as large, or larger than some natural facilities.

All of the five artificial facilities offering the largest volume/animal ratio were facilities constructed or renovated in the 1990s, and four of them were also found among facilities offering the largest surface area/animal. The trend of the late 1990s was clearly towards larger pools in artificial facilities, and the creation of many natural facilities for SWTD programmes likely was influential.

The fact that there are fewer sampled natural facilities than artificial ones might have affected the results. Visits to several natural and semi-natural facilities confirmed that they tend to be larger than artificial ones, however. This difference is probably due to construction and maintenance costs. It is very expensive to build concrete pools and life support systems of dimensions and volumes equivalent to open-sea enclosures; therefore, artificial facilities tend to be downsized compared to natural ones for economic reasons.

Surface Area and Volume of Water per Animal in Europe and Asia

Comparisons were made between European and Asian facilities to determine whether geographical location influenced pool dimensions. Both artificial and natural environments were included, although a great majority of the institutions had artificial environments.

Surface Area per Animal—In sampled European facilities (n = 17), the median surface area/animal was 93 m^2 (minimum = 34 m^2, maximum = 195 m^2). Surface area/animal ratio varied widely from one sampled facility to another. In sampled Asian facilities (n = 10), the median surface area/animal was 48 m^2 (minimum = 14 m^2, maximum = 700 m^2). The surface area/animal ratio varied even more greatly among sampled facilities.

The comparison between European and Asian facilities (Figure 3.8) showed that values are more evenly distributed in European facilities as opposed to Asian facilities. Asian facilities feature both the lowest and highest values across both groups. The median surface area/animal values in sampled Asian facilities were significantly smaller than those in European facilities (Mann Whitney U test, $Z = -1.682$, $p > 0.05$). The median value was almost twice as large in Europe than in Asia.

Volume of Water per Animal—In sampled European facilities (n = 15), the median volume/animal was 388 m^3 (minimum = 240 m^3, maximum = 650 m^3). In sampled Asian facilities (n = 9), the median volume/animal was 211 m^3 (minimum

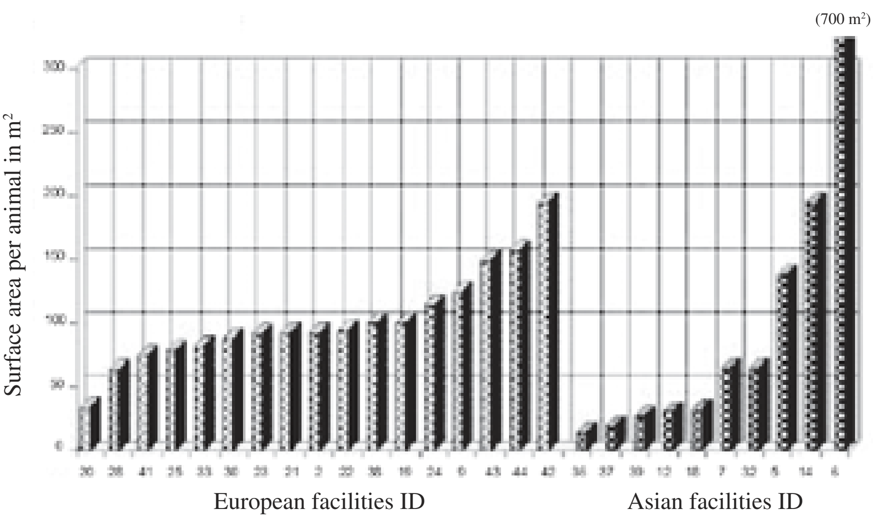

Figure 3.8. Surface area per animal in Europe and Asian facilities

= 46 m^3, maximum = 1,750 m^3). Volume/animal ratio varied widely from one sampled facility to another and features both the lowest and highest values across both groups.

The median volume/animal value in sampled Asian facilities was again significantly smaller than in European facilities (Mann Whitney U test, Z = -1.64, $p > 0.1$). The comparison between European and Asian facilities (Figure 3.9) showed that European facilities offered a volume/animal almost two times larger than those offered in Asian facilities. Among the sampled Asian facilities, 79% of them were located in Japan. It is possible that space constraints and the absence of governmental regulations controling pool dimensions determined these values. In contrast, some European countries (e.g., France and Sweden) (Klinowska & Brown, 1986) have implemented stricter regulations controling pool dimensions and trade during the past fifteen years. The scrutiny of environmental organisations and ministries might also have influenced the space and quality of care provided to the animals during the past decade.

Summary of Survey Findings

- At the time of the survey in 1997, cetaceans were kept in 166 facilities in 42 countries—mainly for display—but some for scientific research and conservation purposes.
- Forty-four institutions from 22 countries participated in the international survey of cetacean facilities, representing 28% of the 157 questionnaires sent out.
- Cetacean facilities were mainly located in the Northern hemisphere in tropical and temperate climates; the majority of them are located in North America, Europe, and Asia. The United States and Japan have the largest number of institutions.
- Construction dates of sampled facilities offered a representative temporal distribution, providing an accurate representation of successes and problems associated with age of facilities.
- The most common group size was between 5 and 10 animals.
- Twenty-one species of cetaceans were kept in human care worldwide. The bottlenose dolphin was the species most commonly displayed. The species variety was greater in Asia than in North America and Europe.
- Facilities with natural environments provided larger habitat dimensions for their animals than facilities with artificial environments. European facilities tended to offer larger habitats than Asian facilities.

The relatively large number of participating facilities in this international survey provided a fair representation of cetacean facilities, with a broad geographical and temporal distribution. They present a wide variety of problems and successes associated with various types of environments, climates, species, and traditions of countries. A larger participation by the USA would have been beneficial because they have some of the largest and most modern facilities; however, the participation of large and renowned facilities in other countries, as well as the visits made to several American, European, Australian, and Asian facilities during this project, allowed the research team to acquire sufficient data on state-of-the-art facilities.

This survey showed that countries around the world do not have similar standards for keeping

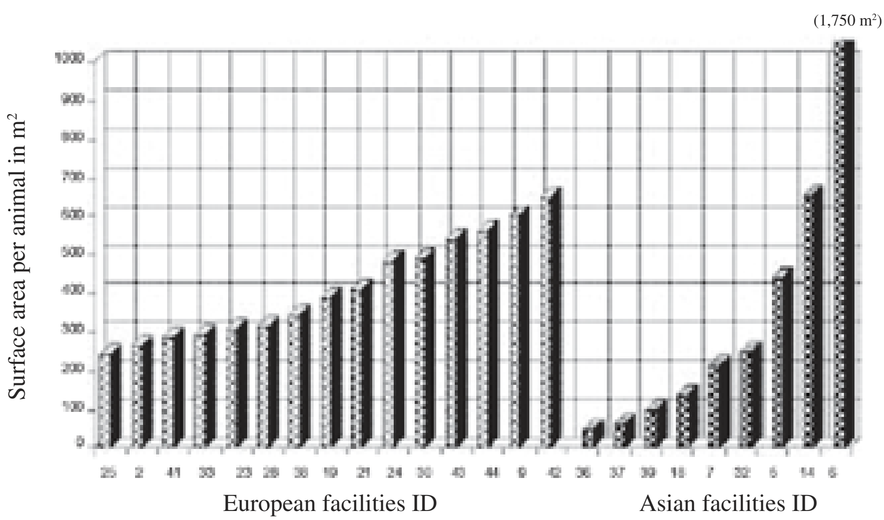

Figure 3.9. Volume of water per animal in Asian and European facilities

cetaceans. They have varied environments, regulations, financial means, and ethical considerations toward cetaceans, which influence the way they display their animals and manage their facilities. This study provided for the first time a clear picture of the average animal group size in display facilities, as well as the species variety on an international scale. It also provided data on housing standards between types of facilities and between geographical areas.

Update

In view of the second edition of the guidebook, I updated the database of facilities in 2005. The latest update shows 199 research and display institutions housing dolphins and whales in 49 countries. One-hundred and sixty-six facilities were recorded just eight years before (164 at the end of the survey because two facilities closed down in the meanwhile)—a progression of 33 new facilities. Some of them existed already in 1997, and some new facilities are still not recorded in China, but at least 30 facilities were created during this time interval. The complete list of cetacean facilities worldwide in 2005 can be found in the Appendix. The following table shows the new geographical distribution. In total, there might be as many as 220 facilities housing dolphins and whales worldwide.

Table 3.2. Geographical distribution of cetacean facilities worldwide

Continent or region	No. of facilities in 1997	No. of facilities in 2005	No. of countries in 1997	No. of countries in 2005
Europe	40	50	17	17
North America	41	41	2	2
Central, South America, and Caribbean Islands	24	43	9	14
Asia/Pacific	54	59	9	12
Africa	3	4	2	2
The Middle East	2	2	2	2
Total	164	199	41	49

This study enabled the creation of the first worldwide database of cetacean facilities. It provided valuable feedback from curators, trainers, and veterinarians of cetacean facilities via the returned questionnaire and the visited institutions. It investigated most aspects of the construction of facilities and many aspects of the management of cetaceans. It provided valuable comments and recommendations for the design of better facilities. I hope this work will be beneficial to all concerned participants involved in facilities' design for the well-being of the animals, their caretakers, and the quality of the experience of visitors that will become more inclined to care for and protect the animals that have touched them.

Literature Cited

Anonymous. (1996). *Membership directory.* Chicago: International Marine Animal Trainers Association.

Anonymous. (2005a). European Association for Aquatic Mammals (EAAM) website. Available online: www.eaam.org. Accessed: 19 July 2005.

Anonymous. (2005b). International Marine Animal Trainers Association (IMATA) website. Available online: www.imata.org. Accessed: 19 July 2005.

Anonymous. (2005c). MarMam listserve. Available online: http://whitelab.biology.dal.ca/marmam.htm. Accessed: 19 July 2005.

Asper, E. D., Cornell, L. H., Duffield, D. A., & Dimeo-Ediger, N. (1988). Marine mammals in zoos, aquaria and marine zoological parks in North America: 1983 census report. *International Zoo Yearbook*, *27*, 287-294.

Asper, E. D., Duffield, D. A., Dimeo-Ediger, N., & Shell, D. (1990). Marine mammals in zoos, aquaria and marine zoological parks in North America: 1983 census report. *International Zoo Yearbook*, *29*, 179-187.

Collet, A. (1984). Live-capture of cetaceans for European institutions. *Reports of the International Whaling Commission*, *34*, 603-607.

Cornell, L. H., & Asper, E. D. (1978). A census of captive marine mammals in North America. *International Zoo Yearbook*, *18*, 220-224.

Defran, R. H., & Pryor, K. (1980). The behavior and training of cetaceans in captivity. In L. M. Herman (Ed.), *Cetacean behavior: Mechanisms and functions* (pp. 319-358). Malabar, FL: University of Hawaii, Krieger Publishing Company.

Dudzinski, K. (1998, Fall/Winter). The best-kept secret in dolphin swim programs is in Japan. *Whalewatcher*, pp. 14-17.

Duffield, D. A., & Wells, R. (1990). Bottlenose dolphins: Comparison of census data from dolphins in captivity with a wild population. *Proceedings of the 18th Annual IMATA Conference* (pp. 4-9), Chicago, Illinois.

Duffield, D. A., & Shell, D. (1994). The past, present and future of marine mammal management. *Proceedings of the 22th Annual IMATA Conference*, Tacoma, Washington.

Duffield, D. A., & Shell, D. (1996). *1996 marine mammal census: Current inventory.* Portland, OR: Portland State University.

Klinowska, M., & Brown, S. (1986). *A review of dolphinaria.* London: Department of the Environment. 247 pp.

Nakahara, F., & Takemura, A. (1997). A survey on the bahavior of captive odontocetes in Japan. *Aquatic Mammals*, *23*(3), 135-143.

Olney, P. J. S., & Ellis, P. (1993). Zoos and aquaria of the world. *International Zoo Yearbook*, *32*, 261-347.

Aquatic Mammals 2005, *31*(3), 320-325, DOI 10.1578/AM.31.3.2005.320

4. Types and Functions of Pools and Enclosures

According to their location, climate, and function, cetacean facilities present a wide variety of shapes, sizes, styles, and materials. Recent construction innovations have allowed new freedom of creativity. The trend towards more naturalistic habitats in zoos is reaching marine mammal exhibits at last. Cetaceans, as compared to pinnipeds or polar bears, are perhaps the last to benefit from this trend. The design of the habitat and the quality of the environmental enrichment are essential to the management of thriving groups of animals and successful exhibits (Doherty & Gibbons, 1994; Stoskopf & Gibbons, 1994). Before detailing the architectural features of the habitats for dolphins and whales in the next chapter, it seemed important to present the various types of existing environments. With the rising development of a wide variety of interactive programmes, such as Swim-With-The-Dolphin (SWTD), Dolphin Assisted Therapy (DAT), feeding and petting dolphins, and Very Important Person's (VIP) educational programmes, in addition to the traditional show, cetacean habitats and displays have become more naturalistic (Samuels & Spradlin, 1994; survey). Allen (1996), in *Wild Mammals in Captivity*, stated that "Visitors are likely to have more favourable impressions of 'natural' exhibits, which, if appropriately designed, can do much to educate and inform them. The challenge to zoos, however, is the construction of environments that not only appeal to visitors, but also fulfil the real needs of the animals living in them. . . . Animals in zoos are neither 'wild' nor truly domesticated."

Dolphins are perhaps in the process of being domesticated; it especially may be true for second and third generations born in captivity. Even so, it is the role of the management to ensure that they are given every opportunity to retain, if not their wild nature, their social and behavioural characteristics.

Often, architects and designers are more interested in responding to the needs of the visitors, especially aesthetically, than the needs of the animals and staff. Their role is to listen to management strategies, husbandry considerations and requirements; to become impregnated by the clear specifications established by the curatorial team before imposing any preconceived view; and to try to combine aesthetically appealing design with safety and welfare considerations.

This chapter presents definitions of the various types of habitat in which cetaceans are housed as well as the function of these various types of pools, and the terms used to name them. These terms, employed by the surveyed and visited facilities, will be used in the following chapters. Rescue facilities are omitted in this chapter. They are designed in a different manner from display and research facilities; they are purely rescue and rehabilitation oriented. Insufficient data on their design were received from the international survey of cetacean facilities for them to be described.

Size recommendations were voluntarily omitted because they should not contradict local regulations. Regulations usually provide the minimum required dimensions and are based on the average body dimensions of animals. Adequate surface area and volume of pools should be determined in compliance with regulations; however, individuals of the species or subspecies regionally may exceed these given dimensions, sometimes by a meter. It is therefore recommended that regulations be exceeded in the best interest of the animals, the staff, and the visitors.

Types of Environments

Two main categories of cetacean habitats have been defined: (1) natural environments and (2) artificial environments. The terms "enclosure" or "lagoon" refer to natural environments, and the term "pool" refers to the artificial environment.

Natural Environments

Cetaceans kept in habitats defined as a natural environment are housed in fenced sea enclosures. The aquatic environment of cetaceans kept in natural facilities is the sea; riverine species are kept in freshwater enclosures. These systems can sustain living organisms such as fish, crustaceans, invertebrates, sea grass, and algae, thus offering cetaceans a greater variety of interactions with their environment. This environment is financially economical because it does not require a filtration system and expensive construction work.

The choice of site for a sea-pen type facility is critical since water quality cannot be controlled. The main disadvantage of natural facilities is the lack of protection against hazards; extreme climatic events; tidal waves; and pollution such as oil spills, drifting objects, and human-made noise. Some facilities have been severely damaged and animals injured or lost because of cyclones, and animals have died because of the ingestion of foreign objects or from severe acoustic disturbances (Geraci, 1986; Sweeney, 1990).

A natural facility preferably will be located where flushing by tidal or wave action ensures an appropriate turnover of the water and the elimination of animal waste. The location will offer the animals a quiet and peaceful environment and will prevent the public from having access to the animals from the sea.

Most facilities of this type are located in tropical or subtropical climates where the water temperature is not subject to extreme variations (e.g., Florida, Bermuda, Hawaii, Israel, Australia, the Philippines, Japan, etc.), but they can be exposed to severe climatic events. Notable exceptions can be found in Denmark, Russia, and Ukraine, where facilities mainly display local species.

Figure 4.1. Interactive wading programme in a natural facility

Semi-Natural or Semi-Artificial Environments

A semi-natural or semi-artificial environment can be defined as naturalistic in the sense that it attempts to recreate natural conditions. I favoured the term semi-natural in this publication.

Such habitats present enclosed lagoons made of natural materials that are physically separated from the sea. Existing lagoons can be used or can be modelled in a natural substrate such as sand and rock. They can be adjacent to the sea or a body of water, or located inland. They can sustain living organisms, such as invertebrates and crustaceans, even fish if possible, which give dolphins greater opportunities to interact with their environment. They add diversity and enrichment as dolphins often are seen playing or chasing them. They also help maintain a cleaner environment and limit algal growth. The water quality is maintained by directly pumping seawater from the sea and/or by a filtration system.

A semi-natural environment can be confused with an artificial environment with naturalistic features (see next section) because the line between them can be thin.

Figure 4.2. SWTD programme in a semi-natural environment

Figure 4.3. Educational wading programme in a semi-natural lagoon

Figure 4.4. Educational wading programme in an artificial pool with naturalistic environment

Artificial Environments

Artificial environments are pools of a geometric or free shape that most often are made of concrete, but sometimes they are made of metal, fibreglass, or plastic. This type of facility allows animals to be housed in places remote from the coast or where the environment does not allow the creation of a semi-natural habitat.

Artificial facilities are more costly to build and maintain than natural enclosures due to the type of construction and the operation of the life support system. Yet, they allow for greater control of the environment and, possibly, better protection against hazards than natural facilities.

Recent artificial environments incorporate naturalistic features and elements. The design of coves and islands, the incorporation of natural boulders or artificial reefs, topography that simulates a rocky shore or beach, and wave generators that recreate an ocean swell are all elements that can bring a certain degree of enrichment to the animal environment according to surveyed facilities. These features often come with greater space, and multiple and free-shape pools that allow for escape from other animals, rest, and diversity of swimming pattern. Although these elements increase the cost of construction and maintenance, they may prove beneficial in the long term by reducing behavioural problems related to poor environmental design.

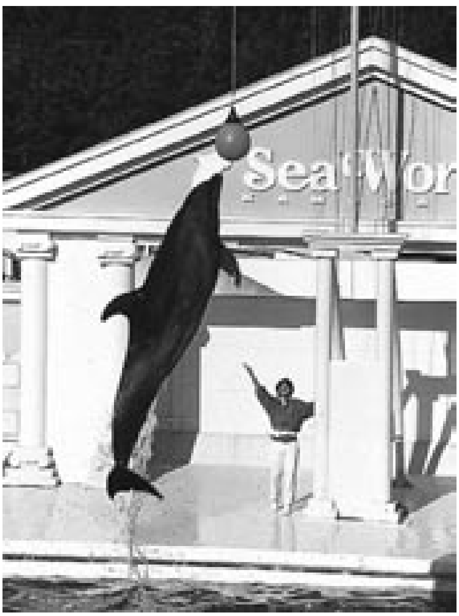

Figure 4.5. Show in an artificial pool

Figure 4.6. Show in an artificial pool

Figure 4.7. Educational programme in an artificial pool

Figure 4.8. Feeding and petting in an artificial pool (Photograph from M. Linet-Frion)

Functions of Pools

Due to the various needs associated with public display, research, training, medical care, breeding, and, most importantly, the animals' social needs, single-pool facilities have evolved into multi-pool complexes. Each pool has a main function, but they each can be used for other purposes as well. Names of pools may vary among facilities. According to USA proposed regulations, the denomination of areas in SWTD facilities varies from those of regular display facilities (Anonymous, 1979-1984, 1995; 1998a; 1998b; 1999). The term "pool" is used in the following section regardless of the environment, but it is applicable to enclosures and lagoons as well.

In practice, pool functions are by no means exclusively defined. Many facilities use pools according to circumstances. In multi-pool facilities, if there is no maternity pool, a pregnant or nursing female is often put into a holding pool not designed for this purpose. If temperamental incompatibility occurs between animals, they sometimes need to be separated. When several species are kept together, but inter-species breeding is not favoured, males and females also need to be separated. Therefore, a facility may very quickly face space limitations. The planning of a new facility too often responds to immediate needs without foreseeing further developments in the size of the colony or its structure, or the many events in the cetaceans' lives. It is therefore important from the planning stage on to anticipate those needs by providing extra space and extra pools, and by designing habitats that ensure that the animals' behavioural requirements are met.

Main Pool

The main pool, also called the show pool in display facilities, is generally the largest pool, which is used for display purposes and often is accessible to public viewing. Animals usually have access to this pool at all times. In the daytime, they may be kept temporarily in another pool before shows or public sessions. All pools where animals are housed permanently, and *a fortiori* not given access to other pools, should follow regulation requirements for the main or permanent, pool. Bassos & Wells (1996), as well as the surveyed facilities, recommend, however, that all animals have access to at least two pools to cater for escape and varied behaviours.

The dimensions and shape of the main pool will be designed to allow the animals to display all species-specific behaviours, including fast swimming, breaching, and unison swimming. It is important to provide ample space to allow for the development of social hierarchy and to provide adequate distance between individuals in the case of multi-species exhibits.

Cetacean species are known to have very different diving and foraging behaviours. It is therefore encouraged that enough space be provided for species known to be pelagic, and thus deep divers, and for those that exhibit gregarious behaviour.

Holding Pool

The pool used for temporary housing or for training purposes is called the holding pool or pen in natural environments. It is also known as the secondary pool or training pool. Holding pools are as important in controlled environments as the main pool. They are used for training, temporary housing, and separation. This pool is usually smaller than the main pool.

Holding pools can be used to house animals permanently only if their dimensions are similar to those of the main pool. Subdividing with partitions to allow a larger number of animals or different species to be maintained separately should be avoided because the dimension requirements will no longer apply. If some individuals need to be separated permanently from the rest of the group because of behavioural problems, pools with space requirements similar to the main pool will be provided.

Many surveyed facilities recommend that a minimum of two holding pools be connected to the main pool to allow for multiple functions. They also advise that a maternity pool be incorporated if breeding is encouraged. If this pool is not incorporated in the design, one of the holding pools will need to be used for this purpose, thereby reducing the overall space available to other animals.

Medical Pool

The medical or quarantine pool, also called the husbandry or isolation pool, is a small pool designed for handling and isolating animals requiring medical treatment (Geraci, 1986). It is preferably connected to the other pools, with the possibility for isolation with watertight gates. In artificial facilities, a medical pool preferably will have a separate water system that can be isolated from the main one to prevent contamination of other pools and so that it can be drained and refilled very quickly. It will ideally provide options for both salt- and freshwater. Dehydrated animals may be successfully rehydrated by being kept in freshwater for limited time (Sweeney & Samansky, 1995). For animals being treated for contagious diseases that require isolation, or for stranded animals, it is preferable to use a pool physically separated from the pools housing resident animals. In natural environments where water control is not possible, a similar option can be considered.

The medical pool can be located between the main pool and one of the holding pools or between two holding pools to facilitate the transfer of an animal. (For the design, equipment, and special features of medical pools, please see Chapter 5.) If the medical pool also is used to treat pinnipeds, appropriate drainage and fencing will be provided.

Maternity Pool

The maternity or nursery pool is an alternative type of holding pool dedicated to pregnant and parturient females, and to nursing females with their calves.

To promote successful breeding and rearing of healthy calves, a facility can have a pool specifically designed for this function. Many problems have occurred in the past due to unsuitable pool design (Amundin, 1986; survey). Pools with a restricted surface area or too shallow depth may interfere with the parturient female's contractions or swimming movements (survey). Calves have been injured and killed by colliding repeatedly with walls because they lack control of their swimming direction in their early days of life. They also have been injured when stranded accidentally in drains or skimmers due to wave action in pools (Amundin, 1986; Sweeney & Samansky, 1995; survey). The pool preferably will have a large, smooth surface area; be free of dangerous protrusions, sharp angles, unprotected drains, or very shallow ledges or beaches where a calf might get stranded; and all fixtures and features should be rounded or padded. It will be equipped with a "baby cot" to prevent the calf from bumping into walls (see "Baby Cot" section in Chapter 5).

To improve the chances of a calf's successful birth and survival, the maternity pool will be large enough to contain the mother, the calf, and at least one additional female. Although the role of the "auntie" dolphin who assists the mother during birth is not conclusive, according to some scientists (Ridgway, 1995), the presence of one or two females during the time of isolation of pregnant or nursing females, and during the delivery, has proved to be beneficial (Evans, 1987). There are many surveyed facilities that have not isolated pregnant females and females with calves, with great success; however, the behaviour of adult males should be closely monitored because male aggression may have resulted in the high incidence of stillbirths and calf mortality in the past (Ridgway, 1995; see also Chapter 8).

Swim-With-The-Dolphin Facilities

Since the beginning of the 1990s, several facilities have initiated programmes proposing interactions with dolphins in the water. There are various types of Swim-With-The-Dolphin (SWTD) programmes—from shallow water or wade interactive programmes to SCUBA-diving with dolphins in open water. In the USA, the Department of Agriculture regulates SWTD facilities, animal care, and personnel training (Anonymous, 1979-1984, 1995; 1998a; 1998b; 1999). Similar programmes exist in countries such as Honduras, Mexico, the Bahamas, Bermuda, Israel, Spain, Australia, Japan, the Philippines, and several Caribbean Islands. USA regulations propose different names for pools such as "interactive area," which corresponds to the main pool, and "buffer area" and "sanctuary area," which correspond to consecutive holding pools.

A majority of SWTD programmes are held in facilities with natural environments, but more and more are proposed in semi-natural and artificial environments. USA regulations propose to discriminate between a "shallow water interactive program," encompassing wade programmes, encounter programmes, or any other programmes in which "a member of the public enters the primary enclosure of a SWTD to interact with the animal and in which the participants remain primarily stationary and non-buoyant," and programmes where members of the public are allowed to swim with the animals in a free or controlled manner. The requirements for the animals, swimmers, or staff will not be covered here. Because the USA regulations for SWTD contain very valuable information and guidelines, please refer to USDA documents available from the *Federal Register* online (Anonymous, 2005).

As is recommended in these regulations, gates between pools should be left open at all times during interactive/swim sessions to allow the dolphins freedom of separation from the public in the water. Both buffer and sanctuary areas are off-limits to the public. The buffer area should be located between the interactive and the sanctuary areas as it "is necessary to ensure that the sanctuary area is an adequate distance from the interactive area" (Anonymous, 1995, 1998a, 1998b). In case this is not the existing layout, if two holding pools are connected to the main pool, it is possible to leave one access gate open and close the other one to transform one holding pool into a buffer area and the other into a sanctuary area. A large shallow ledge or beach will always be available to participants for safety and convenience.

Literature Cited

Allen, M. E. (1996). Introduction: Basic husbandry. In D. G. Kleiman, M. E. Allen, K. V. Thompson, S. Lumpkin, & H. Harris (Eds.), *Wild mammals in captivity: Principles and techniques* (pp. 1-3). Chicago: The University of Chicago Press.

Amundin, M. (1986). Breeding the bottle-nosed dolphin *Tursiops truncatus* at the Kolmarden Dolphinarium. *International Zoo Yearbook*, *24/25*, 263-271.

Anonymous. (1979-1984, 1995). *Specifications for the humane handling, care, treatment and transportation of marine mammals* (9 CFR, Subpart E, §3.100-118). Washington, DC: U.S. Department of Agriculture, the Animal and Plant Health Inspection Service.

Anonymous. (1995). U.S. Department of Agriculture, the Animal and Plant Health Inspection Service, Animal welfare: Marine mammals (9 CFR Parts 1 and 3). *Federal Register*, *60*(14), 4383-4389.

Anonymous. (1998a). U.S. Department of Agriculture, the Animal and Plant Health Inspection Service. Animal welfare: Marine mammals (9 CFR Parts 1 and 3). *Federal Register*, *63*(172), 47128-47151.

Anonymous. (1998b). U.S. Department of Agriculture, the Animal and Plant Health Inspection Service. Animal welfare: Marine mammals (9 CFR Parts 1 and 3). *Federal Register*, *63*(198), 55012.

Anonymous. (1999). U.S. Department of Agriculture, the Animal and Plant Health Inspection Service, Animal welfare: Marine mammals (9 CFR Parts 1 and 3). *Federal Register*, *64*(63), 15918-15920.

Anonymous. (2005a). U.S. Department of Agriculture (USDA) website. Available online: www.usda.gov. Accessed 11 July 2005.

Anonymous. (2005b). USDA, the Animal and Plant Health Inspection Service website. Available online: www.aphis.usda.gov/. Accessed 11 July 2005.

Bassos, M. K., & Wells, R. (1996). Effect of pool features on the behavior of two bottlenose dolphins. *Marine Mammal Science*, *12*(2), 321-324.

Doherty, J. G., & Gibbons, E. F. (1994). Managing naturalistic animal environments in captivity. In E. F. Gibbons, E. J. Wyers, E. Waters, & E. W. Menzel (Eds.), *Naturalistic environments in captivity for animal behavior research* (pp. 126-139). Albany: State University of New York Press.

Evans, P. G. (1987). *The natural history of whales and dolphins.* New York: Facts on File. 343 pp.

Geraci, J. R. (1986). Marine mammals: Husbandry. In M. E. Fowler (Ed.), *Zoo & wild animal medicine* (pp. 757-760). Philadelphia: W. B. Saunders Company.

Ridgway, S. H. (1995). The tides of change: Conservation of marine mammals. In E. F. Gibbons, Jr., B. S. Durrant, & J. Demarest (Eds.), *Conservation of endangered species in captivity: An interdisciplinary approach* (pp. 407-424). Albany: State University of New York Press.

Samuels, A., & Spradlin, T. (1994). *Quantitative behavioral study of bottlenose dolphins in Swim-With-The-Dolphin programs in the United States* (Final report to the National Marine Fisheries Service, Office of Protected Resources). 56 pp.

Stoskopf, M. K., & Gibbons, E. F. (1994). Quantitative evaluation of the effects of environmental parameters on the physiology, behavior, and health of animals in naturalistic captive environments. In E. F. Gibbons, E. J. Wyers, E. Waters, & E. W. Menzel (Eds.), *Naturalistic environments in captivity for animal behavior research* (pp. 140-160). Albany: State University of New York Press.

Sweeney, J. C. (1990). Marine mammal behavioral diagnostics. In L. Dierauf (Ed.), *CRC handbook of marine mammal medicine: Health, disease, and rehabilitation* (pp. 53-72). Boston: CRC Press.

Sweeney, J. C., & Samansky, T. (1995). Elements of successful facility design: Marine mammals. In E. F. Gibbons, Jr., B. S. Durrant, & J. Demarest (Eds.), *Conservation of endangered species in captivity: An interdisciplinary approach* (pp. 465-477). Albany: State University of New York Press.

Aquatic Mammals 2005, *31*(3), 326-350, DOI 10.1578/AM.31.3.2005.326

5. Architectural Design of Pools and Enclosures

Cetaceans, unlike pinnipeds which have access to land, live their entire life in the water. Wild bottlenose dolphins spend up to 75% of daytime traveling and feeding (Shane et al., 1986). They are exposed to a great variety of water movements, and although they tend to avoid extreme conditions and seek shelter during storms, they have been seen surfing big waves and traveling in rough seas. A controlled environment removes many of the choices that animals face in nature. Food, shelter, and medical care are provided, and breeding pairs and group composition is determined by humans (Allen, 1996). Therefore, it is important that human and physical environments propose alternative chances for interactions and behaviours such as playing, resting, mating, and foraging to avoid "boredom" and stress. Dolphins rely on poolmates and humans for social interaction; however, trainers interact with them for only a few hours a day, leaving the animals by themselves the rest of the time. It is therefore important to recreate an environment as close to the wild as possible by promoting the environmental enrichment of the habitat—socially as well as architecturally.

Great efforts have been taken during the past decade to recreate natural environments and enrich the lives of terrestrial animals in zoos (Gibbons et al., 1994). Among marine mammals, sea otters, polar bears, pinnipeds, and even sirenians have been the first to benefit from this trend (Sweeney & Samansky, 1995). Only since the early 1990s have similar concepts been introduced to cetacean exhibits. Curators, veterinarians, animal behaviour specialists, zoologists, and architects are working together to improve the environments of zoo animals to reduce their stress, eliminate stereotypic behaviour, and enhance breeding (Doherty & Gibbons, 1994). By using specialists, a higher level of habitat detail can be achieved without accruing major additional costs (Sweeney & Samansky, 1995). With the increase in the quality of habitats and medical husbandry, survival rates of captive bottlenose dolphins have increased significantly over past years (DeMaster & Drevenak, 1988; Ellis, 1995; Small & DeMaster, 1995a, 1995b). Furthermore, thanks to easier maintenance, there is greater opportunity and time for training and research (Sweeney & Samansky, 1995).

A successful habitat for cetaceans is an environment that promotes well being, where animals cannot injure themselves, where they can interact or avoid each other easily, and where routine medical checks can be performed adequately. Good husbandry of animals that live in the water also means providing a safe and convenient working environment for trainers. Controlled cetacean environments will be planned by studying species-specific behaviours in the wild and introducing significant opportunities for expression of such activities. The need for producing environments that provide greater behavioural richness has to be balanced with the need to provide a safe environment for the animals. Errors in design and the replication of inappropriate facilities will inhibit the creative growth of the environmental design for animals, and may affect the animal's health and wellbeing (Asper, 1982; Markowitz, 1990; Doherty & Gibbons, 1994; Sweeney & Samansky, 1995).

Markowitz (1990) stated a truth that applies today more than ever and that clearly was highlighted by the results of this international survey of cetacean facilities: "In studying the successes and failures of previous habitat design, one can gain considerable information which may help prevent endless replications of the same difficulties. While there is some truth to the notion that sending architects to see existing captive habitats may largely preclude revolution in the design, it is even more apparent that there are an awesome number of identical mistakes in the design of habitats which could easily be avoided if we were better students of each other's facilities."

Innovations in construction technologies, progress in cetacean husbandry, and a better understanding of their behaviour allow curators and architects to design and realise safer, bigger, and more natural exhibits, which better respond to the needs of the animals and the desire of the visitors to see these wild creatures in a more appropriate environment. Larger and more diverse environments provide for a greater degree of socialisation and positive behavioural interactions among individuals and species (Geraci, 1986; Sweeney & Samansky, 1995). The aim of this chapter is to present the suggestions given in the survey questionnaire with the goal of enhancing the quality of dolphin and whale environments in human care.

Please refer to the previous chapter for definitions of "natural," "semi-natural," "artificial," and "semi-artificial" environments; "pools"; and "enclosures." Since many of the suggestions were common to all types of facilities, it seemed clearer to present them by subjects and subdivide them according to the type of environment.

Pools and Enclosures

Many professionals who care for the cetaceans in the visited and surveyed facilities placed a great emphasis on the recreation of environments. Enclosures in which cetaceans are housed should be as naturalistic as possible, considering the fundamental needs of the animals before aesthetic considerations. Environmental enrichment should be considered one of the primary goals to achieve in a facility's design. New enrichment devices will be explored and proposed (e.g., a wave machine, equipment encouraging foraging, etc.). Efforts to promote research and education will be considered early in a facility's design.

Topography, Shape, and Environmental Enrichment

Natural Environment—In open-sea enclosures, topography depends on the selection of the location. Several surveyed facilities with natural environments recommend selecting a spot with a beach that slopes down to adequate depth for ease of access and handling. It is important to provide the animals with both shallow and deep waters. The sides of the enclosure can be made of dry land.

Some open-sea facilities feature pens instead of enclosures. Pens also are used as secondary enclosures. Shapes of pens are usually geometrical as it is easier to install straight portions of chain link fences and wooden walkways. It is important to provide varied shapes and topography in the primary enclosure, and fences will preferably reach the bottom to allow the animals to forage and interact with their environment. Triangular shapes and narrow rectangular pen shapes should be avoided. Larger surface area will be provided if enclosures present inadequate topography, such as rocky mounts or very shallow beaches, that could restrict space available to animals in any way.

Tides are important to consider. The surface area available to the animals might change considerably according to the water level; therefore, surface area for enclosures preferably will be calculated at the lowest tide level. Barrier fences have to extend above the water level at high tide.

Semi-Natural and Artificial Environments—Semi-natural facilities are recreated in much the same way as artificial facilities in regards to topography, shape, and environmental enrichment, but they are built with natural materials or use the existing substrate. Most existing facilities of this type are located near the sea, but it is possible to replicate them inland, provided the nature of the substrate is suitable. The incorporation of live fish and crustaceans allows dolphins greater opportunities to

Figure 5.1. Natural facility

Figure 5.2. Natural facility (Photograph from O. Goffman)

Figure 5.3. Natural facility

Figure 5.4. Sea pens

interact with their environment. They add diversity and enrichment, as dolphins often are seen playing or chasing them. Such fish or crustaceans also are a good indicator of water quality. They help maintain a cleaner environment; however, they also have to be monitored for diseases or parasites. Visitors are more attracted to exhibits displaying cetaceans in a more natural setting, especially if visitors interact with the animals in the water.

Topography is an aspect of the pool design that has been neglected in most artificial exhibits. Many pools present a very bare and sterile environment—flat bottom, straight wall, geometrical contour shape, painted concrete, and still water. Variations in the topography and irregular shapes provide the animals with opportunities for varying their swim patterns. Irregularly shaped areas can prevent potential stereotypic behaviours and help reduce stress by introducing variety. An irregularly shaped area has less reverberation and, hence, provides a better acoustic environment for cetaceans. No matter how large and varied a pool's shape, it will never replace the unlimited space of the ocean, especially for pelagic species. Even so, some elements can make it look closer to inshore waters or coves.

An island, a rocky edge with crashing waves, or a beach present interesting additions to the pool environment. Dolphins may favour deeper and larger areas of the pool for breeching and fast swimming; and shallow waters, confined space, or coves for resting (Bassos & Wells, 1996).

The following is a summary of features recurrently suggested by surveyed facilities for the design of pools and enclosures in regards to topography, shape, and environmental enrichment, as applied to both semi-natural and artificial environments (unless otherwise stated).

- Avoid geometric contour shapes (e.g., circle, square, rectangle, even a "D" shape); prefer irregular shapes. Circular pools tend to create vortex and encourage stereotypical swimming. Angles in square and rectangular shapes are wasted space and often create dead corners in water circulation. An oval shape can be appropriate for a medical pool. "D" shapes or kidney shapes have been especially favoured for show pools and can be enhanced by incorporating an extra space with a shallow recess or an island.
- Curves preferably should have a radius inscribed in obtuse angles (no less that 90º).
- Provide at least one elongated area of great dimension, which allows for bursts in swim speed.
- Design the pool bottom with varied depths and contour shapes. This provides a better, less reverberant acoustic environment and greater variety in swim possibilities.
- Incorporate underwater humps and hills, and beaches and shallows; some species, such as the bottlenose dolphin, like to rub themselves or strand themselves occasionally in shallow waters.
- Favour slanted walls over vertical and parallel walls in concrete pools. It is even better to eliminate the delimitation between the wall and the bottom (e.g., bowl shape) (see "Acoustics" section).
- The pool bottom can be covered with sand to allow the dolphins to forage and play (with protection over drains and sumps), providing the animals have not shown tendencies to swallow sand (see "Construction and Materials" section for restriction of use).
- Add an artificial coral reef or rock work, boulders, and underwater landscaping (provided they are not used as a target to redirect destructive behaviour of the animals (Sweeney, 1990). Similar recommendations apply for the following paragraph. Rocks should either be above or partially above water or safely at more than 2 m deep to avoid an animal mistakenly leaping on an unseen ledge. Their surface should be smooth to prevent skin abrasion and to facilitate cleaning.
- Add live fish and crustaceans, sea grass, and so on, when the water system allows (venomous and spiked animals should be avoided) (see Chapter 6 for requirements).
- Incorporate cascading water, sprinkler, hose, and bubble stream with a possibility of training the animals to activate or turn off these features as part of the enrichment.
- Incorporate a wave generator in concrete pools. A beach will be designed on the other end of the pool to absorb waves. The wave generating mechanisms should be completely out of reach of the animals for safety reasons. Wave generators might destabilise natural substrates such as sand and stones in semi-natural enclosures.
- Each pool preferably will be connected to at least two other pools for easier animal management or to provide refuge space for animals.

Construction and Materials

Facilities must be constructed of such materials and maintained in good repair so as to ensure that the animals, the attending personnel, and visitors are protected from injury and from the entrance of unwanted animals (Anonymous, 1979-1984, 1995). Materials have to be durable, non-toxic, and easily cleaned and disinfected. Consumption of pool components may occur as portions of the pool disintegrate or as the animals disassemble portions of their environment (e.g., peeling paint, crumbling artificial rocks, tiles, burst membranes, protruding pipes, etc.). For this reason, it is important that design efforts be directed not only toward

Figure 5.5. Wave machine

Figure 5.6. Wave action in dolphin pool (Photograph from M. Linet-Frion)

Figure 5.7. Sprinklers

Figure 5.8. Bubbles stream

the development of interesting and stimulating exhibits, but also to make them impervious to the destructive tendencies of the animals (Sweeney, 1990; Sweeney & Samansky, 1995).

The structure of artificial pools and semi-natural enclosures are different. Artificial pools are made with concrete, and semi-natural enclosures are made using natural substrates.

Several materials are omitted in this list such as metal, plastic, and fibreglass. Metal pools are not recommended by the few surveyed facilities using them since they are difficult to maintain, rust easily, are very noisy, and do not allow a wide variety of shapes. Plastic and fibreglass pools, limited in dimension and resistance, are used only as rescue pools and for temporary accommodations.

Structure and Lining

Semi-Natural Environments—Semi-natural lagoons are dug in natural substrate—either rock, sand, or soil. Some suggestions relative to semi-natural environments follow:

- When the substrate is sand, the excavation is lined with a clean 50 cm thick bed of sand, which will compact but still allow the passage of ground water. It is then lined with a geofabric membrane that permits the osmotic transfer of water from the surrounding groundwater while not acting as a filter. The main purpose of this style of membrane is to prevent foreign objects, which could be harmful to animals, from floating through the substrate. The main problem with this type of environment lies with groundwater levels because they influence the surface levels of the pools. In a show environment, this can be critical for slide-out beaches and the stage. Groundwater has an even greater influence when it is subject to tides that can cause great fluctuations in water level. High groundwater requires installing sheet piling with pumps to remove water. If the pool is emptied for technical or medical intervention, the pumping system will need to be reactivated.
- There are other kinds of semi-permeable or impermeable membranes available, depending

on soil conditions. After the membrane is put in place, another layer of sand is placed over the membrane. It needs to be at least 50 cm thick. If the sand bed is less than 50 cm, the dolphins may expose the membrane by their foraging and tail movements. Anaerobic bacterial activity will develop in the sand and stabilise; this should not create problems providing the turnover of the water is appropriate. Metal ions in the water might give the sandy substrate a brownish colour. Metal ion composition of the groundwater at the site location will be investigated to ensure that it is safe for the animals. There is a potential danger of impurities leaking into the system, but if the turnover rate of the water is good, it can be avoided. A second membrane layer above a thick layer of several meters of sand can be added to create a buffer zone and guarantee watertightness when impermeable membrane is preferred.

- The perimeter of the recreated lagoon can be either left sandy with the membrane sealed underneath on a concrete beam or lined with a thick layer of stones, preferably hand-placed on the upper surface of the inclined batter and down to approximately two-thirds of the lagoon's depth. Some portions can slope gently to form a sandy beach. Boulders also can be added. Sump grates should be placed at a higher level than the bottom, and their perimeter should be lined with stones or concrete to maintain the membrane in place around them.
- Bank stability may also be a problem according to the nature of the substrate. The geotechnical qualities of each site must be assessed before this type of environment can be designed or constructed.
- On a rocky substrate, a membrane might be required if the rock is not solid and uniform, or if it is mixed with soil. Lava rock makes an excellent lagoon substrate and acts as an efficient natural filter. Granite also makes a good natural substrate that can provide a dramatic underwater landscape.

The primary advantage of this type of environment is that it offers a more natural habitat with greater possibilities for the animals to interact with their environment because marine organisms such as fish, crustaceans and shrimps can thrive, provided the life support system (LSS) allows it. It can be much larger than concrete pools and offers a wide variety of shapes and topography. It also is easier to maintain and clean because algae are part of the environment and do not need to be removed frequently. It is cost-effective compared to concrete pools for construction and maintenance.

Artificial Environment – Concrete pools have been used to house dolphins since the beginning of modern oceanaria. Facilities have reported several problems related to concrete foundations and have proposed recommendations for adequate construction and maintenance:

Figure 5.9. Semi-natural show lagoon

Figure 5.10. Semi-natural lagoon

Figure 5.11. Semi-natural lagoon

- The structure of the pool will be designed to resist changes in pressure when the pool is empty. This can be achieved by means of a thick slab or multiple anchored ground piles. For budget constraints, this aspect often is neglected, resulting in concrete failure at the first attempt to empty the pool.
- Concrete pools with geometrical shapes and regular surfaces are built traditionally using formwork. To create free shapes and

Figure 5.12. Semi-natural lagoon (Photograph from J. M. Chollet)

Figure 5.13. Semi-natural lagoon built on granite rock

irregular forms, a metal structure is installed following the shape of preformed ground, then sprayed with liquid concrete such as Gunite™ or Shotcrete™.

- Concrete failure is due to movements from settling, shrinkage, surface-spalling, and scaling. If the pools develop leaks, saltwater can travel into the wall, attack and corrode the steel reinforcement rods, and deteriorate concrete further. Preventive maintenance and sealing of surfaces with a concrete sealer protects concrete and rockwork. Krajniak (1988) and several surveyed facilities recommend to re-coat pool surfaces more often than most manufacturers do.
- All of the cracks will be filled up by injecting them with epoxy under high pressure. Cracks around window frames should be given special attention.
- Concrete will be coated with a nonporous and waterproof finish. If paint is used, its composition should be checked carefully to avoid any dangerous chemicals that could be released into the water and be absorbed by the dolphins through their mouth mucosa or skin. No butyltin compounds, such as TBT (tributyltin), commonly found in antifouling paints, nor any paint containing heavy metals should be used as it may jeopardise dolphins' health by suppressing their ability to fight infection (Kannan et al., 1997) (see Chapter 6). Epoxy paint, like all other coatings, has to be applied under very strict hygrometric and temperature conditions. Waterproofing and pool liners should be applied properly when first installed. The surface must be clean and dry, and the proper temperature and procedures recommended by the manufacturer must be maintained at all times. Otherwise, it can result in early scaling and improper waterproofing (Krajniak, 1988). Remedial work to remedy poor installation can be extremely costly and time-consuming, as well as stressful and dangerous for the animals since they can swallow pealing paint or coating. One surveyed facility has reported that dolphins have ingested bits of pealing paints due to the improper polymerisation of the components, and that traces of the same components were found in the blood analysis of the animals. Fibreglass-reinforced polyester, which is waterproof and smooth, makes an excellent coating, preventing algal growth more efficiently. It eliminates the necessity to repaint a pool at regular intervals. Newly constructed pools and applied liners, as well as a new LSS, should be dry and used for several days to weeks before animals are introduced to ensure that there are no toxicity concerns (e.g., concrete leaching, paint fumes, etc.).
- Tiles are not recommended as a pool liner. They are slippery and often crack; broken tiles are difficult to replace; and grout is difficult to clean. Regular scrubbing of grouts wears them in a way that makes an excellent substrate for algal growth.
- Pool surfaces replicating a natural environment are encouraged as long as they do not jeopardise the well-being of the animals.

Figure 5.14. Artificial pools with naturalistic features

Figure 5.15. Artificial pools with naturalistic features (Photograph from M. Linet-Frion)

Figure 5.16. Artificial show facility

Figure 5.17. Artificial show facility

Figure 5.18. Artificial show facility

Figure 5.19. Artificial show facility

Figure 5.20. Artificial show facility

Figure 5.21. Artificial show facility

Figure 5.22. Killer whale show pool (Photograph from M. Linet-Frion)

Sand and Rocks
Although present in nature, some elements may be used differently in a display by both captive and wild-born animals. Sweeney (1990) and several visited and surveyed facilities reported that dolphins often swallow sand, gravel, small stones, seaweeds, and various objects dropped in the pool. Stones of large diameters, around 30 cm, are preferred. Fine crushed shells will preferably be used rather than silica sand because shells can be digested if swallowed. Natural and artificial rocks should not present dangerous protrusions or overhangs. The finish and surfacing of artificial rocks will be discussed with curators according to the potential destructive behaviour of animals and then properly specified to the contractor prior to construction. The surface has to be smooth, and its quality and finish must not be porous and friable. Crumbly material can be swallowed by animals. Abrasive finishes may provoke skin damage and tooth erosion in animals (Sweeney, 1990). Rough surfaces may contribute to increased algal and bacterial growth and need therefore to be frequently and thoroughly cleaned (Sweeney & Samansky, 1995). Proper waterproofing should prevent the corrosion of the internal metal structure. Internal Polyvinyl Chloride (PVC) structure can be preferred over metal, but it does not allow the same freedom of shape. In both cases, it is recommended to fill the sculpted rock with concrete to strengthen it and avoid dirt traps.

Figure 5.23. Artificial rockwork

Figure 5.24. Rockwork structure

Figure 5.25. Artificial coral reef

Partitions and Fences
Natural and Semi-Natural Environments—The division of space within a pen or a pool is made by walls, partitions, or fences. The design and materials used should be convenient and safe for both animals and trainers. Here are some examples observed in or reported by facilities:

- In a lagoon, various types of partitions can separate the main enclosure and the holding areas. They should be adapted to the tidal action and maintenance requirements. Several types of chain link fences can be used—for example, the classical type (diamond-shape pattern, plastic coated metal) or the reinforced type (square or rectangular pattern, stronger welded mesh). Both are convenient and cheap, but they have to be maintained regularly to remove the algal and coral growth and to check for protruding or rusted portions. They corrode quickly and do not have a very long life span. Facilities located where fouling and coral growth is fast will ensure that the fence does not collapse under the weight of incrustation. Chain link fences are preferred aesthetically in areas with little tidal action since they are collapsible. They are quite easy to move or replace. Mesh size will be wide enough (10-15 cm) to allow fish and crustaceans to swim through and to ensure that animals do not get their rostrum stuck between meshes. A regular schedule of inspection by divers is recommended.
- Partitions can be made of stainless steel bars, which are resistant to corrosion and easy to maintain. Since they are rigid, they cannot collapse under tidal action unless they have articulated parts. They can create noise in the dolphin's environment, however, if not carefully padded with rubber at the junctions. They are more costly than chain link fences, but they also are more durable.

- Partitions also can be made of concrete piles. Piles can be solid or hollow and filled with sand. This ensures a resistant and durable partitioning, but cannot be easily moved or modified.
- Timber is frequently used as partitioning, fencing, walkways, and platforms in natural facilities. A variety of wood types can be used for their durability and resistance to marine borers in marine environment such as teak, greenheart, and bakau woods. Wood piles used in marine installations (e.g., wharves, jetties, breakwaters, etc.) usually receive preservative and antifouling treatments to increase their life span. Creosote oil is a mixture obtained from the distillation of coal and tar and is a good preservative. CCA (Copper Chrome Arsenate) is another widely used wood preservative. They both have excellent antifouling properties, but they are toxic. Creosote contains impurities that are toxic, carcinogenic, and mutagenic. In some regions of Canada, its use is banned in protected water supply areas or in swimming areas whether protected or not (Anonymous, 1995). Experiences have indicated that 3.5% of arsenic from CCA is leached from the timber after one month of exposure to running water, and arsenic contamination of potable water supplies occurred due to the leaking from a timber treatment plant. The effect on marine mammals is unknown; however, if toxins from these products affect humans, they potentially could affect cetaceans living permanently in their vicinity. Therefore, I recommend to look for alternative wood treatments, such as silicone and epoxy, or use naturally resistant untreated woods. Wood should always be carefully maintained to prevent algal and fungal growth. It also can become rapidly slippery if not regularly scrubbed. I suggest that the use of smooth-surfaced wood for walking platforms be avoided and that groove patterns be incorporated to increase foothold.
- Nets (nylon fishing nets or rope nets, either loose or mounted on frames) should be banned for use as fences, partitions, or gates in dolphin enclosures because they are extremely dangerous for the animals. As reported by several facilities and by Myers & Overstrom (1978), they have been used in the past, resulting in the death of animals by suffocation because dolphins entangled a part of their body in the net and were unable to withdraw. They can only be used as a device to restrain an animal or push it toward a pool. Only in maternity pools can a special kind of net be used as a "baby cot" (see "Baby Cot" section).

Artificial Environment

- In pools, concrete walls are used to partition space between primary and secondary pools or the medical pool. These do not allow physical, acoustic, or visual communication between animals. Pools normally are connected by gates. If the water surface is level with the concrete platforms, a concrete hump or a low stainless steel separator can be installed between pools to prevent dolphins from sliding over the partition.
- Stainless steel bars and chain link fences also can be used as partitions, but pools should not be subdivided if the original design did not include provisions of space for this purpose. Spacing between bars or meshes will either be wide enough or small enough according to the housed species to prevent their rostrum from getting stuck between them.

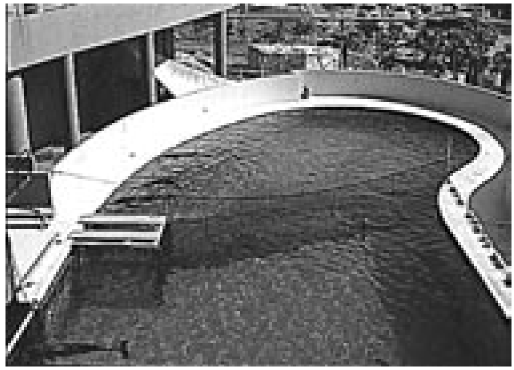

Figure 5.26. Net dangerously used as separator (Photograph from Pr. Lam Khee Poh)

Pool and Enclosure Surroundings

- Platforms and beaches used by dolphins to slide-out will be very smooth and nonabrasive, and edges will be rounded. Trainers should wear appropriate no-slip footwear.
- Walkways around the pools are exposed to water and, therefore, will be made of or covered with no-slip and waterproof materials, and they also should be properly drained. Waterproof-treated concrete and concrete covered with no-slip materials are recommended. Stone paving is suitable for natural or semi-natural facilities. Sand and gravel are not recommended because they can easily be washed away and are too soft and unstable for walking and pushing carts. Furthermore, according to several surveyed facilities, they can be used by the public to throw at the animals. Aggregated sand or gravel are preferred. Rubber and vinyls without no-slip treatment, metal, or wood are not recommended; they are slippery and difficult to maintain. Wood, with the exception of teak and a few other types of wood that are resistant to marine environments, tends to rot quickly. Marine woods are suitable for footpaths and fixed or floating platforms, but should be kept clean and

free of algae. Any selected material needs to be evaluated for its durability, its potential to be disinfected and maintained, and that it does not adversely affect the health of the animals.

- Pillars and columns will be avoided in close proximity of the pools because they are obstacles to the circulation of the staff and the use of equipment.
- Because the air around saltwater pools or near the sea is highly corrosive, poles, pipes, and all fixtures will be selected for their resistance to corrosion and rotting and will not protrude dangerously. PVC and high-quality stainless steel are recommended by a large majority of facilities. Mediocre-quality stainless steel will corrode more quickly and thus will need faster replacement. The use of aluminium is controversial, and some surveyed facilities advise to avoid it because of corrosion problems. All elements in contact with dolphin pool water need to be thoroughly cleaned to avoid build-up of dirt, bacteria, and fungi that could be harmful to the animals. Any potentially dangerous overhangs or protrusions in or around pools that can bruise or harm animals and trainers will be avoided. Cetaceans may damage appendages on overhangs that do not make contact with the water surface. In contrast, animals can navigate to avoid structures that are in the water (Sweeney & Samansky, 1995).
- Any furniture, piece of equipment, or element of decoration in the pool or enclosure will be evaluated in terms of potential danger to the animals and its capacity to be properly maintained and cleaned.
- In windy areas, foliage and litter can be a problem if blown in the pool and swallowed by the animals. Trees and plants will be selected with care accordingly, as some species such as laurel, poinsettas, and some yews can be toxic. Dustbins with lids have to be installed in sufficient number. Pools should frequently and regularly be checked for litter by divers.

Husbandry, Training, and Enrichment Features

Great care will be placed in the design of a safe and convenient area for husbandry and training that benefits the animals as well as their trainers. Adequate features will allow easier and faster handling of the animals, lower stress, greater safety, and more efficient and cooperative training. Architects and engineers hopefully will be very attentive in incorporating into their design the requests of curators, trainers, and veterinarians in regards to circulation and access, husbandry features, medical equipment, materials, drainage, and life support systems, as they are the daily users of the installation and know the animal requirements better than anyone. Inappropriate design can result in poor working conditions for the staff and an unsafe environment for them and the animals. Environmental enrichment for the well-being of the animals and educational programmes for visitors also will be considered and integrated early into the facility's design.

Harassment

Cetaceans should not be housed near animals that cause them stress or discomfort (Anonymous, 1979-1984, 1995). They should be able to escape the harassment of other animals at all times. Outside of training sessions and Swim-With-The-Dolphins (SMTD) sessions or shows, they can be allowed to swim around the main pool and holding enclosures. The topography, shape, and size of all enclosures, and the connections between the pools, should provide animals with a refuge.

All cetaceans will be protected from abuse and harassment by the viewing public through the use of a sufficient number of employees or attendants to supervise the viewing public or by physical barriers such as fences, walls, glass partitions, or distance or both (Anonymous, 1979-1984, 1995). The animals also will be protected from possible injuries caused by the ingestion of foreign bodies that might be intentionally or accidentally introduced by the public. Visitors will be prevented from entering the pool area during non-authorised time frames. The admission of the public near the dolphin enclosure for limited periods also increases the animals' interest and curiosity towards visitors.

Beach

Beaches or slide-outs are gently sloping surfaces made of sand or concrete, which serve various purposes. In SWTD programmes, it is more comfortable for guests to enter the water from a beach to interact with the animals. For medical and training purposes, it is easier for trainers to handle, examine, or restrain an animal from a beach, especially if it is trained to beach itself. Bottlenose dolphins also use beaches and shallow areas to rub themselves or play. Sandy beaches will be made of smooth sand (no pebbles, coral bits, or rocks). Artificial beaches can be coated with a smooth liner to prevent skin abrasion. They can be built as shallow underwater platforms (5 to 15 cm) or sloped with only a portion submerged. I suggest that an artificial beach or platform for training and husbandry be at least 1.5 MAL long and wide (Maximum Adult Length of the largest species housed in the pool; see Cetacean Species Information Table).

Ledge

Many pools are built with submerged ledges and low walls surrounding the perimeter that can be used as training stations. A majority of surveyed facilities

Figure 5.27. Beach in semi-natural facility

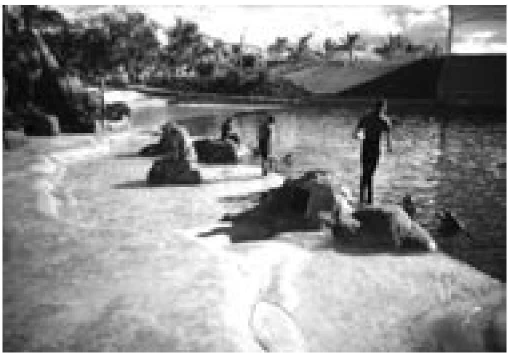

Figure 5.28. Beach and show stage

Figure 5.29. Beach in an artificial facility (Photograph from B. Mercera)

with artificial pools expressed the need to have running ledges around the perimeter of the pool to allow better trainer circulation and to provide a variety of training stations. Ledges often were reported in the survey as being too narrow in existing facilities. They preferably will be submerged and of comfortable dimension—at least 1- to 1.5-m wide. The ledge will be slightly sloped toward the inside of the pool. Animals may jump or be pushed out of the pools while engaged in play or aggressive behaviour (Sweeney & Samansky, 1995). It is important to have a low wall limiting this ledge, 0.6 to 1.0 m in height, or a physical barrier made of rocks or other materials to prevent dolphins from sliding out of the water and getting stranded for extensive periods which potentially could cause dehydration and overheating. Several surveyed facilities have reported that dolphin calves have been trapped in drains or recesses and have been severely injured or even died. Skimmers and drains should be protected by grates, and their design and location will not prevent the animal from sliding back into the water (Amundin, 1986; survey). One surveyed facility suggests that there be wider areas with a removable barrier instead because it is sometimes necessary to bring large or heavy equipment close to the water.

Figure 5.30. Ledge (Photograph from B. Mercera)

Underwater Training Platform

An underwater platform is a very useful feature for medical examination and husbandry training, especially in facilities with no ledge or beach. It allows trainers to stand knee-deep to waist-deep in the water while handling an animal. An underwater platform can be a shallow recess, a protruding lip from the wall, or an extended ledge or beach. It can be between 0.8- and 1.2-m deep and minimum 1.5-m long by 1.2-m wide.

An adjustable underwater platform offers more versatility in its use—for training as well as husbandry purposes. This platform can be made of stainless steel or PVC and have rounded edges. It is important to maintain it properly to avoid corrosion and slipperiness due to algae, and check the mechanism to adjust the level regularly to ensure it is functioning at all times.

Training Station

Training stations and their locations vary according to the pool design and the climate. In cold weather, trainers tend to avoid contact with water. In tropical regions, however, they may prefer to train in the water or stand on a beach, an underwater platform, or a submerged ledge. When not in the water, various training positions may be used.

Figure 5.31. Adjustable underwater platform

Figure 5.32. Underwater training platform

Trainers usually stand on the pool's edge or a platform, but when the pool deck is higher than the water level, trainers have to sit on the edge of the pool or kneel and bend down uncomfortably to reach the animal. A more comfortable training position is to stand behind a waist-high wall or glass panel so that the trainer can reach the animal easily without bending down. This position is not splash proof though. If the level of the water in the pool is flush with the pool deck level, a drop of the ground level adjacent to the pool, or a "pit," would allow this position as well.

Floating platforms offer the advantage of following the tidal action in natural enclosures or the wave action created by the animals in pools. Since it is not intended to act as a slide-out area, it is preferable to cover it with no-slip material to prevent accidents when trainers stand on it when it is in motion. A submerged ledge or underwater step, about 0.8-m deep and wide, on the side can be included in the design. PVC is recommended because it is easier to maintain than wood or metal. It also can have internal compartments that can be filled with water, as a ballast, to submerge it partially.

A training box is another solution to enable training at waist level without being in contact with the water. The box is made of stainless steel, PVC, or fibreglass; installed in the water; and fixed to the pool deck. Edges will be smooth and rounded. Water can be evacuated by a pump.

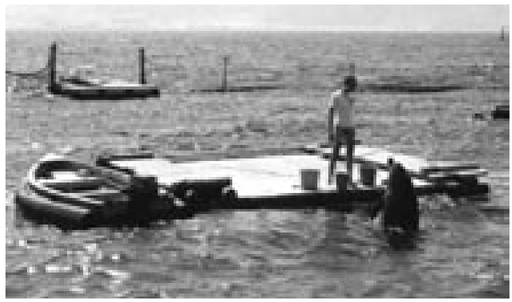

Figure 5.33. Training platform in natural facility

Figure 5.34. Floating platform with ballast system

Figure 5.35. Training box (Photograph from G. Tizzi)

Lifting Bottom and Jig

Some facilities have medical pools equipped with a jig system or a lifting bottom, which occupies the entire space of the pool. Both systems use platforms that can be lifted manually, electrically by a winch, or by a pneumatic or hydraulic system. Proper maintenance prevents any leakage of oil from these systems into the pool. The platform can be made of panels of hard plastic mounted on a supporting structure (recommended for large animals) or can be made of a soft plastic sheet mounted on a stainless steel frame. A rigid system allows a better level of support for the trainers and veterinarian's weight, which eases access to the animal. According to the survey, both systems have proved extremely useful for examining and treating animals while reducing handling and stress. When installed in a large enough medical pool, about 2-m deep, a mother and her calf or a sick animal can be kept there for several days or weeks, offering the possibility of stranding the animals without having to empty the pool. This is very useful if the examination or treatment has to be performed or administered every day.

Figure 5.36. Manual lifting platform or jig

Figure 5.37. Medical pool with lifting platform (Photograph from Zoomarine)

Figure 5.38. Lifting medical platform for large whales

Baby Cot

Several facilities have installed a "baby cot" in their maternity pool, which is a protective net stretched along the perimeter of the pool to prevent the calf from bumping against the walls. It can be installed a few weeks prior to the expected time of birth. The net has very small mesh, like anchovy fishing nets, to prevent the animals from getting caught by its flippers, snout, or teeth, and must be stretched very tightly. This prevents drowning. The baby cot is in use in three surveyed facilities, and they are all very satisfied with this device.

Crane and Hoist

Cranes and hoists are essential devices in the management and husbandry of a captive group of animals. They are used to lift an animal in a stretcher for transportation, medical examination or treatment, and weighing. If a crane is not available near the pools, dolphins and whales would have to be brought in or lifted out of the water by hand-carried stretchers, or by a crane mounted on a truck in the case of very big animals like killer whales. The layout of pools and enclosures sometimes prevent truck access. It is therefore highly recommended that a crane be installed near the pools. It should either rotate to serve several

Figure 5.39. Manual hoist with hooked scale

pools, or it can be mounted on a rail. Some facilities employ a hoist and rail system mounted in the ceiling over the pool in indoor facilities.

Weighing Scale

The body weight of dolphins and whales has to be monitored regularly to control their physical condition and growth rate. Loss of weight is one of the first indications of illness, but it is difficult to assess by observations (Geraci, 1986; Sweeney, 1990, 1993). Body weight should be checked regularly as part of the normal physical assessment or in the case of illness and medical treatment. Concrete beaches and decks allow the installation of permanent or temporary slide-out scales. A single trainer can perform the task of weighing a trained animal with a slide-out scale. If a facility has several sets of pools, a portable slide-out scale is recommended, but the calibration must be checked regularly to maintain accuracy. I advise having a fixed scale for larger animals (false killer whales, pilot whales, killer whales, Risso' s dolphins, etc.), however, because the scale might be too large and heavy to be moved easily.

The alternative to slide-out scales is to put the animal in a stretcher and lift it with a crane equipped with a hooked scale between the cable and the stretcher. It is a nontraumatic experience if the animal is trained to perform it, but the animal can be reluctant to enter a stretcher freely if it is ill. This technique requires more humanpower and can be stressful to the animal.

Figure 5.40. Weighing scale

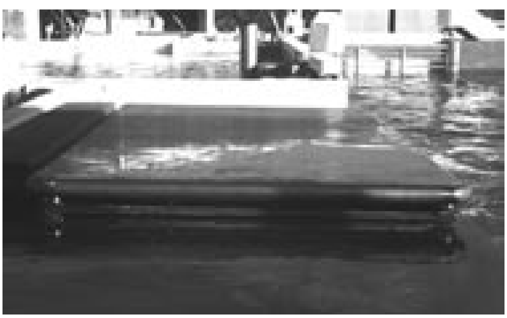

Figure 5.41. Weighing scale for large animals

Fish Flush

Two surveyed facilities mentioned an enrichment device that provides foraging possibilities. It is made of several tubes included in the concrete of the pool or mounted on the walls and bottom. It is equipped with a flushing device, manual or pneumatic, that flushes food into the pool. It allows random distribution in terms of location and time, surprising animals and encouraging them to forage. This device needs to be properly disinfected after every use.

Observation Points

Many surveyed facilities recommend that a high observation stand or platform be installed near a pool to monitor the animals' behaviour unless a nearby building terrace, rooftop, or theatre stand can be used. This facilitates a better view of dolphin movement under water since the reflective glare at water level usually prevents good observation. The platform will be at least 4-m high to allow a good view of the pool. The structure should not represent a hazard for the animals. In indoor facilities, a mezzanine floor can be designed overlooking the pool, but not extending above it unless the proper height is respected. The upper seats of the show theatre also can be used for this purpose. It is also recommended to locate the staff room close to the pools so that they can observe the animals at all times.

Shade and Colour

Cetaceans in the wild do not have access to shade, except the coastal or riverine species. They do not need protection from the sun since they are submerged most of the time. Exposure to direct sunlight is actually important to marine mammals for skin irradiation to ensure adequate vitamin D levels (Asper, 1982). In captivity, however, animals tend to float at the surface more often and get sunburned. It is therefore important to provide them with shade, especially in tropical and equatorial regions where solar intensity is very high. Training stations can be sheltered to provide protection for both animals and trainers. A shade can be built of concrete; wood; fibreglass; or waterproof, reinforced fabric. Metal shades are not recommended because they tend to become hot, corrode easily, and are noisy when it rains.

The intensity of light combined with the colour of the pool walls can have an adverse effect on animals when they have to swim from indoor to outdoor pools. The colour of a pool therefore will be selected with consideration to the colour of the coating and the volume of water. The deeper the pool, the darker the overall colour will become. Light cream coating colour will create a light, "blue lagoon" effect. A grey concrete colour will give the water a deep blue hue. The more yellow is incorporated, the greener the effect.

Special Features and Equipment
The various features suggested by surveyed facilities or observed in several visited oceanaria that are presented below can be incorporated in the design of a facility to enhance monitoring of the animals, behavioural observation, training, and medical care. Research also plays an important role in effective conservation programmes and can be facilitated by adequate equipment and pool design (Klinowska & Brown, 1986; Sweeney & Samansky, 1995; see also Chapter 8):

- Above and underwater observation areas or observation room(s) with underwater windows that are inaccessible to the public
- Computer-connected hydrophones and speakers for listening and producing sounds under water for research
- Cables and power points for audio-video recording systems, computers, and communication systems (e.g., telephone, Internet, etc.); also need a communication line that links the below-water room with pool level
- Waterproof electric sockets and accessible grooves for concealing cables
- Waterproof shelter for equipment, such as computers and ultrasound machines, installed near the water
- Equipment and supplies storage space
- Laboratories (medical, audio-video, data lab, or any other research-specific equipment)

Figure 5.42. Media laboratory

Channels and Gates

Openings are cut between pools and in partitions to allow animals to move from one pool or enclosure to another. Wide openings are important since dolphins are reluctant to swim through narrow pathways. They will be designed to allow a dolphin or a whale to swim through comfortably without feeling scared or restrained.

Channels
Channels should be avoided in the design of pools. Some facilities have dealt with them without much trouble; however, a large majority have reported problems associated with channels. Animals would not "gate" or swim through the channel; would get injured during a struggle in this restricted space; or, in the case of calves, would require months before daring to swim across. Channels are sometimes unavoidable to connect existing pools to a new one. In this case, odd angles should be avoided; and they should be as wide, deep, and short as possible—minimum 2.0-m wide, 1.5-m deep, and preferably no longer than 1.2 m for bottlenose dolphins. No large structure should cover a channel because animals, especially calves, may be reluctant to swim underneath it. The survey also indicated that dark colours in a channel or a pool bottom can make it more difficult to gate animals than lighter colour bottoms.

A tunnel is a feature that must absolutely be avoided in a dolphin pool. Animals are usually very reluctant to swim through tunnels. They can be dangerous because dolphins can suffocate if trapped.

Gates
Gates are used to close openings for separation purposes or to drain a pool. Dolphins and whales have to be trained to "gate," to overcome their reluctance. It can take from one week to six months to train an animal to swim through a gate or a channel, depending on the species, individual, and pool design. The minimum recommended width is 1.5 times the distance between the tip of extended flippers of the largest animal housed in the pool.

Two kinds of gates are used for connecting or isolating pools and enclosures: (1) watertight and (2) non-watertight. The two predominant aspects in the design of a gate are the efficiency of its operating mechanism and its safety.

Gates can slide horizontally or vertically, or swing sideways or up and down. It can be operated manually, with or without a winch or ratchet mechanism, electrically, pneumatically, or hydraulically.

It is very important for safety reasons that all automatic gates have mechanisms to prevent entrapment or the crushing of animals between gates and walls or the pool bottom. They should have locks or mechanisms to prevent accidental or intentional opening or closing by wave action or animal play. It is recommended that the operating mechanism be inspected on a regular schedule. Check that gates are not making a "clanging" noise under water.

Watertight Gates—Watertight gates are full solid panels made of metal, fibreglass, PVC, painted or plastic coated wood, or composite panels. They are used to separate two water bodies (e.g., isolation of medical pool) and to allow a pool to be emptied while the adjacent one remains full. Watertight gates are occasionally needed, especially if a pool is used for quarantine purposes or needs to have repairs.

Watertight gates are used mainly in concrete pools since semi-natural enclosures rarely allow changes of water levels. They are much heavier than non-watertight gates. Hence, they require several people to operate them manually unless they are equipped with a winch or ratchet mechanism. They are easier to operate with a powered mechanism, and their design is very similar to flood or sluice gates.

Watertight gates can slide horizontally or vertically in built-in grooves. For a horizontal sliding gate, a recess is provided within the partition or the pool deck to accommodate the gate. Because the gate is submerged in the water, algae and dirt may accumulate in the sliding mechanism and affect its operation. It is therefore recommended to service and clean the gate regularly to avoid a system breakdown.

A vertical sliding gate is either put in place manually or is operated by a crane. It also can be installed on a frame and lowered into the water by a powered hoist, a ratchet mechanism, or a pulley mechanism equipped with a counterweight. This is called a "guillotine" gate. It is a convenient system, but it is aesthetically unpleasant. It also requires a lot of maintenance because of corrosion. The overhang may also cause problems in gating the animals. Hinged or swinging watertight gates exist, but they were not documented nor reported in the survey. They are designed using the same principles as flood gates, with ratchet mechanisms and very sturdy hinges.

The material used is also important in the design of a watertight gate. Some facilities have reported that fibreglass or plastic-coated wood panels, kept in place by wedges, would pop out after they started emptying the pool. A system to secure the gate in place can be installed to prevent this kind of incident.

All types of watertight gates are heavy and fairly slow to operate. Pneumatic systems are probably the most widely used and reliable mechanisms for gate operation. Because of their slow movement and inertia, watertight gates may represent a potential danger to the animals. Therefore, trainers operating the gate should be able to see the opening at all times to prevent an animal from being trapped. The operating system should be equipped with a device that reopens the gate if it detects resistance while closing. A manual override also should be available as a back-up.

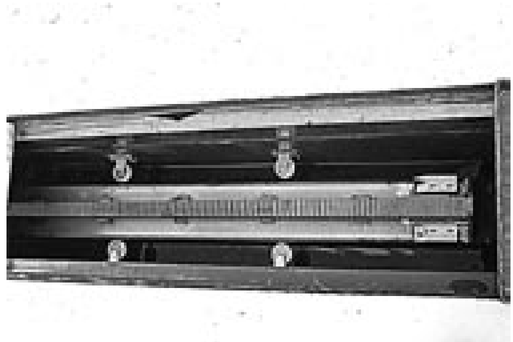

Figure 5.43. Horizontal sliding waterproof gate

Figure 5.44. Vertical sliding waterproof gate

Non-Watertight Gates—Non-watertight gates are used to separate or isolate animals for training and show purposes or for medical reasons. They are usually bars or mesh made of stainless steel, aluminium, fibreglass, PVC, or chain link that are mounted on a metal or plastic frame.

Gates are used frequently in dolphin pools and should therefore be easy to operate and safe for the animals, as well as the trainers. A gate will be designed so that it can be operated by one person. Non-watertight gates are lighter weight and animals sometimes learn to use their rostrum to lift the gate if it is not properly secured.

Manually operated gates can slide vertically, horizontally, sideways, or up and down. Surveyed facilities made the following comments and suggestions regarding manual gates:

- If lifted manually, the gate has to be very light; therefore, PVC bars or chain link mounted on stainless steel frames are recommended. A 34-kg gate, as reported by a surveyed facility, is too heavy for one person to lift manually; therefore, it can only be operated by the strongest trainers or require several persons. A 10-kg gate is the maximum weight recommended. Although aluminium is much lighter than stainless steel, it is not as resistant to strain and corrosion; therefore, it is not recommended by facilities. All other types of gates can be made of stainless steel as it is the most durable and resistant material.
- Avoid covering swing gates with fine mesh because it creates a strong drag that makes them difficult to operate.
- Avoid swinging gates with a horizontal rotation axis; they tend to encourage dolphins to jump over closing gates at the last moment.
- Avoid nets stretched on a frame. They may make the gate light, but they represent a danger for animals (see "Partitions and Fences" section).

Some gates used in large whale pools are too large and heavy to be operated manually. They require a powered system, allowing effortless and remote control. A manual override should always be available as a back-up. Like watertight gates, they can slide horizontally, vertically, or swing sideways, and the most common operating system is pneumatic. Those gates are quite slow, usually allowing plenty of time for the animals to swim through; however, the survey reported that some dolphins, waiting for the last moment to go through, got trapped in the gate and suffocated, unable to reach the surface to breathe. The two concerned facilities recommended to have a control panel near the gate to operate it while watching the animals and the opening. The operating system will be equipped with a device that reopens the gate if it detects resistance while closing. If the gate is covered by a bridge or a catwalk, the opening should remain visible.

To secure a gate, the lock or latch will be easy to open and close. Doors need to be secured when open or closed to avoid reopening or closing by accident or when animals are playing with them; avoid bolts, padlocks, and clasps. It is important to design gates, handles, and locks to ensure animal and trainer safety (i.e., no protruding handles, wide slits, or grooves for sliding gates in the middle of a narrow walkway). Trainers should not have obstacles in their way, especially when carrying heavy buckets of fish or expensive equipment. The operation of gates should not require straining or perilous contortions.

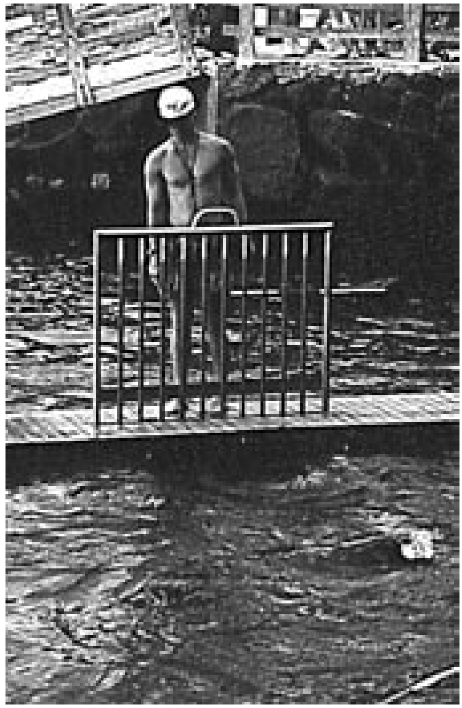

Figure 5.45. Manually operated vertical sliding gate

Figure 5.46. Rotating pneumatic gate with horizontal axis

Bridges and Walkways

Various types of walkways can be proposed to cross over a gate or a channel. Walkways will be independent from the gate to allow the passage from one side of the pool to the other when the gate is open. Walkways need to be large enough for trainers to walk safely. The minimum recommended width is 0.8 m, or 0.9 m if there is a

Figure 5.47. Rotating pneumatic gate with vertical axis

slit for lifting a gate. If a heavier structure like a bridge is built, it will be at least 0.8 m above the surface of the water. They should be much higher for large whales, especially male killer whales, because of the height of their dorsal fin. Several facilities reported recurrent problems with dolphins, especially calves, refusing to gate when there is a structure over the surface of the water. Therefore, the design of such structures should be as light as possible and high above the water level. The principle is to avoid designing tunnel-like openings.

Other Architectural Features and Specifications

Underwater Windows

Underwater viewing windows are a recommended additional feature in a dolphin exhibit and an essential one in a research facility (Asper, 1982; Ellis, 1995; Sweeney & Samansky, 1995). Seeing the animals under water, gracefully swimming and interacting, allows the visitor to become part of the dolphin environment and gives them a better view than from the surface. If the facility's programme allows people in the water with a mask, swimmers will be able to see the animal properly, but only for a brief time. Underwater windows allow prolonged observations without disturbing the animals. They also allow trainers, veterinarians, and scientists to observe behaviour and hierarchical relationships, and to monitor births and research experiments.

Although glass is more resistant to scratches and punctures than acrylic, acrylic allows for a much greater variety of dimensions and shapes. It cannot be used in pools displaying clawed marine mammals, like sea otters and polar bears, but is very commonly used in the display of cetaceans. Acrylic panels can be flat, curved, or hemispherical, and can be assembled without visible joints to make windows of very large dimensions or in shapes such as tunnels through which visitors walk. Windows can be installed fully or only partially under the surface of the water.

Great care will be given to the design and installation of the frame and joints to prevent cracks and

Figure 5.48. Surface window

Figure 5.49. Panoramic windows

Figure 5.50. One piece panoramic window

Figure 5.51. Acrylic tunnel

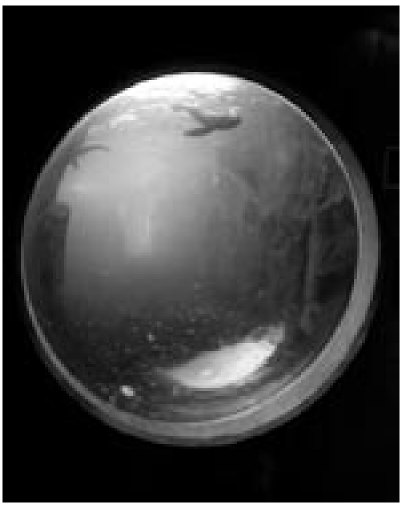

Figure 5.52. Hemispherical acrylic window

leakage. The surface exposed to seawater will have to be cleaned frequently to remove algae. It can be cleaned by divers or a mechanical cleaning device. If a portion of the window is above water level, traces of salt can be removed regularly with soft brushes or sponges.

A large majority of facilities have expressed the wish to have an observation chamber with viewing windows, off-limits to the public, which allows 24-h observations of the animals. This room can be equipped with hydrophones and an audio-video recording system. It is especially useful for monitoring pregnant and parturient females, and newborn calves.

Levels and Access

To achieve easy and efficient circulation around the pools and enclosures for trainers and guests, level changes need to be as gentle as possible. Ramps of proper dimensions, made of no-slip materials, will provide access for wheelchairs, carts, and trolleys carrying fish or equipment.

Every part of the enclosure should be easily accessible to the staff at all times. Avoid having trap doors, ladders, and catwalks as the access the filtration plant room, power switchboards, and food storage.

The food preparation and storage rooms preferably will be at the same level as the pool. Road access to the main freezer allows unloading of frozen fish directly from the truck.

All areas accessible to visitors should comply with regulations for the disabled.

Lighting

Underwater and aerial lighting will be designed to prevent the exposure of animals to excessive or insufficient illumination, which could disturb their behaviour and physiology.

Underwater lighting in pools is optional, but it can be useful for training and observation, in emergency situations, and especially in facilities located in high latitudes during the winter's early nightfall. It can be installed on dimmers to progressively reduce the light intensity. It will be used only occasionally and will be switched off early in the evening. In no case should it be left on all night as it might disturb the animals' diel cycle. The survey recommended that banquets and social events requiring bright illumination and generating loud noise be avoided near the pools because they may disturb and stress the animals. At no time should the lighting be such that it causes the animals discomfort or trauma (Anonymous, 1979-1984, 1995).

In both natural and artificial facilities, aerial lighting is important to ensure the safety of trainers and keepers at nighttime around the pools. Pole lights, bollards, or spotlights will provide uniform and minimal lighting. Mercury vapours and metal halide lamps provide cold spectrum light; sodium or incandescent lamps provide warm colour light. Lights should not be located directly over the pools because they are difficult to access and can shatter in the water.

Acoustics

Dolphins and whales in the wild live in a fairly noisy environment. Creatures like snapping shrimps produce a broadband background noise, and mysticete whales can produce low-frequency sounds as loud as 195 dB (dB re 1μPa at 1 m) under water. In addition, the sounds of crashing waves, precipitation, and seismic rumblings also produce loud, low-frequency, background noise. The auditory system of cetaceans is adapted to these natural sounds. Even so, there is a growing concern that anthropogenic, or human-made noises, such as shipping, seismic exploration for oil or gas, various sonars, and recreational vessels, could adversely affect cetaceans or even cause auditory trauma (see “Audition” in Chapter 2). In captivity, cetaceans cannot escape acoustical interferences, and these can have a dramatic effect on the physiology and behaviour of captive animals (Stoskopf & Gibbons, 1994). Water circulation system, filtration plants, and architectural elements in their environment can create vibrations; low-frequency, mechanical noise; and high-pitch noise that may affect them. Enclosed pools are notorious for their extreme and distorted resonance. It is of great importance to baffle any noise-generating activities (Anonymous, 1992). Noise from even distant construction activities is transmitted with little attenuation through the ground and into pools. The survey indicated that cetaceans seem to be very distressed by drilling activities, explosions, and pile driving activities. Under these conditions, some animals may fail or refuse to perform or feed. Geraci (1986) reported the case of two juvenile dolphins refusing to feed in the presence of the resounding noise from steel work on their enclosure. After weeks of construction activity, the animals became emaciated and eventually died of resulting disease. Cetaceans should not be exposed to loud background or sudden noises, which may be distressing or harmful. In the design of any new marine mammal facility, consideration must be given to its acoustic characteristics with the intention of avoiding a level or quality of sound likely to cause stress or discomfort to the animals. Minimising subaquatic noise should be adequately addressed in the planning of any new facility, with precise operating recommendations given to contractors. In addition, the underwater acoustic environment will be monitored periodically in case new unexpected noises develop.

Irregular pool shapes with varied topography and natural materials, like sand and stones, provide a much quieter environment than artificial and geometrical pools with reflective concrete walls. Sound bounces back repeatedly on parallel walls and can even cause standing waves. It is therefore recommended that the walls of the pools be slanted at about a 10º angle towards the outside of the pool to achieve better acoustic quality.

Floating platforms, partitions, gates, and loose elements in the water, such as latches, might clang repeatedly due to water movement. They will be carefully padded with rubber joints.

Filtration plants and pumps can create loud, high-pitched background noise potentially annoying for dolphins and whales. Filtration plants are frequently located adjacent to the pools in artificial facilities. I recommend that they be located away from the pools. If they are located on concrete platforms, they can be mounted on vibration pads. Outdoor concrete platforms can be isolated from the pools with planted soil buffer areas that absorb vibrations. Noise and vibrations also are conducted into the pools through pipes. Noisy pumps can be replaced with quieter ones, and pipes and all elements connected to the filtration plant can be insulated.

Natural enclosures should not be located near marinas, harbours, or shipyards. Shipping, dredging, and drilling activities create loud noise within the auditory frequency range of many odontocetes and can be harmful to them (Richardson et al., 1995).

Captive cetaceans lose hearing as they age (Ketten, 1998); this is a natural condition called presbycusis. Long-term exposure to noise might create stressful conditions and even damage the hearing of captive animals.

A quiet aerial environment also is recommended for both animals and staff, and the proximity of noisy equipment and activity should be avoided. The noise associated with trucks, forklifts, and machinery near pools can be limited.

Indoor Facilities

Dolphin and whale exhibits frequently are located indoors, especially in cold countries or where winter conditions are severe.

At present, indoor facilities imply artificial environments. No indoor natural or semi-natural facility exists at this time, with the partial exception of one facility using natural granite as part of the pool. With the development of new construction techniques and the preference of the public for more natural environments, semi-natural facilities can be considered a better choice than artificial ones. Most of the following suggestions apply to both types of environments.

Location of Pools and Building Structure

One of the drawbacks of an indoor facility is the dimensions of the roof span, which is limited by the support structure. In turn, the limitations of the roof span restrict the dimensions of the pools. With innovations in materials and structural design, it is now possible to build roof beams with very large spans without the support of columns, therefore allowing new exhibits to be of similar dimensions as some outdoor pools, and making the re-creation of semi-natural habitats indoors possible.

Pools and immediate surroundings should be free of structural piles. Piles create obstacles and can be dangerous for animals and staff. I strongly recommend to build pools at ground level. Artificial pools can be excavated or built above ground. Semi-natural lagoons have to be excavated. The indoor situation of a pool does not limit the incorporation of free form and naturalistic elements in the design. The indoor environment already reinforces the sense of artifice, therefore efforts to create a more natural environment for the benefit of the animals and the pleasure of the visitors are important.

As observed in several existing facilities, pools located in the upper levels of a multi-story building tend to be much smaller and shallower than those located on ground level because of the structural limitations due to the weight of the water. They usually have one or several structural piles in the pool. Upper levels create problems in animal transport, as well as causing an inconvenient environment for the staff. Thus, the construction of cetacean pools in upper levels of buildings should be avoided.

It is important to provide an unobstructed air space above the water surface to a height appropriate to the leaping capacity of the species kept there (Anonymous, 1992). A minimum clear height of 6 m above water level is recommended. The surface above the pool will be clear of overhangs, balconies, and mezzanines. The surface lining of the roof and all areas adjacent to the pools should be constructed of materials that will not shed particulate or toxic material into the water.

Every space, stage area, pool deck, platform, etc., that is accessed by the staff should have a minimum standing room of 2.2 m. I recommend that there be a clear height above the stage of at least 3 m. The stage has to be easily accessible. A ramp with a no-slip finish is preferred over steps.

Incorporating an outdoor pool in the design of new indoor facilities is desirable. To allow the animals to use the outdoor environment during suitable climatic conditions, this outdoor pool will be connected to indoor pools by a short and wide channel, with a large sliding or removable panels of the same width as the channel and located in the wall above to prevent the impression of a tunnel. During cold months, the indoor pool can be isolated from the outdoor pool by a watertight gate and insulating wall panels. This combination of indoor and outdoor pools allows the animals to experience natural sunlight, fresh air, and the outdoor environment, all of which are beneficial to the animals' health and are enjoyable to visitors. Animals should not be introduced to an outdoor pool if the difference in air temperature between the inside and the outside is too great. Water temperature will be similar in both indoor and outdoor pools.

Temperature

The air and water temperature in indoor facilities will be sufficiently regulated by heating or cooling to protect captive cetaceans from temperature

Figure 5.53. Indoor artificial show pool

Figure 5.54. Indoor pool with natural rock and large roof span

Figure 5.55. Outdoor-indoor crane

extremes. The air temperature around the pools will be maintained between 10° and 28º C and should not be subject to rapid changes (Anonymous, 1992). Lower temperatures are required for polar species and preferably will not exceed 20° C. Water temperature in the pool should remain within the range suitable for each species (see Cetacean Species Information Table in Chapter 2). In maintaining captive cetaceans, it is essential to know and provide for their thermoregulatory characteristics and capacities. These capabilities are behaviourally as well as physiologically controlled by the animals (Anonymous, 1992).

Rescued cetaceans should be maintained under conditions that approximate those from which they were removed, and acclimatisation to other conditions should be achieved gradually. Following its initial adaptation, an animal can then be further adapted to other temperatures with provision for behavioural and physiological thermoregulation. Rapid changes in ambient temperature will be avoided, and all changes will be effected cautiously and carefully (Anonymous, 1992). Most surveyed facilities record the air and water temperature twice daily.

Ventilation and Lighting

Indoor facilities have to be ventilated by natural or artificial means to provide an adequate supply of oxygen for the animals and staff; to prevent the accumulation of potentially noxious gases and unpleasant odours; to dilute airborne pathogens; and to reduce saltwater humidity, which is corrosive (Amundin, 1986; Asper, 1982; Geraci, 1986). Queensland's regulation (Anonymous, 1992) recommend a minimum of 10 air changes per hour for air-conditioned areas and 20 air changes per hour for areas otherwise ventilated. At least two intake and outlet points will be provided, and their location carefully selected, to create appropriate air movement and to eliminate pockets of still air. It is highly recommended that a ceiling with adjustable openings above the surface of the pool be designed to provide additional natural ventilation and daylight. Several facilities in the survey expressed a need for more ventilation and natural light, mentioning that a roof that could be open would be a very useful and convenient feature.

Lighting recommendations for outdoor pools also are applicable to indoor pools. Indoor facilities should have ample natural and artificial illumination of a quality (e.g., intensity and spectral composition) and distribution appropriate to the species involved. The lighting level will be sufficient to provide uniform illumination across the pool and designed to prevent the animals from being exposed to excessive or insufficient illumination (Anonymous, 1992).

Quality, duration, and intensity of lighting exposure will ensure that normal physiological and behavioural functions are minimally disturbed, and approximate the lighting conditions encountered by the animals in their natural environment. At no time should the lighting cause discomfort or trauma, which would be visible by behavioural or physiological changes. Lighting will try to recreate as closely as possible the photoperiod, the photospectrum, and the photointensity of natural outdoor sunlight, but at no time cause discomfort or trauma, which would be visible by behavioral or physiological changes. Geraci (1986) suggested that "Natural spectrum" fluorescent lamps and automatic time switches be coupled to outdoor ambient light-sensitive photocells to replicate outdoor light conditions. The lighting system can be equipped with dimming devices to allow a progressive reduction and amplification of the light intensity.

All pools housing cetaceans should receive natural daylight. Ample window openings and skylights will be provided around and above the pools to allow a maximum penetration of daylight (Asper, 1982).

Acoustics

Acoustic recommendations for outdoor pools are also applicable to indoor pools (see earlier "Acoustics" section).

It is difficult to achieve a good acoustical environment inside a building containing large pools of water and areas of flat, hard surfaces. Many facilities complained about the poor acoustic quality of their buildings. They mostly complained about excessive resonance and echo. It seems that designers pay little attention to the acoustic environment of dolphinaria, considering the extraordinary acoustic quality they can achieve in auditoriums and theatres. Most indoor display facilities include a grandstand or theatre to allow the public to enjoy dolphin performances. They will be designed to minimize the echo by incorporating sound traps, and walls can be lined with sound-absorbing materials, which can be selected for their noise reduction capabilities as well as their ability to resist humidity and mould. Backstage areas also will be treated in the same way. Walls and floors adjacent to the pools will be lined with waterproof no-slip materials. Walls above the splash zone, as well as the ceiling in this area, will be lined with water resistant and sound absorbent materials.

Security and Emergency Protocols

Security

Establishments keeping, or seeking permission to keep, cetaceans must have a perimeter fence and security measures available to secure their premises at all times from unwanted intrusion by humans or animals (Anonymous, 1979-1984, 1995; 1992). To ensure that animals are safe and equipment and facilities are secure, a member of the staff preferably will be present at all times when the public has access to the pools. A patroling guard will ensure that no member of the public is found loitering in or near the pools after hours. The guard should have the contact numbers of all members of the curatorial team in case of emergency. Areas that are off-limits to the public will be fenced-off, with signs to inform people about restrictions and potential dangers.

Emergency Protocols
Little is known about the way dolphins protect themselves during violent storms and cyclones in the wild. In controlled environments, they cannot escape a storm by traveling away or seeking refuge in protected bays. Some facilities have been severely damaged and animals injured during extreme climatic events in the USA, Honduras, the Bahamas, Bermuda, France, and Japan by tents, roofs, and building elements collapsing in the pools or by the wave action. Even if the animals do not get injured, they may be highly stressed by the event itself and the following repair work. At the approach of a violent storm or cyclone, it is important to secure every mobile element and check all fixtures. An indoor pool should be provided to shelter animals temporarily in facilities located in regions subject to extreme weather conditions. In natural facilities that are subject to cyclones, tidal waves, or earthquakes, an emergency protocol will be developed to transfer the animals to a safer place or to temporarily release them. Several tropical facilities housing bottlenose dolphins have contingency plans to release resident dolphins in case of a hurricane, so the animals will not be hurt by fences or collapsing platforms. Terrell (1997) recommends that animals be tagged with microchips, replacing cryogenic markings, which allow them to be tracked and identified when recaptured.

Emergency protocols also should be established in case of equipment breakdowns, such as the malfunction of pumps, filtration system, or major power failure, as well as severe pollution or virus outbreaks. Such situations are potentially harmful to the animals and disruptive for visitors if not dealt with promptly and efficiently.

A simple event can generate dramatic consequences. Power failure for a freezer can spoil weeks or months worth of fish supplies. Apart from the financial loss, animals' health can be jeopardised by food poisoning if the problem goes undetected. A failure in the pumping system can slow down the water turnover rate or increase the temperature and bacterial load, leading to harmful consequences for the animals.

Alarm systems should be installed on all control panels to detect and signal failures. Backup generators will be available in case of power failure. Backup equipment that can assume the functions of adequate temperature control, ventilation, lighting, water flow, and water filtration must be maintained in good working order in case of primary system failure or while repairs are being undertaken (Anonymous, 1992).

Emergency protocols for the evacuation and temporary relocation of animals will be established with other nearby facilities when possible.

Support Facilities

Support facilities are essential to the operation of all institutions. Their convenience and quality of design ensure the comfort of the staff, a high level of hygiene, and maintenance.

Staff Quarters
Adequate staff quarters include wet and dry spaces. The wet area, located immediately at the entrance, is composed of a changing room, lockers, showers, and washbasins, in such numbers that it promotes high personal hygiene and complies with regulations. Staff can enter with swimsuits or wetsuits. Floors and walls will be covered with no-slip waterproof finishes, easily sanitised to prevent mould and fungal growth. Appropriate drainage will be provided. A space should be reserved to hang wet suits and store snorkelling gear.

It also is advised that an outside shower and hose located near the entrance of the indoor wet area be installed to allow the rinsing of wetsuits and equipment.

A laundry room with washing machines and dryers will be provided and preferably will be located near the changing room.

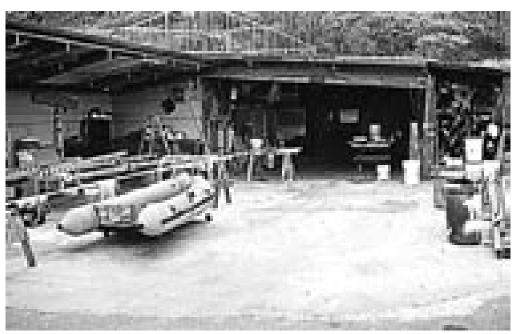

Figure 5.56. Workshop

Dry office space will be provided to hold meetings; record daily behavioural, medical, and husbandry data on the animals; and rest between training, research sessions, educational programmes or shows. I recommend to equip this area with a pantry for microwave snacks. Staff rooms ideally will have access to the outside and windows to the pools, allowing observation of the animals. A space heater or air conditioner can be available if needed, and a footbath is recommended at every outside doorstep.

Workshop and Diveshop

A workshop (fully or partially indoors) will be installed to store spare parts, pipes, and tools; and for the repair of small vehicles, boats, toys, and training and scientific equipment. It preferably will not be located close to the pools as repair work can be noisy and dirty. Diving gear can be stored in a dedicated room close to the pool where it is most frequently used. It is suggested to use compressors far away from pools because they are noisy, and that trolleys be used to carry full tanks.

Literature Cited

Allen, M. E. (1996). Introduction: Basic husbandry. In D. G. Kleiman, M. E. Allen, K. V. Thompson, S. Lumpkin, & H. Harris (Eds.), *Wild mammals in captivity: Principles and techniques* (pp. 1-3). Chicago: The University of Chicago Press.

Amundin, M. (1986). Breeding the bottle-nosed dolphin *Tursiops truncatus* at the Kolmarden Dolphinarium. *International Zoo Yearbook*, *24/25*, 263-271.

Anonymous. (1979-1984, 1995). *Specifications for the humane handling, care, treatment and transportation of marine mammals* (9 CFR, Subpart E, §3.100-118). Washington, DC: U.S. Department of Agriculture, Animal and Plant Health Inspection Service.

Anonymous. (1992). *Guidelines for the care, holding and transport of marine mammals in Queensland* (Draft). Brisbone: Queensland Department of Primary Industries (QDPI). 39 pp.

Anonymous. (1995). *Environment Act* (SN 1995 c E-13.1, Sections 9 to 11). St. John's, NL: Government of Newfoundland and Labrador, Department of Environment and Labour.

Asper, E. D. (1982). Design features of marine mammal exhibits. In K. Sausman (Ed.), *Zoological parks and aquarium fundamentals* (pp. 99-104). Wheeling, WV: American Association of Zoological Parks and Aquariums.

Bassos, M. K., & Wells, R. (1996). Effect of pool features on the behavior of two bottlenose dolphins. *Marine Mammal Science*, *12*(2), 321-324.

DeMaster, D. P., & Drevenak, J. K. (1988). Survivorship patterns in three species of captive cetaceans. *Marine Mammal Science*, *4*(4), 297-311.

Doherty, J. G., & Gibbons, E. F. (1994). Managing naturalistic animal environments in captivity. In E. F. Gibbons, E. J. Wyers, E. Waters, & E. W. Menzel (Eds.), *Naturalistic environments in captivity for animal behavior research* (pp. 126-139). Albany: State University of New York Press.

Ellis, S. (1995). Marine mammal behavior: Conservation through research. In E. F. Gibbons, Jr., B. S. Durrant, & J. Demarest (Eds.), *Conservation of endangered species in captivity: An interdisciplinary approach* (pp 441-463). Albany: State University of New York Press.

Geraci, J. R. (1986). Marine mammals: Husbandry. In M. E. Fowler (Ed.), *Zoo & wild animal medicine* (pp. 757-760). Philadelphia: W. B. Saunders Company.

Gibbons, E. F., Wyers, E. J., Waters, E., & Menzel, E. W. (1994). Naturalistic environments in captivity for animal behavior research: Synthesis statement. In E. F. Gibbons, E. J. Wyers, E. Waters, & E. W. Menzel (Eds.), *Naturalistic environments in captivity for animal behavior research* (pp. 283-288). Albany: State University of New York Press.

Kannan, K., Senthilkumar, K., Loganathan, B. G., Odell, D. K., & Tanabe, S. (1997). Elevated accumulation of tributyltin and its breakdown products in bottlenose dolphins (*Tursiops truncatus*) found stranded along the U.S. Atlantic and Gulf Coasts. *Environmental Science and Technology*, *31*(1), 296-301.

Ketten, D. R. (1998). *Marine mammal auditory systems: A summary of audiometric and anatomical data and its implications for underwater acoustic impacts* (NOAA-TM-NMFS-SWFSC-256). Washington, DC: U.S. Department of Commerce, National Oceanic and Atmospheric Administration, National Marine Fisheries Services, Southwest Fisheries Science Center. 97 pp.

Klinowska, M., & Brown S. (1986). *A review of dolphinaria.* London: Department of the Environment. 247 pp.

Krajniak, E. (1988). Potential marine mammal exhibit physical plant failures. *Proceedings of the 16th IMATA Conference*, San Antonio, Texas.

Markowitz, H. (1990). Environmental opportunities and health care for marine mammals. In L. Dierauf (Ed.), *CRC handbook of marine mammal medicine: Health, disease, and rehabilitation* (pp. 483-488). Boston: CRC Press.

Myers, W. A., & Overstrom, N. (1978). The role of daily observation in the husbandry of captive dolphins (*Tursiops truncatus*). *Cetology*, 29.

Richardson, W. J., Greene, C. R., Malme, C. I., & Thomson, D. H. (1995). *Marine mammals and noise.* London: Academic Press. 567 pp.

Shane, S. H., Wells, R. S., & Würsig, B. (1986). Ecology, behavior, and social organisation of the bottlenose dolphin: A review. *Marine Mammal Science*, *2*(1), 34-63.

Small, R. J., & DeMaster, D. P. (1995a). Survival of five species of captive marine mammals. *Marine Mammal Science*, *11*(2), 209-226.

Small, R. J., & DeMaster, D. P. (1995b). Acclimation to captivity: A quantitative estimate based on survival of bottlenose dolphins and California sea lions. *Marine Mammal Science*, *11*(4), 510-519.

Stoskopf, M. K., & Gibbons, E. F. (1994). Quantitative evaluation of the effects of environmental parameters on the physiology, behavior, and health of animals in naturalistic captive environments. In E. F. Gibbons, E. J. Wyers, E. Waters, & E. W. Menzel (Eds.), *Naturalistic environments in captivity for animal behavior research* (pp. 140-160). Albany: State University of New York Press.

Sweeney, J. C. (1990). Marine mammal behavioral diagnostics. In L. Dierauf (Ed.), *CRC handbook of marine mammal medicine: Health, disease, and rehabilitation* (pp. 53-72). Boston: CRC Press.

Sweeney, J. C. (1993). Dolphin Quest protocol for veterinary health care management of *Tursiops & Steno*. San Diego: Dolphin Quest.

Sweeney, J. C., & Samansky, T. (1995). Elements of successful facility design: Marine mammals. In E. F. Gibbons, Jr., B. S. Durrant, & J. Demarest (Eds.), *Conservation of endangered species in captivity: An interdisciplinary approach* (pp. 465-477). Albany: State University of New York Press.

Terrell, K. (1997). Microchipping in Atlantic bottlenose dolphins (*Tursiops truncatus*) for positive identification using Avid microchip I.D. system. *Proceedings of the 25th IMATA Conference*, Baltimore, Maryland.

Aquatic Mammals 2005, *31*(3), 351-363, DOI 10.1578/AM.31.3.2005.351

6. Life Support Systems

Because cetaceans spend their entire life in the water, its quality is one of the most important aspects of their environment. Water quality for marine mammals is not always "visible," contrary to Cornell's (1982) statement that, in general, water that looks clean is clean. Good water quality not only means clean water, it also means adequate temperature according to the species housed, stable and appropriate pH level and salinity, and low bacterial count, among many other considerations.

The water quality in a controlled environment relies almost entirely on the quality of the life support system (LSS), but also on the ecological interactions within the pool. Maintaining a clean and healthy environment is essential because animals are prevented from swimming away from any situation that is unsettling or harmful (Manton, 1986). Animal health concerns should clearly take precedence over the aesthetic quality of an exhibit; however, ideally the two should be complementary. Facilities should make proper assessment of the cost of building and operating an adequate LSS (Boness, 1996; Doherty & Gibbons, 1994). Various systems to maintain a balanced aquatic environment have to be present. Often, the appropriate systems are determined by the distance of the facility to the seashore, the quality of available water, and the species kept.

The aim of this chapter is to give an overview of various systems without getting into intricate detail. Creating and maintaining a water system of high quality is a difficult process that should be supervised by LSS engineers and biologists in collaboration with the institution's curatorial team. It is important that the people in charge of maintaining the quality of their animals' environment on a daily basis and at various levels know the general principles.

Water Systems

Open-Ocean

The aquatic environment for cetaceans in natural facilities is the ocean; it is freshwater for river species. The choice of a site for a sea-pen type facility is critical since water quality and climatic events cannot be controlled. This system offers great advantages because it can sustain other living organisms, such as crustaceans, invertebrates, sea grass, and algae, thus offering cetaceans a greater variety of interactions within their environment. It also is financially advantageous because it does not require a filtration system.

The main disadvantage of natural facilities is the lack of protection from weather hazards and pollution. Pollution can be biological (e.g., red tides), physical (e.g., debris), chemical (e.g., oil spills), or acoustical (e.g., shipping or construction activities). Floating booms can be installed at the entrance of the main channel or around fences or pens to contain drifting oil, debris, and waste, but a contingency plan will be set up to evacuate animals to another facility in case of severe pollution or if there is biological pollution that cannot be prevented. Acoustical pollution can be avoided by selecting quiet spots for the installation of the facility. Proximity to harbours, marinas, and shipyards will be avoided. Toxic butyltin compounds, including mono-(MBT), di-(DBT), and tributyltin (TBT), which are used in ship antifouling paints to prevent barnacles and algae from attaching to hulls, may contribute to suppress an animal's ability to fight infection, especially in the liver, kidney, heart, and brain. Such paints also can be found on pier piles and harbours, and the installation of dolphin facilities in their vicinity should be avoided. A study on the effects of TBT on bottlenose dolphins in U.S. coastal waters concluded that it is a "very serious" problem that may pose a greater risk to dolphins than PCBs (polychlorinated biphenyls) (Kannan et al., 1997).

Ideally, a natural facility will be located where flushing by tidal or wave action ensures an appropriate turnover of water and elimination of animal waste. Regular checks, at least once a day, help locate and eliminate potentially harmful debris in the water such as plastic bags, pieces of nets, hooks, ropes, or driftwood. These can be swallowed by or become entangled with a dolphin's limb, causing harmful or lethal injuries. All foreign objects should be considered a potential health risk to the animals (Sweeney, 1990).

Open Systems

Facilities located near the sea, where water quality is good and has adequate salinity, often use open systems. Although they have the advantage of being economical, as with sea pens, they do not provide protection against pollution unless a primary filtration system is installed. If the water is pumped from a depth of 5 to 10 m, floating oil, most debris, and waste can be avoided.

Open System—Open or flow-through systems do not require mechanical filtration devices. Pumped seawater from the nearby ocean, or tap- or groundwater for freshwater species, flows directly into the pools and is discharged without being recirculated. The exchange is continuous, and the control of temperature, colour, concentration of suspended particles, and bacteria depends on the frequency of the water replacement (Spotte, 1991). Raw seawater, however, often is pumped through a silt-screen to remove the largest particles and therefore reduce turbidity. To obtain very clean water, some facilities, such as two surveyed facilities in Hawaii, pump seawater through natural lava substrate, which acts as a primary filter.

Water is discharged back into the sea, usually at a location far away from the pumping station, or into waste or storm drains if local regulations allow. The quality of the water discharged should comply with regulations and not affect marine organisms living near the discharge point or down current.

Open System with Chlorine—This system is the same as above, with the addition of a small amount of chlorine to limit algal and bacterial growth. If chlorinated water is discharged into natural waters, it can be dechlorinated unless it already complies with regulation levels. Non-dechlorinated water will be discharged away from shore to limit its effect on living organisms.

Semi-Closed and Closed Systems

Facilities located near the sea where water quality is such that it needs to be treated, as well as those facilities located inland, use semi-closed (also called semi-open) or closed systems. Since these systems involve the partial or total recirculation of water, they both require mechanical, chemical, and biological filtration. Both systems use natural or artificial seawater or freshwater (if appropriate).

Semi-Closed System—A semi-closed system relies on the continuous replacement of water lost through evaporation or waste; however, replacement occurs at a lower rate than in open systems. Typically, less than 10% of the total volume is replaced every 24 h (Geraci, 1986; Spotte, 1991). The rest of the water is treated as it is in a closed system.

Closed System—All water is recycled through filters, sterilised, and returned to the pool (Spotte, 1991). On occasion, partial water replacement occurs to replace water lost through evaporation and recycling. (See "Water Purification" section for more details.)

Saltwater and Seawater—There are various kinds of "sea water" in controlled environments. Saltwater is made of tap water, groundwater, or available natural freshwater mixed with marine sodium chloride (NaCl). Artificial seawater can be made from the same sources of freshwater mixed with commercial mixes of sea salts available in bulk from marine supply houses (Faulk, 1990). Artificial seawater usually is recirculated because it is expensive to produce. The composition of artificial seawater has to be very close to natural seawater because the presence of some trace elements, such as copper, zinc, manganese, or potassium, may be essential to the health of the animals (Manton, 1986). It does not have an impact on marine mammals, but it is crucial for keeping fish and invertebrates alive. Tap water or freshwater might have to be filtered before adding salts because the ions or chlorine present in this water may change the composition of the artificial seawater and might jeopardize animal health. Refer to Dierauf (1990) and Faulk (1990) for brand names of ocean salts. Natural seawater delivered by trucks also can be used to fill and top-off pools.

Freshwater—Only four cetacean species can live permanently in freshwater (see Cetacean Species Information Table in Chapter 2). Tap water, groundwater, or freshwater from a nearby water body (e.g., lake, river) can be used in open, semi-closed, or closed systems. If tap water is used, the amount of chlorine in the water should be checked before adding any more. The composition of natural freshwater has to be analysed to ensure its suitability. A high concentration of metal ions and pollutants could make it unfit for use.

Marine cetacean species cannot be kept permanently in freshwater. When necessary, a veterinarian may recommend that a dolphin be kept in freshwater for very short periods of time for rehydration purposes. When this occurs, the LSS preferably will allow the medical pool to be filled with freshwater. The danger is that after some time, a degeneration of epidermal cells will occur. Necrosis and ulceration of the epidermis will result if the animal is not returned to seawater (Manton, 1986).

Water Parameters

Temperature

Water temperature is monitored to ensure the comfort of the animals and that all mechanical systems are operating efficiently. If the water temperature rises or drops slowly but steadily, it can reveal a malfunction in the system (Faulk, 1990).

In natural facilities, water temperature cannot be regulated; therefore, the species housed should be adapted to that type of climate. Similar recommendations are applicable for artificial outdoor facilities. It is not recommended to keep coldwater species in tropical climates or vice versa. Despite this, with the increase in new facilities, cetaceans more frequently are being placed in unnatural climates (Geraci, 1986; survey). Institutions located in tropical regions sometimes display Arctic species, such as beluga whales, in water lacking temperature regulation; this results in apathetic animals. Cetaceans, however, have efficient means of temperature control, using blubber and uniquely adapted vascular mechanisms. Most cetaceans can exceed their natural range, but excessive heat is as life threatening as excessive cold (Geraci, 1986). Therefore, I recommend that the temperature range indicated in the Cetacean Species Information Table be followed, and variations from these given ranges only should be for a brief time and should occur gradually. Chillers and heaters can be added to lower or increase water temperature in semi-closed and closed systems. The system will gradually increase or drop the temperature through successive filtration cycles.

Air temperature is also critical to cetaceans. In most surveyed facilities, air and water temperature are measured twice daily (AM and PM). Handheld thermometers are adequate to monitor the temperature in the tank, but digital wall-mounted thermometers or laser thermometers allow easy and efficient tank temperature readings day or night (Faulk, 1990; survey).

Salinity

Salinity is the measurement of dissolved salts in water. The symbol ‰, parts per thousand (ppt), is commonly used to express salinity (Faulk, 1990). It is almost equivalent to g/l. Measurements of specific gravity are more troublesome because they are correlated to the water temperature. Salinity of seawater differs throughout the world. A natural and safe salinity range is 25 to 35‰ (Geraci, 1986). Although the balance and proportions of salts and ions in seawater is critical to fish and invertebrates, it does not seem crucial for marine mammals, for which osmolarity is the main factor. This is the reason why they survive well in saltwater. The composition of seawater is given in Table 6.1.

Both evaporation and rainwater have an impact on salinity level. In regions with heavy rainfall, it is important to install efficient drainage around the enclosures to limit rainwater from entering the pool (see "Water Flow and Drainage" section). In Mediterranean and desert regions with strong evaporation, salinity will be closely monitored, and freshwater will be added if there is a strong rise in salt concentration. Cetaceans can handle some variations within a safe range. If animals are transferred from one facility to another, however, the salinity level should be close to that of the facility of origin. Saline water assists in buoyancy, as well as certain osmotic processes to which cetaceans have adapted (Faulk, 1990). A sudden change in salinity may reduce or increase the animals' buoyancy and affect their metabolism. A reduction in buoyancy would require the dolphin to spend more energy to stay afloat and therefore unbalance its energy budget. This could have severe consequences for sick and newborn animals (van der Toorn, 1987).

Table 6.1. The composition of natural seawater at 35‰ salinity (Castro & Hubert, 1992)

Chemical constituents	Concentration	% of total salinity
Chloride (Cl)	19.345	55.03
Sodium (Na)	10.752	30.59
Sulfate (SO_4)	2.701	7.68
Magnesium (Mg)	1.295	3.68
Calcium (Ca)	0.416	1.18
Potassium (K)	0.390	1.11
Bicarbonate (HCO_3)	0.145	0.41
Bromide (Br)	0.066	0.19
Borate (H_2BO_3)	0.027	0.08
Strontium (Sr)	0.013	0.04
Fluoride (F)	0.001	0.003
Everything else	<0.001	<0.001

Salinity measurements can be done daily with a refractometer that is temperature-compensated (Faulk, 1990) or by easier-to-use handheld devices measuring conductivity.

pH

pH is the measurement of the acidity or alkalinity of a solution. A value of 7 represents neutrality; lower numbers indicate increasing acidity and higher numbers indicate increasing alkalinity. Each unit of change represents a ten-fold change in acidity or alkalinity. Natural seawater pH is 8.2. If chlorination is used as a disinfectant, its properties are optimal at a lower pH range—between 7.2 and 7.6. A safe range of pH for keeping cetaceans is generally between 7.5 and 8.2 (Geraci, 1986).

pH should be measured daily with a properly calibrated unit (Anonymous, 1979-1984, 1995; Faulk, 1990; Sweeney & Samansky, 1995; survey). Carbonate salts, such as calcium carbonate or sodium hydrogen carbonate, can be added to raise pH, and acids, such as hydrochloric acid, can be added to lower it (Arkush, 2001; Spotte, 1991). If a system cannot be consistently maintained at the proper pH level, it might reflect a stress in the filtration or sterilisation systems. It also can be due to a major leak in the system. Filter materials, bacterial activity, and sterilisation chemicals will then be examined.

Alkalinity

Alkalinity is the quantitative capacity of water to neutralise an acid—that is, the measure of how much acid can be added to a liquid without causing a significant change in pH (Anonymous, 1999).

Natural seawater is buffered, meaning it can resist sudden, large changes in pH. Seawater copes with it through the carbon dioxide buffering system (van der Toorn, 1987). It is also referred to as the "hardness" of water. Biological activity, such as nitrification, can alter pH. Alkalinity control is important when chlorination is the primary means of disinfection as the chlorination removes the alkalinity and makes the pH more acidic, and also when autotrophic denitrification mechanisms are installed.

Alkalinity is expressed in mg/l of equivalent calcium carbonate ($CaCO_3$), or meq/l (milliequivalent per litre, 1 meq/l = 50 mg/l $CaCO_3$). The best levels are between 100 and 150 mg/l. Alkalinity levels can be determined by using test tablets or by titration. The presence of salt and chlorine may affect the accuracy of these pH-sensitive dyes, and the latter might "bleach" out the colour and give false results (Manton, 1986).

Turbidity and Visibility

Turbidity is the amount of small solid particles suspended in water as measured by the amount of scattering and absorption of light. Turbidity blocks light and makes the water opaque. It is influenced by the size, shape, colour, and concentration of suspended particles in the water. Turbidity is measured in Nephelometric Turbidity Units (NTU) (Anonymous, 1999). Water preferably will not exceed 0.5 NTU. Levels > 0.1 NTU, combined with turbulences in the water circulation system, can enhance air entrapment (Fasik, 1991) (see "Water Flow and Drainage" section). Turbidity measurements are usually taken daily. It is measured by a nephelometer, which is an instrument measuring how changes in light scatter compared to a sealed calibration vial (Faulk, 1990).

Turbidity affects transparency and, therefore, visibility. Although high levels of particles do not usually affect the animals, it is not aesthetically appealing to the public. It affects the ability of the trainers to observe the animals, and might also reflect some malfunction in the LSS system. Visibility can be measured in vertical or horizontal distance with a Secchi disk placed in the pool (survey).

Dissolved Oxygen

Dissolved oxygen (DO) refers to oxygen gas that is dissolved in the water. Oxygen enters the water by photosynthesis of aquatic biota and by the transfer of oxygen across the air-water interface. The amount of oxygen that can be held by the water depends on the temperature, salinity, and pressure. Colder water holds more oxygen than warm water does, and freshwater holds more oxygen than does saltwater (Smith, 1990). Even though they are air breathers and therefore do not depend on DO in the water, cetaceans benefit from good water quality that is maintained with the help of acceptable DO levels. Mechanical filters operate more efficiently, and water is less turbid when properly oxygenated (Faulk, 1990).

Ideally, seawater DO levels will be maintained between 5.0 and 8.8 ppm DO (equivalent to mg/l) and testing performed once weekly (Faulk, 1990; survey). Measurements can be made with various methods—the most reliable being the iodometric method. It is time-consuming and difficult to interpret. Care must be taken when collecting the samples for DO measurements; they can neither be left in contact with air, nor agitated—both problems change the gaseous contents of the water. Alteration in temperature also affects results. The membrane electrode method, used in field testing (Anonymous, 1992a), is faster and more convenient provided that the electrodes are well-maintained.

Ammonia, Nitrite, and Nitrate

Ammonia is a product of urinary waste and has to be removed for health and aesthetic reasons. Ammonia levels should be kept below 0.05 mg/l of water (Boness, 1996). Chlorination, ozonation, filtration, and water exchange help reduce ammonia levels. Nitrites and nitrates are the other byproducts of urine breakdown. Nitrite occurs after *Nitrosomonas* bacteria (among other "nitritifier" bacteria) break down ammonia. Nitrite is converted into the less toxic nitrate by *Nitrobacter* bacteria (again, among other "nitratifier" bacteria) (Faulk, 1990; van der Toorn, 1987). Ten mg/l is indicated as a safe drinking water limit by the Water Quality Association. A build-up of nitrate causes a drop in pH level because nitrification produces acids. The side effect of nitrate presence is a build-up of algae. Ammonia, nitrite, and nitrate are not known to be toxic to marine mammals, but they can lead to an unsightly and unhealthy system due to excessive algae (Faulk, 1990). Build-up of nitrate in a system can be avoided by the denitrification of a constant side-stream of the water volume.

Readings of nitrites and nitrates will be taken once daily, and once weekly when the system becomes stable. Ion analysers, with specific ion probes, are efficient and reliable. They have to be calibrated for saltwater (Faulk, 1990). Specific test kits are also available.

Bacteria

Bacterial contaminants, some of which are pathogenic, must be monitored to ensure protection against disease (Faulk, 1990). Bacteria, such as *Streptococcus* sp., *Pseudomonas* sp., and *Pasteurella* sp., among others, have been isolated from fish fed to dolphins. This revealed that food is probably the prime source of potentially harmful bacteria in the biological water treatment system (Overath et al., 1999). *Erysipelothrix rhusiopathiae* is another pathogenic bacterium, which also is presumed to be in food. It causes erysipelas, which can be lethal in its septicemic form (Dunn, 1990; Sweeney, 1993).

The main bacteria that is monitored is coliform bacteria, which can cause water contamination. It can be recognized, though, that pathogens could be present in marine mammal pools when coliforms are low in number or absent (Spotte, 1991). Faecal coliform bacteria, *Escherichia coli*, is released in the water through dolphin faecal matter. A high concentration of *E. coli* can be harmful to animals and indicates an unbalanced sterilisation system or water exchange. Measurements are obtained by the MTF (Multiple Tube Fermentation) method. The concentrations of these bacteria is expressed in MPN (Most Probable Number). Another method called MF (Membrane Filtration) can be used and is expressed in number of colony-forming units/100 ml of water (Spotte, 1991). Coliform count should be performed weekly (*E. coli* is the most significant of the family Enterobacteriacae, which also includes most species of *Citrobacter*, *Enterobacter*, and *Klebsiella*) (Arkush, 2001; Spotte, 1991). Results above 100 MPN require a correction of the sterilisation system or an increase in the changing of water (Sweeney & Samansky, 1995).

A weekly count of total bacteria should be evaluated against the historical norm of the location. If the tests indicate a contamination, they will be repeated and the source identified (Sweeney, 1993).

A water- and soil-borne bacterium, *Burkholderia pseudomallei* (formerly named *Pseudomonas pseudomallei*), endemic to Southeast Asia, but now recognized in humans and animals worldwide, can be found in dolphinarium waters, especially after heavy rains when soil is washed into the pools. It is responsible for a dangerous infectious disease called melioidosis, which is lethal in most cases (Liong et al., 1985; Reeves et al., 1994; Sweeney, 1986). A vaccine is now available for melioidosis (Kennedy-Stoskopf, 1990), but it is still recommended that the presence of this bacterium be monitored closely in regions at risk.

Where pools holding cetaceans have sandy bottoms, regular inspections will be undertaken to ensure that bottom sediments do not become anoxic (Anonymous, 1992b).

Algae

Algae are single-celled (e.g., phytoplankton) or simple multi-celled (e.g., macrophyte or seaweed) organisms commonly found in surface water and produce their own food through photosynthesis. Excessive algal growth may cause the water to have undesirable odours or tastes, and decay of algae can deplete the oxygen in the water (Anonymous, 1999). Algal growth is favoured by warm temperature and daylight Ultra-Violet (UV) rays (Sweeney & Samansky, 1995), and it occurs on all submerged surfaces. It is not dangerous for the dolphins, but it is unsightly—especially in concrete pools. In pools and lagoons made of natural materials, the algae only needs to be removed once in a while. Chlorine, ozone, copper, biological filtration, and UV disinfection can control algae.

The following table presents a summary of water tests that are carried out on a regular basis by many surveyed facilities. Sweeney (1993) recommended keeping records on site for at least three years in a digital format.

Table 6.2. Frequency and targeted range of water quality measurements

Measurement	Frequency	Range
Air temperature	2 x day	N/A
Water temperature	2 x day	see Cetacean Species Information Table
Salinity	Daily	25-35 ‰
pH	Daily	8.2; if chlorination, 7.5-7.6
Turbidity	Daily	≤ 0.5 NTU
Dissolved oxygen	Weekly	5.0-8.8 ppm DO
Coliform bacteria	Weekly	≤ 100 MPN/100 ml; more frequent if > 200 MPN until level drops below 100
Total bacteria	Weekly	see historical norm of location
Ammonia	Weekly	≤ 0.05 mg/l of water
Nitrite		
Nitrate	Weekly	≤ 1 mg/l rapidly converted to nitrate ≤ 10 mg/l
Free chlorine	2-3 x day	≤ 0.2 ppm
Total chlorine		≤ 0.5 ppm
Alkalinity	Weekly	100-150 mg/l

Water Purification

The water purification system removes animal wastes, prevents the growth of microorganisms and the build-up of organic carbon compounds, provides a relatively toxic/chemical-free environment, and maintains some degree of clarity (Geraci, 1986). Purification systems are crucial to the quality of the animals' environment and should all have back-ups and safety procedures in case of emergency, malfunction, or equipment failure. The survey results recommended that medical pools have pumping and purification systems separate from the primary pool system to prevent disease transmission and cross-contamination (Anonymous, 1992b).

With the heavy load of waste matter discharged by whales and dolphins, water needs to be replaced or cleaned very frequently. Mechanical filtration is used to remove particulate matters such as epidermal cells, faecal material, food debris, plant material, and dust. Other soluble byproducts of animal excretions, mainly organic nitrogen products, bacteria, and algae, are removed by chemical processes (e.g., chlorine, ozone, or activated carbon), also referred to as chemical oxidation, disinfection, sterilisation, and absorption (the terms *disinfection* and *sterilisation* often are used interchangeably, but they are not synonyms. *Disinfection* is the selective destruction of infectious organisms; *sterilisation* is the nonselective destruction of all life [Spotte, 1991]). Biological purification uses bacteria to remove undesired organic matters. These terms will be used to explain the various processes of water purification.

Mechanical Filtration

Mechanical filtration can be achieved with various kinds of filters such as gravity sand filters, diatomaceous earth filters, high-rate pressure sand filters, membrane filters, or protein skimmers. Nowadays, pressure sand filters are the most commonly used and can treat large volumes of water; however, they often come in combination with other systems. Gravity and diatomaceous earth filters are no longer optimal systems for marine mammal pool filtration and, therefore, are not recommended in new efficient systems.

Pre-Filter and Drum Filters—Pre-filter baskets are cylinders positioned before the pumps that send the pool flow to the sand filters. They are equipped with a straining basket made of steel mesh no larger than 5 mm, and they are useful in removing large particles, thus preventing unnecessary clogging.

Drum filters are composed of rotating drums that retain particles on a fine membrane. This membrane is washed automatically to eliminate accumulating matter. These filters often are installed as pre-filters. The size of the particles they can filter depends on the mesh of the filtering membrane. These filters are not very reliable because they do not retain the majority of particles, have a fairly small capacity, tend to clog up often, and break down easily. Therefore, they are not recommended for other use than pre-filtration.

Figure 6.1. Pumps and pre-filter baskets (Photograph from I. Smit)

Sand Filters—Sand filters, commonly installed for large volumes, have an average diameter of 2.8 to 3 m, offering a filtration surface between 6 and 7 m^2. Recommended speed of filtration is between 29 and 34 m/h, allowing a volume of filtration between 170 and 240 m^3 per hour per filter. These tanks can be made of fibreglass, polyester, or steel. Steel is recommended for large-volume tanks and for durability, but they have to be equipped with cathodic protection to prevent corrosion. Both tower, or vertical, filters and horizontal bed filters exist. Tower filters prioritise the depth of filtering substrate, while horizontal ones prioritize the filtration surface (survey). Filter beds usually are made of a support substrate and a filtering medium of fine silica sand, ranging in granulometry between 0.5 and 1.6 mm. Finer sand could clog more quickly, and coarser sand would not be efficient in trapping particulate matter down to 30-35 μm. An optional layer of hydroanthracite (a derivative from coal, granulometry 0.8 to 1.6 mm) can help remove additional dead organic matter and reduce turbidity. These filters are backwashed regularly to remove accumulated particles and maintain filtration capacity. This is done after emptying the tank of its water and reversing the flow inside the tank. To increase cleaning efficiency, the substrate can be decompressed and agitated by air scour through the injection of air from below. High-rate pressure sand filters do not sustain a stable microbial population that can act as a biological filter, although some microorganisms might survive backwashes and help to break down dissolved substances in a minimal way. This is why a sterilisation process has to be added to the mechanical filtration.

Flocculation—Flocculation is a useful additional process used in substrate filtration. A flocculant is used to coagulate small particles into bigger ones—the flocs—which will be more efficiently trapped in the filtering substrate and, thus, reduce oxidant demand (Anonymous, 1999; Gregory, 1989; Spotte, 1991). The flocculant most commonly used is aluminium sulphate or alum, but polyaluminium chloride and ferric chloride are also used. The flocculent and water should be mixed properly; thus, a mixing chamber is recommended (Boness, 1996).

Figure 6.2. Steel tower sand filters (Photograph from I. Smit)

Figure 6.3. Horizontal steel filters

Figure 6.4. Filters for large whale pools

Protein Skimmers/Foam Fractionators—Another mechanical system to remove organic matter and reduce nitrogen compounds (e.g., ammonia, nitrite, nitrate)—a protein skimmer or foam fractionator—is now commonly installed in marine mammal facilities. This dispersed-air flotation system usually is combined with an oxidising system; thus, it also could be presented in the "Chemical Filtration" section. The polluted water is mixed with an air/ozone mixture through an injector and pumped into the reaction chamber. Here, vortexing causes turbulence and intimate mixing of the water and air bubbles. Proteins with a lower specific weight than water are the first to flow upwards through the internal riser. Solids become attached to gas bubbles already loaded with proteins and also flow upward. Pollutants are concentrated into foam, which then is skimmed, leaving clean water to flow through a different outlet (Shuran Sea Water Equipment: *Aquaflotor Technical Guide*; van der Toorn, 1987). This system usually is installed as a complement to sand filters and is combined with ozone as an alternative to other means of disinfection or sterilisation.

Figure 6.5. Protein skimmers with ozone injection (Photograph from I. Smit)

Activated Carbon—Activated carbon is among the most effective materials for the physical absorption of organic carbon (Spotte, 1991). They are classified in the paragraph on mechanical filtration because they function on a physical principle of absorption. These filters are made of absorbents manufactured from a number of carbon-based materials. They typically have a 0.5 to 1 µm filtration capability, making it helpful for particulate filtration. Activated carbon commonly is used for dechlorination, for reducing soluble materials such as Dissolved Organic Carbon (DOC)—chloramines issued from chlorination—and to prevent the accumulation of total organic carbon (TOC) precursors of trihalomethanes (THM; mutagenic and carcinogenic compounds present as trace concentrations in chlorinated water and ozonated seawater) in closed systems (Anonymous, 1999; Spotte, 1991). It can be installed on a side-stream after mechanical filtration in combination with various oxidation systems. Activated carbon does not have a long life span and is quite expensive to change. The disposed material has to be treated as hazardous waste (Spotte, 1991).

Chemical Purification

Ridgway (1972) calculated that a dolphin, weighing 136 kg and eating 6.6 kg of fish, passes over 4 l of urine and 1.4 kg of faeces per day. The majority of this waste matter is soluble, and high levels of organic nitrogen make a very good culture medium for bacterial and fungal growth. It is important to remove these matters by eliminating the water through an open circuit or through the use of oxidation in a closed system (Manton, 1986). Chemical reactions also are related to temperature. The development of bacteria is accelerated by high temperature.

The objective of water sterilisation is to place the agent in contact with microorganisms by the most efficient means. Two methods are used: (1) point-contact sterilisation (e.g., ozonation, UV irradiation) or (2) bulk-fluid (e.g., chlorination) (Spotte, 1991). Chemical purification also includes catalysts such as copper and silver. The most widely distributed systems used are chlorination and ozonation. Chlorination still is used by a majority of facilities, but ozonation has become quite popular. Chlorination as a single choice is in decline.

Silver and Copper Catalysts—Metal ions, such as silver and copper, used in conjunction with chlorination, are efficient for maintaining good water clarity, removing bacteria, and controling algae; however, copper toxicity has been reported in marine mammals (Boness, 1996; McDevitt, 1986) and copper might accumulate in the liver (Ford, 1997). Its use will therefore be avoided.

Chlorination—Chlorine is one of the most popular chemical agents used for the elimination of microorganisms and algae (Faulk, 1990); however, it is a difficult process and can give rise to a number of problems (Boness, 1996; van der Toorn, 1987). Injudicious use of such chemicals can be more life-threatening than the organisms and excreta they control (Geraci, 1986). As opposed to ozone, chlorine generally is used as an oxidising agent in the bulk-fluid and has a prolonged time of action. Chlorine-based products are inexpensive, possess strong antimicrobial properties, and are easy to store and inject; however, they present important disadvantages in that they form products with lower oxidation potential—the chloramines—as well as mutagenic and carcinogenic byproducts (Spotte, 1991). Therefore, byproducts of the transformed chlorine are hazardous if not removed by chemical processes or frequent water renewal.

Chlorine is available as liquified gas (under pressure and referred to as liquid chlorine) and as solutions of chlorine compounds such as sodium hypochlorite (Manton, 1986); however, as Geraci (1986) stated, chlorine as sodium hypochlorite is perhaps the most commonly misused chemical treatment.

When chlorine gas (Cl_2) or sodium hypochlorite is mixed with water, it is transformed into hypochlorous acid (HOCl), which is an excellent oxidiser (Faulk, 1990; Manton, 1986; Spotte, 1991). For optimal oxidation, pH has to be maintained between 7.5 and 7.6. At higher pH levels, the concentration of hypochlorous acid, the active agent, will drop, therefore reducing its sterilisation capacities. Hypochlorous acid is highly unstable, however, as it breaks down with sunlight (Faulk 1990). Free chlorine is the amount of hypochlorous acid and hypochlorite ions (OCl-) free and available to react. Hypochlorous acid reacts with ammonia from urinary waste to form different chloramines (monochloramine and subsequently dichloramine and nitrogen trichloride). They persist longer than free chlorine, but they possess lower oxidising potential (Spotte, 1991).

Pool water is tested in dolphinaria for free and total chlorine. The survey showed that a safe range is below 0.2 ppm for free chlorine and 0.5 ppm for total chlorine (combined residual chlorine). Saltwater test kits for chlorine and chloramines can be used. Most of them give free and total chlorine readings. Faulk (1990) recommends to delineate chloramines as well. Surveyed facilities perform these tests two to three times per day.

Chlorine is a good bactericide; however, many protozoans, yeasts, cysts, and viruses are resistant (Boness, 1996). Skin infections are quite common in chlorinated water. It can be the result of the destruction of beneficial microflora and the inactivation of antimicrobial substances secreted by the skin (Geraci et al., 1986; van der Toorn, 1987). It potentially can interfere with the dolphin's ability to detect pheromones produced by other dolphins for social and sexual interactions (Herman & Tavolga, 1980; van der Toorn, 1987). Free chlorine used in the presence of humic and fulvic acids (e.g., natural dissolved organic carbon [DOC] or substances derived from humus) or some algae can produce carcinogenic trialomethanes (THM) (Boness, 1996; Spotte, 1991). Cetaceans ingesting a minimal amount of pool water are at minimal risks, however, if any at all (Spotte, 1991). Although their oxidation potential is lower, chloramines are capable of reducing concentrations of THM substantially (Spotte, 1991).

Another chlorine-based oxidant is chlorine dioxide (ClO_2), which is mixed on-site and presents the advantage of not forming chloramines directly, does not yield THM, does not react with bromide, and has comparable sterilising potential at lower dosages and contact times. It is sensitive to temperature, pressure, and light, however, which means that it has to be generated on site like ozone. It also is more expensive than other chlorine-based oxidants.

Seawater and artificial seawater contain bromide. Chlorination of these waters results in various elements together called active bromine that convert later into bromate and potentially brominated THM. Moreover, the presence of bromide reacts with chlorine and increases chlorine demand (Spotte, 1991).

Chlorine always will be added to the water after the mechanical filtration through a high-quality injection system and be properly mixed. Manually adding any type of chlorine is unsafe for both technicians and animals, and it does not properly distribute the chemical in the pool (Boness, 1996).

If dolphins are seen frequently with closed eyes, show signs of eye irritation, occasionally cough, or if the water smells of chlorine, the chlorination system probably is unbalanced. Action should be taken immediately to correct the problem as it can damage the animal's skin and eyes. Proper ventilation above and around the pools is crucial (Amundin, 1986; survey).

Chlorine also can be used in very small amounts (close to drinking water) in an open system or in conjunction with ozonation. Chlorination is incompatible with biological purification, however.

Ozonation—Ozone (O_3) is a point-contact sterilising agent that is also used for water discoloration. It also can have some effect in removing turbidity. Ozone is an excellent oxidising agent and bactericide. It also is effective in oxidising organic matter, iron, and manganese. It is, therefore, a convenient and efficient sterilising agent for marine mammal pools. It produces no taste or odour in the water. Ozone destroys bacteria and mold; eliminates spores, yeast, and fungus; and possibly inactivates viruses and cysts. Its effectiveness decreases at higher densities of organisms (van der Toorn, 1987). Since ozonation does not leave a disinfectant agent in the water, it does not have a long-lasting effect like chlorine, hence requiring a high water turnover.

Ozone is a highly reactive form of oxygen and can be produced by sending a high voltage electrical discharge through air or oxygen (such as what occurs in a lightning storm). Some degree of ozone also can be produced by certain types of UV lamps. As a gas, ozone is unstable and has a very short life, so it must be generated at the point of use. UV ozone generators are relatively simple and economical but are limited in output capacity. For larger volume treatment, a Corona discharge ozone generator is used. Air or concentrated oxygen is used to produce ozone on-site. A Corona discharge system uses very high voltage (5,000 to 14,000V) to split oxygen molecules, which will bond with other oxygen molecules and create ozone. Ozone is mixed with water in a reaction tank where the oxidation process occurs. The undissolved ozone/air mixture must be collected in the reaction tank and destroyed; otherwise, this dangerous gas will be entrained in the water flow and released, for example, into the atmosphere, at the pool water surface (Anonymous, 1997; Boness, 1996). Moisture in the gas stream should be reduced to a minimum as it can cause a serious drop in ozone production and the formation of nitric acid (Anonymous, 1997; Krajniak, 1988).

The concentration (C, in mg/l or ppm) of dissolved ozone multiplied by the time (T, in minutes) of contact between the dissolved ozone and the contaminants in the water provides a "CT value." Ozone, as a primary sterilising method, achieves adequate sterilisation with a CT of 1.6 (e.g., 0.4 mg/l of dissolved ozone maintained for 4 min equals a CT of 1.6) or greater. If the concentration is too low or the contact time too fast, sterilisation will not be optimal. High turbidity, cold temperatures, and high pH decrease the sterilisation capacity as well. Guidelines of CT values for the efficient destruction of microorganisms can be found in the *Guidance Manual for Compliance with the Filtration and Disinfection Requirements for Public Water Systems Using Surface Water Supplies*, published in 1989 by the USA Environmental Protection Agency in Washington, DC. Countries, such as France and Canada, have adopted the same standards (Anonymous, 1997). The production of ozone usually is expressed in g/l. In relation to the variable load and pollution in the water, the ozone quantity is controlled and adjusted by a Redox (Reduction/Oxidation) measuring apparatus. An ozone detector should be installed in the direct vicinity of the ozone installation to warn against the presence of ozone in the surrounding atmosphere. The ozone installation can be switched off when the measured value exceeds 0.1 ppm.

The chemistry of ozone is influenced by the presence of bromide; therefore, it is modified mainly by seawater, artificial seawater, and freshwater, which contain this element. Ozonation of seawater oxidises bromide to bromine and, ultimately, as bromate (as described similarly with chlorine above). Also like chlorine, in the presence of humic acid, it can yield toxic compounds, possibly at a higher level than chlorination. Again, the very limited amount of ingested pool water by cetaceans makes it most likely harmless. Ozonation in the presence of bromide is less efficient and increases ozone demand, but it also forms persistent oxidants that serve as weak bulk-fluid sterilising agents (Spotte, 1991).

Chlorination in small dosages can be used in conjunction with ozonation to prolong the sterilisation effect or limit algal growth. The use of ozone is compatible with protein skimmers (see combined techniques below). One of the greatest advantages of ozone, compared to chlorine, is that fish and crustaceans can survive in ozonated water (if there is no hypobromic acid). This creates a richer and potentially more interactive environment for the animals and a visually more attractive display to the visitors. The animals also can help maintain a cleaner environment by grazing on the algae. With the increasing popularity of ozone systems to sterilise marine mammal pools as well as potable water, ozonators are becoming cheaper, safer, and more reliable. Ozone is considered one of the best sterilising options for dolphinaria at present.

Biological Purification

Biological filtration is the process by which waste products, principally ammonia, are broken down by bacteria using them as a food source. Systems, such as trickling filters, seldom have been used in oceanaria; therefore, we will not examine them. Biological filtration mainly is used in the denitrification process to remove accumulating nitrate in a system by transforming it to nitrogen gas. The denitrification process is installed on a side-stream to treat a percentage of the water each day. There are two kinds of biological denitrification: (1) heterotrophic and (2) autotrophic. Heterotrophic denitrification is based on anaerobic bacteria using nitrite and nitrate as a food source and ethanol or methanol as carbon substrate (Hignette et al., 1997). This method requires a very slow water circulation speed in the filter (0.2 to 2 m/h) and a high consumption of carbon substrate, making it more appropriate for small volumes in aquariology than for marine mammal water treatment.

Figure 6.6. Ozone control cabinet (Photograph from I. Smit)

Figure 6.7. Small ozone contact chamber

Autotrophic denitrification is based upon a sulphur oxidising bacterium (*Thiobacillus denitrificans*) in tanks filled with sulphur balls (1 to 4 mm diameter) and calcium carbonate. Calcium carbonate is present to remove the sulphates produced and to balance the acidification of the pH induced by the denitrification (Hignette et al., 1997). This technique sustains much higher filtration speed (2 to 9 m/h) and is more tolerant to dissolved oxygen than the heterotrophic denitrification method. Denitrification is not yet common in marine mammal LSSs, but it is gaining in popularity, and the autotrophic method is certainly the most appropriate.

UV-Light Irradiation

Ultra-Violet (UV) light irradiation is neither a mechanical, chemical, nor biological process. It is more of a physical process, and it is quite widely used—mainly for small volumes or when other marine organisms, such as fish and invertebrates, which do not tolerate the presence of a residual disinfectant like chlorine, are present. It also can be used for the destruction of residual ozone after the contact chamber. Microorganisms, such as bacteria, algae, yeast, and viruses, are killed or inactivated by the UV radiation produced by the UV lamp that penetrates the microorganism's cell wall and membrane, destroying the inner material. These microorganisms require a precise wavelength and duration of irradiation to be destroyed; otherwise, they can resist, repair, and regrow (Mofidi et al., 2002). UV sterilisation is a point-contact process. Efficiency depends on the time of contact; therefore, the speed of the flow is crucial. UV sterilisation efficiency is impeded by turbidity; the higher the number of suspended particles in the water, the less light penetration (Spotte, 1991). The precise UV dosage will be determined by the quality of water and the type of microorganisms targeted. UV dose is recorded as a product of irradiance (milliwatts per square centimetre) and time (second), which provides units of milliJoules per square centimetre (mJ/cm^2) (Mofidi et al., 2002). Low-pressure lamps are preferred as they have a longer life and are more economical to replace. Recommended dosage is between 20 and 35 mJ/cm^2 (calculated at the end of lamp life) at a wave length of 254 nanometer (nm). UV light sterilisation is not the most efficient system for large volumes (Spotte, 1991); it is also one of the less economical (Boness, 1996); however, it is safer in comparison with other means of sterilisation in terms of potential production of harmful byproducts.

I recommend that a newly installed systems be run for several days to several weeks before introducing animals into the pool to ensure that there are no toxicity concerns, leakage, or loose materials from the LSS.

In conclusion, there is not one solution that has all the advantages. The survey showed that new LSSs tend to be more complex and that a combination of techniques is the most efficient solution. Nowadays, it is common to see an LSS equipped with high-pressure sand filters as the basic mechanical installation. This is used in combination with either a high dosage of ozone or protein skimmers, combined with a lower dosage of ozone. This also is associated with activated carbon filters or denitrification filters on side-streams, sometimes a side-stream treated by UV, and often a small dosage of chlorine as a residual oxidant in bulk-fluid to prolong oxidation capacity and control algal growth. This combination of techniques allows all of the different and extremely complex reactions occurring in cetacean pool water purification to be tackled with more subtlety.

Figure 6.8. UV unit (Photograph from I. Smit)

Water Flow and Drainage

Flow mechanics in pools are a delicate and complicated matter, and they are difficult to summarise. Water circulation depends mainly on the volume of water and the topography and depth of the pool. In general, water flows into the pool through several inlet pipes located in the upper section, and sumps located at the bottom of the pool collect water to outlet pipes. Both inlet and outlet pipes should be placed in such a way that they leave no "dead corner" in the pool. The survey indicated that it is sometimes obvious that such pockets of poorly agitated water exist when leaves, food debris, and dirt regularly accumulate at the bottom.

Water inlet pipes also can be located all around the perimeter of the pool, close to the water surface. This system is found in semi-natural pools where pipes can be hidden behind rocks and are designed to distribute water evenly.

Air entrapment in the water column is a common problem in a water circulation system. High turbidity levels increase air entrapment as suspended solids bind with air bubbles and prevent them from ascending to the surface and escaping. It is caused by a high water flow rate that produces turbulence in some parts of the circulation system. It does not cause an immediate health threat to the animals, but it can severely disrupt their echolocation abilities (Fasik, 1991).

Water turnover rate depends on various factors such as the volume of water to treat or replace, the number of animals, the water temperature, and the sterilisation system. The survey indicated that dolphin exhibits usually have a water volume between 1,500 and 3,000 m^3. New exhibits have larger volumes—between 4,000 and 10,000 m^3. According to the number of animals and climatic conditions, turnover rate ranges between 1½ h for the smallest pools and 6 h for the largest ones. In semi-natural facilities where volumes are often much larger, turnover rate can be as long as 8 h. The survey data did not allow calculating an adequate formula to determine an appropriate turnover rate for any given situation.

Grates covering sumps should be carefully secured to the bottom of the pool. While playing, dolphins might remove or push away the grates covering the sumps and might be sucked into the drain. If they are unable to withdraw, they may drown (Sweeney, 1990; Sweeney & Samansky, 1995; survey).

The contents of grids, meshes, skimmers, and similar equipment will be carefully monitored and debris regularly and frequently removed. Any unusual material will be promptly investigated (Klinowska & Brown, 1986).

Piping design in an artificial environment should allow the pools to be emptied and filled rapidly for maintenance and animal husbandry. Pool cleaning should not be done by emptying the pool. It is not recommended to leave the animals stranded on the bottom of the pool because it induces a high level of stress and great physical discomfort due to the lack of buoyancy. It is preferable to use special underwater devices that are operated automatically, remotely, or with the assistance of divers.

Pools and lagoons need an appropriate draining system to discharge unwanted water. Water spilled by the animals, rainwater, and cleaning water should not be allowed to re-enter the pool. Linear grate-covered drains can be placed around the perimeter of the pools. Linear drains collect much larger amounts of water than gully traps, provided that appropriate piping is installed. In tropical regions, storm drains have to be installed to divert the maximum amount of rainwater and to maintain salinity level since the pool surface already collects a large volume of freshwater. Pool decks will be designed to slope toward the drain and not toward the pool.

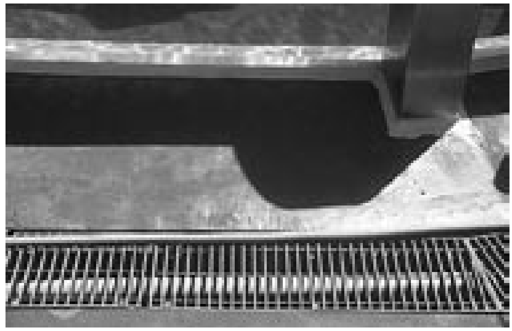

Figure 6.9. Floor drain

Figure 6.10. Vortex due to a too high aspiration of sump

Literature Cited

Amundin, M. (1986). Breeding the bottle-nosed dolphin *Tursiops truncatus* at the Kolmarden Dolphinarium. *International Zoo Yearbook*, *24/25*, 263-271.

Anonymous. (1979-1984, 1995). *Specifications for the humane handling, care, treatment and transportation of marine mammals* (9 CFR, Subpart E, §3.100-118). Washington, DC: U.S. Department of Agriculture, Animal and Plant Health Inspection Service.

Anonymous. (1992a). *Standard methods of water and wastewater* (18th ed.). Washington, DC: American Public Health Association, American Water Works Association, Water Environment Federation.

Anonymous. (1992b). *Guidelines for the care, holding and transport of marine mammals in Queensland* (Draft). Queensland, Australia: Queensland Department of Primary Industries (QDPI). 39 pp.

Anonymous. (1997). *Corona discharge ozone systems: Application guidelines.* San Luis Obispo: DEL Industries. 22 pp.

Anonymous. (1999). *WQA glossary of terms* (4th ed.). Lisle, IL: Water Quality Association.

Arkush, K. D. (2001). Water quality. In L. A. Dierauf & F. M. D. Gulland (Eds.), *CRC handbook of marine mammal medicine* (2nd ed.) (pp. 779-790). Boston: CRC Press.

Boness, D. J. (1996). Water quality management in aquatic mammal exhibits. In D. G. Kleiman, M. E. Allen, K. V. Thompson, S. Lumpkin, & H. Harris (Eds.), *Wild mammals in captivity: Principles and techniques* (pp. 231-242). Chicago: The University of Chicago Press.

Castro, P., & Hubert, M. E. (1992). *Marine biology*. Dubuque, IA: WCB Publishers. 57 pp.

Cornell, L. H. (1982). Husbandry of marine mammals. In K. Sausman (Ed.), *Zoological parks and aquarium fundamentals* (pp. 283-287). Wheeling, WV: American Association of Zoological Parks and Aquariums.

Dierauf, L. A. (Ed.). (1990). *CRC handbook of marine mammal medicine: Health, disease, and rehabilitation.* Boston: CRC Press. 735 pp.

Doherty, J. G., & Gibbons, E. F. (1994). Managing naturalistic animal environments in captivity. In E. F. Gibbons, E. J. Wyers, E. Waters, & E. W. Menzel (Eds.), *Naturalistic environments in captivity for animal behavior research* (pp. 126-139). Albany: State University of New York Press.

Dunn, J. L. (1990). Bacterial and mycotic diseases of cetaceans and pinnipeds. In L. Dierauf (Ed.), *CRC handbook of marine mammal medicine: Health, disease, and rehabilitation* (pp. 73-87). Boston: CRC Press.

Fasik, J. I. (1991). Echolocation interference in the presence of air bubbles. *Proceedings of the 19th IMATA Conference*, Vallejo, California.

Faulk, E. Y. (1990). Water quality considerations for marine mammals. In L. Dierauf (Ed.), *CRC handbook of marine mammal medicine: Health, disease, and rehabilitation* (pp. 537-542). Boston: CRC Press.

Ford, B. K. (1997). An update on the copper-silver water purification system in use at Gulf World. *Soundings*, *22*(2), 14-15.

Geraci, J. R. (1986). Marine mammals: Husbandry. In M. E. Fowler (Ed.), *Zoo & wild animal medicine* (pp. 757-760). Philadelphia: W. B. Saunders Company.

Geraci, J. R., St. Aubin, D. J., & Hicks, B. D. (1986). The epidermis of odontocetes: A view from within. In M. M. Bryden & R. J. Harrison (Eds.), *Research on dolphins* (pp. 3-21). Oxford: Clarendon Press.

Gregory, J. (1989). Fundamentals of flocculation. *CRC Critical Reviews in Environmental Control*, *19*(3), 185-230.

Herman, L. M., & Tavolga, W. N. (1980). The communication systems of cetaceans. In L. H. Herman (Ed.), *Cetacean behavior: Mechanisms and functions* (pp. 149-209). Malabar, FL: University of Hawaii, R. E. Krieger Publishing Company.

Hignette, M., Lamort, B., Langouet, M., Leroy, S., & Martin, G. (1997). Elimination des nitrates par filtration biologique autotrophe sur soufre en aquariologie marine. *Mémoire Institut Océanographique Paul Ricard,* pp. 7-13.

Kannan, K., Senthilkumar, K., Loganathan, B. G., Odell D. K., & Tanabe, S. (1997). Elevated accumulation of tributyltin and its breakdown products in bottlenose dolphins (*Tursiops truncatus*) found stranded along the U.S. Atlantic and Gulf Coasts. *Environmental Science and Technology*, *31*(1), 296-301.

Kennedy-Stoskopf, S. (1990). Immunology of marine mammals. In L. Dierauf (Ed.), *CRC handbook of marine mammal medicine: Health, disease, and rehabilitation* (pp. 115-125). Boston: CRC Press.

Klinowska, M., & Brown, S. (1986). *A review of dolphinaria.* London: Department of the Environment. 247 pp.

Krajniak, E. (1988). Potential marine mammal exhibit physical plant failures. *Proceedings of the 16th IMATA Conference*, San Antonio, Texas.

Liong, E., Hammond, D. D., & Vedros, N. A. (1985). *Pseudomonas pseudomallei* infection in a dolphin (*Tursiops gilli*): A case study. *Aquatic Mammals*, *11*(1), 20-22.

McDevitt, A. (1986). Dolphins and whales improving. *Minneapolis Zoo Magazine*, *1*, 1.

Manton, V. J. A. (1986). Water management. In M. M. Bryden & R. J. Harrison (Eds.), *Research on dolphins* (pp. 189-208). Oxford: Clarendon Press.

Mofidi, A. A., Rochelle, P. A., Chou, C. I., & Mehta, H. M. (2002). Bacterial survival after ultraviolet light disinfection: Resistance, regrowth and repair. *Proceedings of the Water Quality Technology Conference 2002*, American Water Works Association. 10 pp.

Overath, H., Hartmann, M. G., & Gellissen, M. (1997). *Bacteriological studies of the water of the Duisburg Zoo Dolphinarium with respect to fish food as a source of contamination.* Duisburg: Forshungszentrum Jülich GmbH and Duisburg Zoo.

Reeves, R. R., DeMaster, D. P., Hill, C. L., & Leatherwood, S. (1994). Survivorship of odontocete cetaceans at Ocean Park, Hong Kong, 1974-1994. *Asian Marine Biology*, *11*, 107-124.

Ridgway, S. H. (1972). Homeostasis in the aquatic environment. In S. H. Ridgway (Ed.), *Mammals of the sea: Biology and medicine* (pp. 590-747). Springfield, IL: Charles Thomas.

Smith, R. L. (1990). *Ecology and field biology* (4th ed.). New York: HarperCollins.

Spotte, S. (1991). *Sterilization of marine mammal pool waters* (Technical Bulletin No. 1797). Washington, DC: U.S. Department of Agriculture, Animal Plant Health and Inspection Service. 59 pp.

Sweeney, J. C. (1986). Infectious diseases. In M. E. Fowler, (Ed.), *Zoo & wild animal medicine* (pp. 777-781). Philadelphia: W. B. Saunders Company.

Sweeney, J. C. (1990). Marine mammal behavioral diagnostics. In L. Dierauf (Ed.), *CRC handbook of marine mammal medicine: Health, disease, and rehabilitation* (pp. 53-72). Boston: CRC Press.

Sweeney, J. C. (1993). *Dolphin Quest protocol for veterinary health care management of* Tursiops & Steno. San Diego: Dolphin Quest.

Sweeney, J. C., & Samansky, T. (1995). Elements of successful facility design: Marine mammals. In E. F. Gibbons, Jr., B. S. Durrant, & J. Demarest (Eds.), *Conservation of endangered species in captivity: An interdisciplinary approach* (pp. 465-477). Albany: State University of New York Press.

van der Toorn, J. D. (1987). A biological approach to dolphinarium water purification: I. Theoretical aspects. *Aquatic Mammals*, *13*(3), 83-92.

Aquatic Mammals 2005, *31*(3), 364-370, DOI 10.1578/AM.31.3.2005.364

7. Food and Fish House

Food

Most cetaceans in the wild are opportunistic feeders and consume a wide variety of fish, molluscs, and crustaceans (Geraci, 1986). Some large-toothed whales also consume other marine mammals; however, their diet is more restricted in captivity because of the commercial availability of frozen fish and cost restrictions (Worthy, 2001). A balanced diet and fresh food are essential for keeping the animals in good health. Emphasis is put on the quality of food selection and storage, as well as the importance of hygiene in preparing and handling the food and in feeding the animals.

Food Types

In most cases, mackerel (Scombridae, *Scomber* sp.), herring, and sardines (Clupeidae, *Clupea harengus* and *Sardinops* or *Sardinella* sp.), as fatty fish species, are supplemented by two or more leaner fish species, such as whiting (Gadidae, *Gadus merlangus, Merlangus merlangus*), capelin, or smelt (Osmeridae, *Mallotus villosus,* O*smerus mordax*). Squid (*Illex* or *Loligo* sp.) is the most widely used invertebrate. Other species quite commonly fed to cetaceans in human care are the mullet (*Mugil* sp.), anchovie (*Engraulis* sp.), sprat (*Sprattus sprattus*), pilchard (*Sardina pilchardus*), cod (*Gadus morhua*), pollock (*Pollachius* sp.), octopus (*Octopus* sp.), cuttlefish (*Sepia officinalis)*, and a wide variety of local species (Geraci, 1986; Worthy, 1990; survey). Coastal surveyed facilities sometimes supplement the dolphins' diet with live local fish. Nutritional characteristics vary according to the species and also are dependent on the location and the season of capture (Geraci, 1986; Ridgway & Harrison, 1994; Crissey, 1998).

Little information was received through the survey on river dolphins' diet except for the Baiji, which is fed with several species of carp (Cyprinidae, e.g. *Cyprinus carpio*, *Carassius carassius*) in captivity (survey). Indus and Ganges River dolphins' diet in the wild includes prawn (*Palaemon* sp. and *Paneus* sp.), clam (*Indonia coerulea*), catfish (Siluridae, including *Wallago attu, Macrones aor*), freshwater shark (*Saccobranchus fossilis*), gobie (*Glassogobius giuris*), and carp (*Catla buchanani, Labeo rohita*). They also have been given live dace (*Leuciscus leuciscus*) in captivity (Ridgway & Harrison, 1989; Klinowska, 1991). *Inia* feeds in the wild on Sciaenidae, Curimatidae, Cichlidae, and Siluridae (Leatherwood & Reeves, 1983; Ridgway & Harrison, 1989). In captivity, it is fed live and dead carp, as well as live trout (*Salvelinus* sp. or *Salmo* sp.), (Ridgway & Harrison, 1989; Burrows et al., 1990). The Tucuxi's diet varies according to its location along the southern Atlantic coast of South America. It comprises mostly fish of the families Sciaenidae, Engraulidae, and Clupeidae, as well as Siluridae—crustaceans (prawn and crab) and cephalopods (squid and cuttlefish) (Ridgway & Harrison, 1989; Terry, 1990; Klinowska, 1991).

For further details on wild cetacean diets and energy budgets, please refer to Evans (1987), Ridgway & Harrison (1989, 1994, 1999), Klinowska (1991), and Worthy (2001).

The variety of fish species used by 35 institutions among the 44 in the survey, worldwide, is shown in the following table. Cetaceans are fed on average five fish species (facilities = 35, median = 5, minimum = 2, maximum = 10) at any one time. It was not indicated in the survey if some species were used only seasonally.

Table 7.1. Percentage of facilities in the survey using cetacean food by species

Fish type	% facilities (35)
Mackerel (*Scomber* sp.)	94
Herring (*Clupea harengus* sp.)	77
Squid (*Illex* or *Loligo* sp.)	63
Capelin (*Mallotus villosus*)	43
Sardine (*Sardinella* sp.)	31
Whiting (*Gadus merlangus*, *Merlangus merlangus*)	31
Smelt (*Osmerus mordax*)	29

Diet

Fish fed to captive cetaceans should be of the finest quality, similar to human standards (Anonymous, 1979-1984, 1995; Geraci, 1986; Crissey, 1998). A bacteriological study examining contamination of the poolwater revealed that food is probably the prime source of potentially harmful bacteria in the water treatment system (Overath et al., 1997). Bacteria such as *Streptococcus* sp., *Pseudomonas* sp., and *Pasteurella* sp., among others, have been isolated in fish fed to dolphins. *Erysipelothrix rhusiopathiae* is another pathogenic bacterium, which also is found in food. It causes erysipelas, which, in its septicaemic form, can be lethal (Dunn, 1990; Sweeney, 1993). Feeding inferior-quality fish is wasteful; it can lead to severe health consequences due to poor nutritional value, and it can even lead to death if there is contamination.

A mixed diet of fish and invertebrates provides a balanced source of fat, protein, vitamins, minerals, and water, as well as a mixed diet of fatty and lean species and invertebrates like squid, the leanest of all. Similar amounts of different species of fish provide a great disparity in energy content. Many prey species show changes in composition, and, thus, energy content, according to age, sex, season, and location of capture (Kastelein et al., 2003; Logerwell & Schaufler, 2005). This is why it is advised to conduct the analysis of fish composition to determine the appropriate diet (Worthy, 2001). Cetaceans in human care will not be fed only one or two fish species because this may lead to possible nutritional deficiencies (Geraci, 1986; Worthy 1990, 2001); five or more are preferable (Anonymous, 1992). With the exception of river dolphins, cetaceans do not have access to freshwater; therefore, they depend on the water absorbed through their food—directly available or derived from the combustion of fat. The fatter the fish, the more water and energy are available (Worthy, 1990, 2001).

The smallest species of odontocetes, the harbour porpoise (*Phocoena phocoena*), requires a daily portion of food estimated to be up to 13% of their body weight (Evans, 1987), whereas bottlenose dolphins may require between 3 and 6.5% of body mass per day (Barros & Odell, 1990), depending on various parameters. Nevertheless, food intake is an individual parameter related to caloric calculations based on metabolic rate and depends on the species, individual, age, sex, season, activity level, participation in shows, and reproductive status (Reddy et al, 1994; Kastelein et al., 2003). It is important that the profile of a captive animal's diet reflects that of its species in the wild. For example, the Risso's dolphin (*Grampus griseus*) feeds on bottom-dwelling cephalopods (Ridgway & Harrison, 1989) and should be offered a diet of cephalopods in captivity. Although Inias have been fed mackerel in the past in Japan (Sylvestre, 1985), river dolphins should be offered only freshwater fish, if possible, or species similar to the ones in their natural habitat.

Crissey (1998) recommended that whole fish be used as often as possible because cut-up fish loses important nutrients from beheading, evisceration, and leaching. Vitamin supplements are important to replace loss through storage, thawing, and processing. Fish such as smelt, herring, capelin, and some types of mackerel contain thiaminase, which induces thiamine deficiencies in cetaceans that can provoke severe to fatal disorders (Geraci, 1986; Worthy, 1990, 2001). It is therefore important to supplement the food ratio with a dose of thiamine—25mg/kg of fish—when feeding fish containing thiaminase (Geraci, 1972; Worthy, 2001). Please refer to Worthy (2001) for the table of fish containing thiaminase.

For details on food intake, calculation of caloric requirements, vitamin supplements, and nutritional disorders, please refer to Dierauf & Gulland (2001), Geraci (1986), and Worthy (1990, 2001).

Purchase and Storage

Crissey (1998) recommended that the fishery or fish supplier supply the history and location of the catch and record the catch date on the boxes. Occurrences of pesticide and heavy metal contamination, as well as biological contamination (e.g., red algae bloom), should be recorded and flagged. Fish supplied by fisheries for cetacean consumption should be caught, processed, and stored as if they were intended for human use; fish will remain whole, however.

Fish will be quick-frozen—individually or in blocks (Crissey, 1998)—packaged in plastic-lined boxes or in plastic bags, with the date of the catch printed on it. Packaging will be impervious to air and moisture to retain its quality and water content (Geraci, 1986). Crissey (1998) reported that package weight preferably will not exceed 10 to 15 kg to allow proper thawing, but this size is not always available from fisheries. Because thawed fish should be consumed within 24 h, packet size will not exceed the necessary daily amount to avoid waste. Individually Quick Frozen (IQF) packs are preferred over blocks because the fish thaw more quickly and more uniformly and therefore the quality is retained; blocks usually contain broken or crushed fish. IQF packs are more expensive than boxes, but the amount of waste is much less. According to the survey, packs are usually of better value and easier to handle. At least one of the fish species that will be used in training or during shows will be of small size (e.g., smelt, sardine, anchovy, etc.) to avoid the necessity of cutting them into small pieces, which causes nutrient loss and increases preparation time (Crissey, 1998).

Ideally, samples of each fish species will be analysed from every new shipment prior to the delivery. Facilities often test for several of the following: caloric content, bacterial count, fat, protein, carbohydrates, moisture, vitamins, minerals, histamine, putrescine and cadaverine, volatile ammonium, peroxide, and heavy metals (most common are arsenic, cadmium, chromium, copper, lead, mercury, nickel, selenium, and zinc [Skoch, 1990]), dichlorodiphenyltrichloroethane

(DDT), polychlorinated biphenyls (PCB), and tributyltin (TBT) (Amundin, 1986; Kannan et al., 1997; survey). These analyses are fairly expensive and are not available everywhere. Furthermore, results can significantly vary among laboratories (Logerwell & Schaufler, 2005). It is therefore recommended to be consistent with the choice of laboratory and analysis methods. The fat content may vary according to the catch season and location (Logerwell & Schaufler, 2005), and it affects the amount of food required by the animals' caloric needs. If heavy metals—DDT, PCB, and TBT—are detected, or if the health history of fish is doubtful, the shipment should be refused.

Once ordered, the shipment will be inspected before and during the delivery for any sign of pests, or thawing and refreezing. The type, size, quantity, and quality of fish and the number of boxes or packs should be checked. If there is water or ice build-up on the boxes or the floor beneath, if the wrapping is moist or slimy, or if the flesh is flabby or exudes a sour odour, it should be rejected. These signs mean that temperature has not been constant during storage or transportation. Once thawed, fish has bright red gills, prominent clear eyes, and firm elastic flesh. If it appears dull, eyes cloudy or red-bordered, or if finger impressions remain on the flesh, it means that the fish is old or has been thawed and refrozen and is therefore unacceptable (Crissey, 1998; survey).

Before storing a new shipment, the remaining stock should be placed so that it will be used before the new one. Crissey (1998) and those surveyed recommended that the date of reception and expiration date should be written or stamped on the boxes or packs. All the data related to fish purchase, analysis, delivery, and storage will be recorded. If frozen fish has to be transported from a central freezer to a secondary freezer, as often happens in large facilities, similar procedures will be followed. Fish supply should be stored in walk-in freezers between -25° and -30° C (Geraci, 1986; survey), and for no longer than 6 months. Scombroid fish (i.e., mackerel or tuna) have a short shelf-life and deteriorate quickly. They can cause scombroid poisoning if eaten after an extended period (Worthy, 2001). The toxic effects can be quite serious, even without visible evidence of putrefaction (Geraci, 1986; Worthy, 1990, 2001). Scombroid fish should not be kept longer than 3 to 4 months (Anonymous, 1992; Worthy, 2001).

Relative humidity will be maintained between 85 and 90% to decrease dehydration of the frozen items (Crissey, 1998). Refrigerators and freezers will be used for perishable food only. No substance known to be, or which may be, toxic or harmful to an animal should be stored or maintained in areas used for food storage (Anonymous, 1992).

Thawing and Feeding Procedures

The officially recommended way to thaw fish is in a refrigerated space. Fish will be thawed overnight, or as close to feeding as possible, in a refrigerator at a temperature of 4º to 6º C (Geraci, 1986; Crissey, 1998; survey). It is allowed to thaw fish in cool running water (max. 8º C), but it causes nutrient loss—especially water-soluble nutrients. It is better to thaw it in slightly salty water to maintain osmotic gradient and avoid losses (Geraci, 1986). Fish should never be thawed by leaving it to soak in water, at room temperature, or in the sun. The use of fans to speed thawing causes loss of fluid through dehydration. Crissey (1998) recommended that fish be left in its wrapping or container to provide insulation and to allow for uniform thawing. "Dry" thawing takes longer than "wet" thawing, and there is a potential risk of pathogen multiplication. The debate over which method is preferable and safer has not yet been resolved.

All fish should be fed to the animals within 24 h following its removal from a freezer for thawing. If not used, fish must be discarded (Anonymous, 1979-1984, 1995). Fish dropped on the floor also should be discarded. Thawed fish will be kept iced or refrigerated until feeding time. Frozen fish, once thawed, should NEVER be refrozen. Fish waste will be kept in a separate refrigerator or an insulated container, and it will be thrown away as quickly as possible.

Food should be wholesome, palatable, free from contamination, and of sufficient quantity and nutritive value to maintain the animal in good health (Anonymous, 1979-1984, 1995). During preparation, each fish is inspected for quality and cut into pieces for training purposes only if necessary. Fish of smaller size are preferred over cut fish for this purpose. Fish will be fed cool, but not frozen. Animals should be fed at least twice daily, unless otherwise stated by a veterinarian, but preferably three to four times daily. Prior to feeding, individual buckets, labelled with the name of each animal, or engraved, are prepared with their individual ration according to their predetermined diet. Vitamins and mineral supplements should be given daily. Refer to Worthy (2001) for details. Trainers should never consume vitamins prepared for animals. Tablets are usually hidden by inserting them in fish gills or into the abdominal cavity and should be fed first (survey). To ensure that conditions are appropriate and that methods for storage and handling fish are proper, validation of the conditions and procedures are needed before they become policy and practice. They will be reexamined at periodic intervals to ensure compliance, as well as when they are changed (Crissey, 1998). Several surveyed facilities recommend to establish a preparation and feeding procedure and provide the staff with adequate training.

Cetaceans should be fed individually with previously cleaned hands, and the staff in charge of feeding will learn to recognize any alterations from the normal state of health and behaviour in order to adjust food intake, consulting a veterinarian when necessary. The quantity and type of food consumed by each animal should be recorded after each feeding session (Crissey, 1998; survey). Food can be used as a reward, but it should never be withheld as a punishment.

When the animals are fed outside in cool or cold conditions, no extra cooling is needed. When they are fed inside, or outside at a temperature of 18º to 20º C, the fish is kept in cooled containers. If they are fed outside in warm to hot conditions, it is important to keep the fish iced and covered in pool-side coolers to avoid microbial build-up and contact with pests (Crissey, 1998; survey). Melted ice and fish juices will be drained from the buckets periodically (survey). Ice can be placed separately in a plastic bag, or the fish can be placed in a covered, insulated container.

If members of the public are allowed to feed the animals, it only should be done under staff supervision. The quantity of fish given to individual animals will be recorded to adjust the subsequent rations. Only fish provided by the institution can be allowed (Klinowska & Brown 1986). Close contact between dolphins and people facilitates the transfer of microorganisms that have pathogenic potential. It is recommended that strict hygienic rules be observed by members of the public who contact dolphins directly. They should wash their hands prior to handling the fish, and not place anything in an animal's mouth that they would not place in their own (Geraci & Ridgway, 1991).

Fish House

Freezer and Refrigerator

The survey respondents recommended that there be two access points to a freezer—one from the outside, wide enough for direct loading from a truck using a forklift or from an electric cart, and the other from the preparation room. Bunk freezers are not recommended, even for small facilities, as they do not allow a proper turnover of the boxes or packs and are difficult to access. Bags of IQF can be stored on shelves or in special containers (survey).

The surface and volume of a freezer required by a facility can be calculated in the following ways:

- The duration of the supply according to the amount of food eaten by the animals in one day/week/month
- The number of boxes/packs this total represents, including waste, and the amount of space (surface and volume) required for them, stacked at a maximum height of 1.8 m to ease manipulation (The supplier can help with the evaluation.)
- Additional 0.6 m open space around the perimeter of the freezer and above the stacks to allow good air circulation, and a central path between stacks of at least 0.9 m

Large, stainless steel, professional kitchen-type refrigerators can be placed inside the preparation room, or a walk-in refrigerator can be placed between the freezer and the preparation room. Walk-in refrigerators, shelves, and fittings will be made of materials that are easy to clean, disinfect, and maintain (e.g., stainless steel) (survey).

Refrigerators and freezers will have a temperature display panel located near the front door, and the temperature will be checked and recorded daily. They should be equipped with an alarm system to signal temperature increase. In case of power failure or system breakdown, an emergency generator is necessary to maintain the required temperature until the stock is transferred to another freezer or until repair is completed.

In the unfortunate case of a dolphin's death, the carcass should not, under any circumstances, be kept in the food supply freezer. A special freezer located in the necropsy room will be kept solely for this purpose or the carcass should be dealt with by the veterinarian.

Preparation Room

The preparation room is the place where fish are thawed and processed. In most institutions, trainers handle the food preparation. They spend several hours daily in the fish house, processing the food, and thereafter, sanitising the space. Changes of level and stairs make the trainer's task of carrying food to the animals several times a day very difficult. Therefore, the locations of the fish house and freezer are of great importance. The survey recommended that they be located at the same level as the dolphin pool and as close to it as possible, preferably not be under the stage or pool deck. Trainers should not have to carry the food over different levels nor carry excessive weight over unreasonable distances.

Stainless steel sinks and tables are recommended over concrete or enamel because they are easy to clean and disinfect and are resistant to punctures and cuts. Concrete is resistant, but it requires thorough scrubbing since its surface is not smooth. Concrete can be used for floors if the finish is very smooth and receives a waterproof treatment or a high resistance epoxy finish. Industrial ceramic or aggregate tiles can be used, but grouts are difficult to clean. Plastic or vinyl liners are not recommended for fish house flooring as they are not resistant enough to punctures, cuts, and disinfectants. Floor lining will be applied on wall plinths as well (15 to 20 cm high). Floors should be made of no-slip materials. All materials used in the preparation room, from floor to ceiling, should be resistant to detergents, disinfectants, hot water, and high pressure water. Preparation rooms also will be equipped with a pressure hose and hot water taps.

Survey respondents recommended that the preparation room be equipped with an ice machine (even in facilities located in cold countries) and a manual or electronic stainless steel scale to weigh the amount of food given to the animals. The accuracy of the scale calibration will be checked regularly. Near the scale, a board will indicate the name of each dolphin, its daily food intake, and the portions of fish species given. Data should then be recorded daily and stored with other records, preferably digitally. Data can be more easily analysed if entered into a database or spreadsheet application.

Figure 7.1. Fish kitchen

Plastic or stainless steel buckets are used for food distribution. Stainless steel buckets are definitely preferred over plastic. Although more expensive, they are easier to clean, more resistant to intensive manipulation, and less likely to tip over (survey). It is recommended that there be an elevated stainless steel mesh or perforated sheet at the bottom of the bucket to allow melted ice and fish juices to be drained away from the remaining food. Stainless steel lids should be used to cover the food during transportation from the kitchen to the pool to protect it from outside elements such as birds and wind-carried objects. Each animal requires two or three buckets if the food is prepared in the morning for the whole day; the feedings also can be placed in layers in the same bucket. After the buckets are prepared, there needs to be space available to store them. Buckets can be hung upside-down on long pegs fixed in a wall

Figure 7.2. Grinder under sink

Figure 7.3. Weighing individual rations

to air dry after cleaning and sanitising. The layout of the preparation room will provide ample space to allow people to carry out several tasks simultaneously. No cleaning and disinfection should be conducted until the food is safely put in buckets and removed from the room.

Sanitation

Special attention will be paid to sanitation. Utensils, processing surfaces, walls, and floors must be cleaned and sanitised prior to and after fish processing (Crissey, 1998). Staff must wash their hands prior to handling food, vitamins, and medicine tablets. A footbath with disinfectant should be placed at the entrance of the preparation

Figure 7.4. Buckets and coolers

Figure 7.5. Fish cart

room, and staff must wear appropriate footwear like rubber boots.

Containers, such as buckets, tubs, and tanks, as well as utensils, such as knives and cutting boards, along with any other equipment which has been used for holding, thawing, or preparing food, should be cleaned and sanitised at least once daily if the animals are fed more than once a day (Anonymous, 1979-1984, 1995). Kitchen and other food handling areas should also be cleaned and sanitised daily. Fish prepared with unclean equipment may become contaminated, rendering it unfit for consumption (Crissey, 1998). The ice machine, refrigerators, and freezers should be cleaned and disinfected weekly (survey).

Small equipment should be cleaned with a detergent and sanitised by washing with hot water of 82º C or higher in a washing machine. Tables, sinks, buckets, scales, and all surfaces and large equipment should be disinfected by using a sanitising rinse, which should remain in contact with the surface for at least a minute and then that surface should be thoroughly rinsed with water. Sodium hypochlorite disinfectants (chlorine) such as Chlorox (dilution disinfectant – water; 1:32) are cheap and efficient. Phenolic disinfectants, such as Lysol (1:32), are also cheap and efficient. Glutaraldehydes, such as Cidex (1:50), are cheap and among the most efficient disinfectants. Quaternary ammonium, such as Roccal (1:200), is cheap but only moderately efficient. Iodophores, such as Betadine and Metadine (undiluted), are surgical antiseptics; they are more expensive but very efficient. Hot water of at least 77º to 82º C can also be used for disinfection. Please refer to more precise lists of characteristics of common disinfectants in Crissey (1998), Dierauf (1990), and Dierauf & Gulland (2001). Survey respondents recommended to rotate the different disinfectants to avoid any possible microbial resistance. Microbiological swabs from sinks, buckets, and various locations in the kitchen can be taken randomly to confirm the efficiency and consistency of sanitising procedures.

Gully traps and drains should also be cleaned and sanitised daily to remove food debris. Their number and location have to provide efficient drainage. They will not be concealed, and the grates will be removable and easy to clean. Grinders located under the sinks help to eliminate food debris and prevent clogging of drainpipes. Both grinders and pipes will be carefully cleaned, maintained, and sanitised to avoid pests.

Fish waste should not be kept in the same refrigerator as fresh food, and it will be disposed of daily. The disposal of waste should comply with regulations. Parts of fish, such as heads, providing they are of sufficient quality, can be given to nearby zoos or aviaries as bird food or to local fishermen to use as bait (survey).

Literature Cited

Amundin, M. (1986). Breeding the bottle-nosed dolphin *Tursiops truncatus* at the Kolmarden Dolphinarium. *International Zoo Yearbook*, *24/25*, 263-271.

Anonymous. (1979-1984, 1995). *Specifications for the humane handling, care, treatment and transportation of marine mammals* (9 CFR, Subpart E, §3.100-118). Washington, DC: U.S. Department of Agriculture, Animal and Plant Health Inspection Service.

Anonymous. (1992). *Guidelines for the care, holding and transport of marine mammals in Queensland* (Draft). Queensland, Australia: Queensland Department of Primary Industries (QDPI). 39 pp.

Barros, N. B., & Odell, D. K. (1990). Food habits of bottlenose dolphins in the Southern United States. In S. Leatherwood & R. R. Reeves (Eds.), *The bottlenose dolphin* (pp. 309-328). New York: Academic Press.

Burrows, A., Schreib, S., & Smith, T. (1990). Sole use of non-food reinforcers in daily training sessions with an Amazon river dolphin, *Inia geoffrensis*. *Proceedings of the 18th IMATA Conference* (pp 206-210), Chicago, Illinois.

Crissey, S. D. (1998). *Handling fish fed to fish-eating animals: A manual of standard operating procedures*.

Washington, DC: U.S. Department of Agriculture, Agricultural Research Service, National Agricultural Library, Animal Plant Health Service. 21 pp.

Dierauf, L. (1990). Appendix 4: Characteristics of common disinfectants. In L. Dierauf (Ed.), *CRC handbook of marine mammal medicine: Health, disease, and rehabilitation* (p. 706). Boston: CRC Press.

Dierauf, L. A., & Gulland, F. M. D. (Eds.). (2001). *CRC handbook of marine mammal medicine* (2nd ed.). Boston: CRC Press. 1,063 pp.

Dunn, J. L. (1990). Bacterial and mycotic diseases of cetaceans and pinnipeds. In L. Dierauf (Ed.), *CRC handbook of marine mammal medicine: Health, disease, and rehabilitation* (pp. 73-87). Boston: CRC Press.

Evans, P. G. (1987). *The natural history of whales and dolphins*. New York: Facts on File. 343 pp.

Geraci, J. R. (1972). Dietary disorders in marine mammals: Synthesis and new findings. *Journal of the American Veterinary Medicine Association*, *179*, 1183-1189.

Geraci, J. R. (1986). Marine mammals: Nutrition and nutritional disorders. In M. E. Fowler (Ed.), *Zoo & wild animal medicine* (pp. 760-764). Philadelphia: W. B. Saunders Company.

Geraci, J. R., & Ridgway, S. H. (1991). On disease transmission between cetaceans and humans. *Marine Mammal Science*, *7*(2), 191-194.

Kannan, K., Senthilkumar, K., Loganathan, B. G., Odell, D. K., & Tanabe, S. (1997). Elevated accumulation of tributyltin and its breakdown products in bottlenose dolphins (*Tursiops truncatus*) found stranded along the U.S. Atlantic and Gulf Coasts. *Environmental Science and Technology, 31*(1), 296-301.

Kastelein, R. A., Staal, C., & Wiepkema, P. R. (2003). Food consumption, food passage time, and body measurements of captive Atlantic bottlenose dolphins (*Tursiops truncatus*). *Aquatic Mammals, 29*(1), 53-66.

Klinowska, M. (1991). *Dolphins, porpoises and whales of the world. The IUCN red data book.* Gland, Switzerland, and Cambridge, UK: IUCN. 429 pp.

Klinowska, M., & Brown, S. (1986). *A review of dolphinaria*. London: Department of the Environment. 247 pp.

Leatherwood, S., & Reeves, R. R. (1983). *The Sierra Club handbook of whales and dolphins*. San Francisco: Sierra Club Books. 309 pp.

Logerwell, E. A., & Schaufler, L. E. (2005). New data on proximate composition and energy density of Steller sea lion (*Eumetopias jubatus*) prey fills seasonal and geographic gaps in existing information. *Aquatic Mammals, 31*(1), 62-82.

Overath, H., Hartmann, M. G., & Gellissen, M. (1997). *Bacteriological studies of the water of the Duisburg Zoo Dolphinarium with respect to fish food as a source of contamination.* Duisburg: Forshungszentrum Jülich GmbH and Duisburg Zoo.

Reddy, M., Kamolnick, T., Curry, C., Skaar, D., & Ridgway, S. (1994). Energy requirements for the bottlenose dolphin (*Tursiops truncatus*) in relation to sex, age and reproductive status. *Marine Mammals: Public Display and Research*, *1*, 26-31.

Ridgway, S. H., & Harrison R. J. (Eds.). (1989). *Handbook of marine mammals. Vol. 4: River dolphins and the larger toothed whales*. London: Academic Press. 442 pp.

Ridgway, S. H., & Harrison R. J. (Eds.). (1994). *Handbook of marine mammals. Vol. 5: The first book of dolphins and porpoises.* London: Academic Press. 416 pp.

Ridgway, S. H., & Harrison R. J. (Eds.). (1999). *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises*. London: Academic Press. 486 pp.

Skoch, E. J. (1990). Heavy metals in marine mammals: Presence and analytical methods. In L. Dierauf (Ed.), *CRC handbook of marine mammal medicine: Health, disease, and rehabilitation* (pp. 127-138). Boston: CRC Press.

Sweeney, J. C. (1993). *Dolphin quest protocol for veterinary health care management of* Tursiops *&* Steno. San Diego: Dolphin Quest.

Sylvestre, J. P. (1985). Some observations on behaviour of two Orinoco dolphins *Inia geoffrensis humboldtiana* (Pilleri & Gihr, 1977), in captivity, at Duisburg Zoo. *Aquatic Mammals, 11*(2), 58-65.

Terry, R. P. (1990). A short guide to the biology of the Tucuxi: Lessons from captivity and the wild. *Proceedings of the 17th IMATA Conference*, Harderwijk, The Netherlands.

Worthy, G. A. (1990). Nutritional energetics for marine mammals: Addendums. In L. Dierauf (Ed.), *CRC handbook of marine mammal medicine: Health, disease, and rehabilitation* (pp. 489-520). Boston: CRC Press.

Worthy, G. A. (2001). Nutrition and energetics. In L. A. Dierauf & F. M. D. Gulland (Eds.), *CRC handbook of marine mammal medicine* (2nd ed.) (pp. 791-827). Boston: CRC Press.

Aquatic Mammals 2005, *31*(3), 371-381, DOI 10.1578/AM.31.3.2005.371

8. Husbandry

Good husbandry and animal care are the underlying motivations of all environmental and habitat enhancements. For an animal maintained in a controlled environment, there are many factors that can affect its health and behaviour: social parameters, public display-related factors, breeding, pathogens, age, etc. In the wild, many animal species mask signs of illness to avoid falling prey to other species. This is especially true for cetaceans. Because of the aquatic environment, marine mammals exhibit symptoms of illness very late in the disease process and differently from terrestrial animals (Cowan et al., 2001; McBain, 2001; Sweeney & Reddy, 2001). Therefore, cetacean veterinarians need to spend time observing the animals in their care and adjust their practices to accommodate these animals (McBain, 2001).

The purpose of this chapter is to present a brief overview of the various aspects of husbandry, the necessities of medical training, and the importance of implementing a veterinary programme to maintain the physical and psychological health of cetaceans in human care. It is by no means an exhaustive and detailed reference on diagnosis and treatments. One book is the international and uncontested reference in this field—the *CRC Handbook of Marine Mammal Medicine* (2nd ed.), published in 2001 by Dierauf & Gulland (Eds.). This overview chapter is compiled from this reference, as well as several governmental regulations, such as the USA regulations (Anonymous, 1979-1984, 1995) and the Australian regulations for Queensland (Anonymous, 1992), and it also presents some results from the international survey of cetacean facilities. Husbandry also relies heavily on adequate training and conditioning, and frequent references to these areas will be made throughout this chapter. For professional vocabulary, training methods, and problem solving, the international reference is Ramirez (1999).

Husbandry and Veterinary Care

The basic foundation for a successful captive cetacean group is preventive medicine (Allen, 1996). Husbandry training can help minimise stress and encourage the animal's cooperation. The frequent and regular monitoring of physiological and behavioural parameters allows for early diagnosis and treatment of any health condition (Sweeney, 1993). Variation from usual behaviour often occurs at the beginning of many disease episodes. Hence, evaluation of an animal's behaviour in its environment is of crucial importance both in the diagnosis of disease and in preventing the consequences of disease. Husbandry problems with cetaceans in human care come partly from exhibiting animals in an enclosed environment. This presents a challenge to curatorial staff and veterinarians (Sweeney, 1990).

Monitoring and Examination

Due to the fact that early recognition of behavioural symptoms facilitates prompt medical diagnosis and treatment, it is recommended that all animals be monitored regularly, which includes a daily visual assessment and a regularly scheduled full physical assessment. Sweeney (1993) recommends a full examination every three months.

Husbandry staff and trainers are very important in the daily observation of the animals and the careful recording of each animal's behaviour, appearance, and food intake. Through their daily interactions, they develop the ability to detect abnormal cetacean behaviour, change in appetite, and motivation. They are responsible for alerting the veterinarian if they detect abnormal behaviours, loss of appetite for 24 h, or a change in body appearance. Efficient and frequent communication between husbandry staff, trainers, and veterinarians is important to ensure a rapid transfer of information (McBain, 2001). They should comply with the training program established by the curator and veterinarian, which includes husbandry and handling techniques, reporting data, and keeping records. It is important to motivate husbandry and training staff to develop their skills through courses and participation in international symposiums and conferences.

Every facility should have a veterinarian responsible for the health of the animals or one who is quickly available when not directly attached to the institution. Depending on his or her experience, this person will have ready contacts with other experts and specialised professionals. The veterinarian is responsible for establishing the medical protocol, the examination schedule, the healthcare programme, the behavioural conditioning to facilitate animal handling, the dietary goals (e.g., food type, quantity, caloric content, and vitamin supplements), the administration of vaccines, the management of pregnancy and birth control, the therapy and treatment of disease, and the control of records. In addition, the veterinarian will control the hygiene of humans and the life support system parameters. The veterinarian and/or curator serves as the liaison with the Convention on International Trade in Endangered Species of Wild Fauna and Flora (CITES) and various other regulatory offices (see “Regulatory Offices and Organisms” section).

The clinical examination of a cetacean includes its history, the examination of the animal, and its environment. To understand what is considered normal, it is essential to know the life, social history, and clinical records of an animal. Visual examination and behavioural observation, both from the surface and under water, also are important to understand how the animal feels. An animal’s behavioral changes with its conspecifics or trainers can reveal that it is not feeling well (McBain, 2001). Lack of appetite, abnormal swimming or buoyancy, reduced activity, increased respiratory rate, unusual breath odour, among others, are symptoms signalling the possible onset of disease. Problems related to stressful social interactions can be exhibited by behavioural and physiological symptoms as well such as spontaneous vomiting, foreign body consumption, self-inflicted trauma, rake marks, excessive tooth wear, aggressiveness, excessive sexual behaviour, submission, nervousness, failure to perform trained tasks, and destructive behaviour (Sweeney, 1990; Stoskopf & Gibbons, 1994; survey).

After reviewing the animal’s history and performing a visual, hands-on examination, a routine medical examination is given, which usually includes documenting physical parameters such as weight, size, mouth inspection, blood, animal exudates, and fluids for cytology examination. Fluids from the blowhole, stomach, and intestinal tract, as well as urine and faeces, provide valuable measures of health conditions (Sweeney, 1990; Sweeney & Reddy, 2001). Ultrasonography and, to a lesser degree, radiography are additional diagnostic aids (McBain, 2001). For details on training procedures and result interpretations, please refer to McBain (2001) and Sweeney & Reddy (2001).

When a disease from a bacterial, viral, fungal, or parasitic origin is diagnosed, the veterinarian is responsible for determining the adequate therapy and the methods of medication administration. He or she might be assisted by dedicated trainers in the case of prolonged treatment, restraint, and any treatment requiring behavioural training. Access to sick animals can be greatly facilitated by appropriate medical facilities such as an elevating platform or pool bottom, a sliding scale, or a shallow medical pool (see Chapter 5 for details).

Medical Training

Animals trained to accept routine veterinary examinations and sample collections without coercion will be less stressed and more easily monitored. Until recently, examination and collection procedures required that the animal be physically restrained. Such procedures can have a negative effect on the behaviour of the animal and can cause the alteration of samples due to stress (Sweeney & Reddy, 2001). Additionally, there are risks of trauma and injury to the animals and trainers during handling. Conditioning techniques requiring only a few weeks to a few months of training allow medical procedures to be performed reliably without restraint. Regular repetition maintains these behaviours and allows for routine health monitoring (Abel, 1986; Sweeney, 1990; Sweeney & Reddy, 2001). Gating and mattress or stretcher training, although not really considered medical training, are equally important in animal husbandry. Nevertheless, this is mainly true for monitoring, sample collections, some endoscopic examinations, bandaging, and small surgical procedures (Lacave et al., 2003; survey). Unfortunately, when an animal is very sick, it is often reluctant to perform medical behaviours, which makes it necessary to catch and restrain the animal for treatment. Still, medical husbandry training is highly advisable as a routine and standard part of every training program. Both animals and keepers benefit from trained procedures because they greatly reduce stress, handling time, and costs, allowing an earlier diagnosis of medical conditions and, therefore, reducing the chances of severe illnesses and potential loss of an animal.

Institutions are encouraged to formalise their behavioural training and management plan in writing, detailing the objectives and goals, the methods, and the evaluation criteria. Professional organisations, such as the European Association for Aquatic Mammals (EAAM) (Anonymous, 2005e), the International Marine Animal Trainers Association (IMATA) (Anonymous, 2005i), and the Alliance of Marine Mammals Park and Aquariums (AMMPA) (Anonymous, 2005a), can assist by proposing criteria for the evaluations and training programs. Training is a very useful tool to enhance research, education, and environmental and behavioural enrichment.

Sample Collections

Table 8.1 presents a list of the physiological and behavioural examinations and sample collections that are performed in the surveyed institutions and recommended to be part of a routine medical management plan (Sweeney, 1986, 1993; Dunn, 1990; survey); however, the selection of parameters and their frequency remains the choice of the treating veterinarian. The role of the veterinarian also is to decide what shall be recorded and collected by trainers or by the veterinarian him- or herself.

Records

A detailed and easily accessible database of all records for each animal should be kept as part of

Figure 8.1. Blood sample

Figure 8.2. Blowhole sputum sample

the preventive healthcare programme. Storage of frozen blood and serum for all individuals may be necessary for future analysis. Regular collection of samples allows for the establishment of a strong database. The processing and analysis will be done using consistent techniques and by the same laboratories.

Comprehensive records help to document the animal's history and should be available to the veterinarian. Records are kept on an individual basis, preferably in database applications that are easy to examine and compare with other establishments' records or for scientific purposes. It is

Figure 8.3. Milk sample

Figure 8.4. Stomach sample

Figure 8.5. Stretcher training

preferable to keep back-up copies that are stored safely (Anonymous, 2003). An animal moving to new locations will be accompanied by copies of its records. Records for each individual contain the following information:

- The species name
- Date of birth
- Animal identification, specific markings, and photographic identification
- Origin in captivity or in the wild; parents if known

- Previous location, if any
- Growth and development; morphometric log (weight and body size measurements)
- Diagnostic ultrasound log, haematology, and cytology findings; veterinary observations and treatments; medication log
- Social behaviours and status; known incompatibilities and unusual or interesting behaviour; behaviours during training, performances, and handling
- Breedings, mates, outcomes, and offspring identification
- Date of death and results of post-mortem analysis

A routine report is prepared on all animals, including behaviour, health observations, treatments administered, transfer to other enclosures, time of feeding, quality and amount of food, feeding behaviour, the animal's birth and/or death, and the arrival of new animals. The staff on duty, any maintenance carried out or required, and the results of water and environmental quality tests also are recorded daily. Training received by each animal and the animal's progress also should be recorded.

Table 8.1. Frequency of physiological and behavioural tests and records (W=Week, M=Month, Y=Year)

Parameters and tests	Frequency
Activity level and behaviour	Daily
Food intake	Daily
Vitamin intake	Daily
Eye, mouth, and blowhole inspection	Daily
Respiration monitoring	Daily
Body weight	1/W to 4/Y
Body dimensions (morphometrics), growth rate	1/M to 4/Y
Heart rate	1/W to 4/Y
Body temperature	1/W to 4/Y
Blood (haematology)	1/M to 4/Y
Blowhole fluids (sputum)	1/W to 4/Y
Faecal fluids	2/M to 4/Y
Eye mucus	1/M to 4/Y
Gastric fluids	1/M to 4/Y
Urine	2/M to 4/Y
Parasite control	4/Y
General physical examination	4/Y
Ultrasound check	4/Y to 1/Y; regularly during pregnancy
Endoscopy or X-ray	1Y based on prior history
Milk sample	When animal is lactating

Institutions are encouraged to analyse their records and publish their findings in recognised journals, through international professional organisations, and through symposiums, with the objective to promote better husbandry and encourage scientific research.

Handling

Sometimes an animal needs to be segregated from others. This can be the case for newly arrived animals as a quarantine measure or for an animal suspected of being ill and contagious. Any communicable disease has to be remedied before the animal is placed with other animals unless they were together during incubation.

Institutions will find it necessary to have adequate quarantine facilities. Isolation pools are only efficient if the location is remote from the other pools and if the LSS is also separate. Any enclosure that has contained an animal with an infectious or contagious disease should be cleaned and sanitised.

How cetaceans will be handled and restrained needs to be decided and supervised by curators and veterinarians and should be done as quickly and carefully as possible by trained and experienced staff in a manner that does not cause physical harm for the animal or the staff involved. The length and invasiveness of the procedure will be anticipated to predict which drugs and analgesics to use, the stress involved, and possible degree of discomfort. During restraint, the animal's vital signs will be monitored. Behavioural training, such as desensitisation and habituation, also can assist in procedures involving physical restraint. Experience and acclimation attenuate the response to potentially stressful procedures (St. Aubin & Dierauf, 2001). When cetaceans are removed from the water, care should be taken to prevent skin abrasion; water should be constantly sprayed to prevent hyperthermia.

The curatorial staff will be attentive in placing animals together that are compatible. They should not be co-housed with animals that could cause them stress or discomfort or those that could interfere with their good health by inappropriate or aggressive behaviour. Experienced veterinarians and curatorial staff are able to identify and resolve social, hierarchical, and behavioural problems. Whenever behaviour causes harm or injury to vulnerable animals, such as neonates or juveniles, the offending animal preferably will be isolated or removed from the area until the problem is solved. Apart from removal, caretakers can lessen compatibility problems to some extent by extinguishing the aggressive behaviour through training techniques—not introducing new animals into an established group, removing the dominant animal, reintroducing a new animal only after the other animals are established, or introducing new animals into an established group as a pair or a group (Geraci, 1986). If the decision is made to remove the animal permanently, proper housing will be provided. This animal should not be deprived of the company of other animals, however.

Cetaceans should be allowed to rest between performances. They can be rotated to ensure that they rest adequately and perform readily. The regulations of different countries may vary greatly on this issue. An animal that suffers from a disease or does not behave adequately will not participate in performances or swim programmes until its condition is remedied. This includes animals that are reluctant to perform.

Breeding

Nowadays, every institution should consider promoting breeding to maintain a stable, global captive population. Laws and regulations in most countries forbid the collection of wild animals and favour captive breeding programmes. Species in danger of extinction are of particular concern. Yet, many populations are kept in small groups or same-sex groups, therefore, excluding these animals from contributing to the collective captive gene pool (Odell & Robeck, 2002). An efficient animal management programme is encouraged to achieve long-term population stability.

Nevertheless, breeding will be considered only if the welfare of newborn animals is ensured by an adequate environment. Maintenance of pregnant females and females with a calf requires facilities to have adequate spatial requirements with which to isolate animals. Breeding will be supervised by a veterinarian with expert knowledge in reproduction. Reproductive strategies and physiology can vary significantly among cetacean species (Robeck et al., 2001). The reproductive status of males and females can be monitored by hormonal status and/or by ultrasonography and regular check-ups performed during pregnancy and the rearing of the calf. The genetic origin of newborn animals has to be identified to control the diversity of the captive population. Precious information on reproductive physiology and anatomy can be obtained from routine veterinary examinations. Mating patterns and sexual and rearing behaviours can be recorded to enhance the understanding of the reproduction of the species.

Whenever possible, the veterinarian and training staff will be present during birthing to gain experience in parturition and post-natal care, to record the event and monitor the timing and physiological parameters of the mother and calf, and to provide assistance and support in case of a problem. Contingency plans should be developed by the veterinarian when pregnancy is detected in case of emergency situations. Adequate safeguards will be implemented to maximise the survival of offspring, including prevention action to ensure that the young are not subject to injury by other individuals in cases such as improper rearing by the mother or if the newborn is weak or does not nurse well. If hand-rearing is chosen for medical reasons, adequate facilities will be used to provide easy access to the young animal, and it will be kept in the company of its mother or a surrogate female. During the past decades, some facilities have faced the difficulties of breeding and the resulting loss of a calf. By communication and information sharing, improvements have been made in pregnancy detection and management, in environmental enhancements for birthing and neo-natal care, and in post-natal care and calf husbandry.

Inbreeding is one of the problems of captive groups (Klinowska & Brown, 1986). This is not acceptable in view of available techniques for population management such as segregation, contraception, artificial insemination, and possible relocation. The veterinarian can assist an institution in creating a plan for population management and breeding control. The role of the European Endangered species Programme (EEP), European Studbook (ESB), or a Species Survival Plan (SSP) is to help institutions in the management of captive populations to maintain genetic diversity.

Please refer to, among others, Robeck (2001) and Odell & Robeck (2002) for information on reproduction and artificial insemination and to Townsend & Gage (2001) for information on hand-rearing of a calf. Reproduction is one of the fast evolving topics in the care and maintenance of marine mammals. Artificial insemination has seen many positive results during the past few years. Many articles have been published in various marine mammal science, veterinary, or medicine journals. Workshops and symposia by the European Association for Aquatic Mammals (EAAM), the International Association for Aquatic Animal Medicine (IAAAM), the International Marine Animal Trainers Association (IMATA), the Society for Marine Mammalogy (SMM), and other professional organisations recently have been organised on these subjects. These organisations have website links that discuss artificial insemination (Anonymous, 2005e, 2005h, 2005i, 2005k). Two chapters of the *CRC Handbook* by Dierauf (2001) and Dierauf et al. (2001) provide extensive online references and links to marine mammal websites. Two journals are dedicated to marine mammal sciences: (1) *Aquatic Mammals*, published by the EAAM, and (2) *Marine Mammal Science*, published by the SMM. Both are accessible online (see the *Aquatic Mammals* and

Marine Mammal Science websites [Anonymous, 2005c, 2005j]).

Contact with Cetaceans and Hygiene

Until the last decade, only scientists, rescue workers, veterinarians, and trainers had close contact with wild and captive cetaceans. Many facilities now offer interactive and "Swim-With-The-Dolphins" (SWTD) programmes, dolphin-assisted therapy programmes, or allow the feeding and petting of dolphins. These programmes increase contacts between humans and cetaceans and, therefore, the possibilities of disease transmissions. Transmission of diseases from cetaceans to humans seems rare and hypothetical; however, taking appropriate precautions for protection through proper hygiene may prevent future cases of infections. Examples of infections are mainly bacterial and fungal infections as a few are known pathogens in humans. Viral, protozoal, and fungal infections are rarer, and, in most documented cases, come from handling wild stranded or captured animals (Cowan et al., 2001). Even less is known about diseases that are transmittable from humans to marine mammals; however, strict rules of hygiene should govern contacts between humans and cetaceans, even for the general public, such as the compulsory washing of hands before feeding fish to the animals; using disinfectant footbaths when entering an enclosure with a marine mammal; and regularly cleaning and disinfecting wet suits, toys, and equipment.

Other hygiene rules will include the control of insects, ectoparasites, and avian or mammalian pests. It is preferable to prohibit the access of pets in pool areas. Insecticides or other chemical agents should not be applied unless specifically recommended by the veterinarian.

Death

In the event of the death of an animal, a complete necropsy should be conducted by a qualified veterinary pathologist as soon as possible—within 24 h. A necropsy report will be prepared, listing all lesions observed and the cause(s) of death. All diagnostic tests will be listed in the report and the results recorded. Relevant authorities should be notified of the animal's death. The remains of the animal can be made available for further research by authorised institutions and disposed of in a manner consistent with public health practice (e.g., incinerated, buried, or donated to a museum).

Mixed Species Exhibits

Mixed-species grouping occurs in the wild. Commonly associated species are bottlenose dolphins, pilot whales, spotted dolphins, Risso's dolphins, rough-toothed dolphins, and humpback whales (Wells & Scott, 1999; Herzing et al., 2003). In human care, exhibits of mixed cetacean species have been successful. A wide range of species can be compatible as long as ample surface area, volume, and structural variability are provided to allow some natural separation of habitat space (Klappenback, 1988; Sweeney & Samansky, 1995). Bottlenose dolphins, Pacific white-sided dolphins, common dolphins, rough-toothed dolphins, Risso's dolphins, pilot whales, and false killer whales have been successfully housed together, but they also all have mated with a bottlenose dolphin and produced hybrids—some of them living for several years. The "wolphin" Kekaimalu, a female hybrid bottlenose dolphin x false killer whale, also gave birth to three offsprings sired by bottlenose dolphins. Several female bottlenose dolphins also have given birth to four hybrids sired by a long-beaked common dolphin, and one of these hybrid calfs later gave birth to a backcross calf sired by a male bottlenose dolphin, again showing that dolphin hybrids are fertile (Odell & McClune, 1999; Zornetzer & Duffield, 2003). It is interesting to note that this is a cross between two genera, which should not be possible under currently accepted taxonomic classifications.

Hybridisation among cetacean species has been documented in the wild between several of the above-mentioned species, as well as between Dall's porpoise and harbour porpoise, blue whale and fin whale, and possibly between beluga whale and narwhal (Sylvestre & Tasaka, 1985; Arnason et al., 1991; Heide-Jorgensen & Reeves, 1993; Baird et al., 1998), but it seems to be rare. While this situation may indicate social compatibility, hybridisation should be avoided when cetaceans are kept in human care and especially when managing endangered species (Sweeney & Samansky, 1995). As in the wild, social and territorial dominance can be exhibited in captivity; when inadequate space is provided, the dominant species have displayed traumatic aggression toward subdominant counterparts, regardless of the species (Sweeney & Samansky, 1995).

Many species of cetaceans and pinnipeds cohabitate in the wild and are commonly seen in mixed-species groupings. The survey yielded contradictory results regarding the mixing of cetaceans and pinnipeds. Some facilities have reported incompatibilities between sea lions, seals, and bottlenose dolphins whereas others (a majority) have reported that the presence of sea lions is considered a valuable environmental enrichment. They provide a harmless distraction, and they are seen chasing and playing with each other. Adequate space and haul-out areas, as well as proper fencing, should be provided if dolphins and pinnipeds are exhibited together.

Rescue and Rehabilitation

Occasionally, institutions might be involved in the rescue and rehabilitation of stranded cetaceans. A facility should participate in rehabilitation only if adequate accommodations are available. Rescued animals will not be housed in the same enclosure as resident animals because they can carry contagious diseases or parasites and contaminate resident animals. Specific serological tests, such as *Brucella* and *Morbillivirus*, should be carried out upon arrival for strict quarantine and prior to release to receive clearance from authorities to proceed. Rescued animals will be housed in a quarantine facility specifically built for this purpose. The LSS also preferably will provide separately treated water. Rescued animals that are diagnosed with an illness should not be housed in natural facilities with other animals as the water system does not prevent the spread of disease. Staff will respect strict hygiene protocols to prevent healthy residents from being infected.

Animals that are diagnosed as unreleasable will have to be housed permanently, meaning that the institution might have to provide new accommodations or transfer the animal to another facility.

Rescued animals usually require extra care, observation time, and a lot of dedication from the staff (survey). Cetaceans all belong to protected species and are subject to special regulations concerning rehabilitation and release. Animals belonging to endangered species might be subject to even stricter rules. Special effort can be made to gain scientific information that could help protect the species in the wild. Its reintroduction should be monitored medically and behaviourally.

Laboratory

Most of the bigger facilities that house a large number of cetaceans or other marine mammals have an in-house veterinary clinic, including a laboratory, a surgery room, and a necropsy room that is equipped with a freezer for carcasses.

Smaller facilities do not always have clinics, but they usually do have a laboratory. Several surveyed facilities recommend to have an on-site laboratory that is equipped to run cytological analysis and, if possible, haematological and biochemical analyses as well. Considering the cost of equipment and the required trained staff, this might be possible only in the larger facilities. Unfortunately, external analyses are costly and might discourage preventive medicine.

If outside laboratories perform the analyses, they should use established techniques to ensure the consistency of measured parameters. Prior arrangements can be made with other facilities, zoos, and local veterinary hospitals to use their surgery or necropsy rooms and equipment should the need arise.

Sweeney (1993) recommended these minimal laboratory items:

- Basic medical supplies: syringes, needles, tubes, vials, flexible stomach tubes, funnels, etc.
- Pharmacy cabinet
- Microscope
- Centrifuge
- Bacteriological incubator
- Medical box with emergency kit
- Portable scale or means of assessing animal weight (that can be installed on pool-side)
- Autoclave
- Equipment to perform water quality tests, including coliform counts

Other equipment, such as an endoscope and a portable ultrasound machine, can belong to either the institution or to the consulting veterinarian, but it is recommended that there be access to one. Additional equipment may include a sample freezer (-70° C), a bench or walk-in type freezer to store carcasses, a laminar flow cabinet, a stainless steel table with drain, and a portable X-ray machine. The medical laboratory preferably will be separated from any staff areas for better hygiene, and it should be equipped with a computer to store biological and behavioural data.

Research Laboratory

Research is vital to any effective conservation programme and is recommended to be included in the design of new marine mammal facilities (Klinowska & Brown, 1986; Sweeney & Samansky, 1995). It can consist of at least one room, preferably air conditioned, dry, and properly drained located adjacent to a pool, with an underwater window for the observation and monitoring of animals, as well as for conducting experiments

Figure 8.6. Dolphin clinic (Photograph from I. Smit)

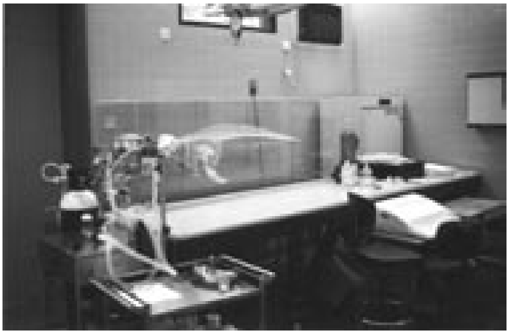

Figure 8.7. Surgery room

Figure 8.8. Laboratory equipment

Figure 8.9. Laboratory

requiring an underwater view such as acoustic and behavioural experiments. Cabling and storage for hydrophones, video cameras, a computer, a communication system between the upper deck and trainers' room, Internet connections, and underwater lighting also can be included in the design (for more details, see Chapter 5).

Transportation

Transportation of animals from one facility to another is a long and potentially stressful operation for the animals. It involves precise logistics, high costs, extensive personnel, and usually several transportation methods such as cargo airplanes, trucks, and sometimes boats. Techniques and equipment have been developed over the past 30 years to cope with the unique physiology of cetaceans, thus ensuring safe and successful transportation. They are reviewed in detail by Antrim & McBain (2001) and by the Alliance of Marine Mammal Parks and Aquariums (Anonymous, 2003) and will not be discussed here. Nevertheless, transportation of cetaceans is subject to regulations in most countries and, therefore, both the national and international regulations of the countries involved must be followed.

In a large facility, dolphins and whales sometimes have to be moved between pools or for medical or husbandry purposes. Transportation can be facilitated by the layout of the pool surroundings, and adequate equipment can be available in or near the enclosure (e.g., a lifting pool bottom, a crane, a hoist, or a beaching area). To facilitate transportation, animals can be trained to slide into a stretcher, which can then be lifted manually by trainers or by a hoist or a crane, and then transported by truck if necessary. Because it is a stressful operation for the animal, it should be avoided unless deemed necessary for animal management or veterinary purposes. Animals can be desensitised through training, thereby reducing stress and potential harm to the animal and staff.

Regulatory Offices and Organisms

Cetacean movements are subject to the Convention on International Trade in Endangered Species of Wild Fauna and Flora (CITES), which is an agreement between governments (Corkeron, 2002). Its aim is to ensure that international trade in specimens of wild fauna and plants does not threaten their survival. Today, 167 countries are members. These species are listed in three different appendices, depending on their status. Appendix I lists species threatened with extinction and for which trade is permitted only in exceptional circumstances.

Appendix II includes species not necessarily threatened with extinction, but for which trade must be controlled to avoid utilisation incompatible with their survival. Appendix III lists species that are protected in at least one country, which has asked other CITES parties for assistance in controlling the trade (Anonymous, 2005d). All cetacean species are included in the CITES Appendices I or II; therefore, all imports, exports, re-exports, and introductions from the sea are controlled through a licensing system. Exchange or trade of wild-born cetaceans between institutions belonging to member countries are subject to CITES permits. Animals bred in captivity are subject to permit variations. CITES has been in operation in the European Union since 1984. In 1997, two new regulations replaced the old legislation, which since that date are the core of the EU wildlife trade legislation with some differences from the CITES texts (Anonymous, 2005g).

The World Conservation Union (IUCN) is an international union of 140 countries, which establishes a Red List of Threatened Species. The IUCN members originally adopted the resolution that allowed the CITES agreement in 1973. This list provides the conservation status of each species threatened with extinction. It also proposes action plans (Reeves et al., 2003) and publications with more extensive listings such as Klinowska (1991), and a newsletter of the Cetacean Specialist Group from the Species Survival Commission.

Apart from these international regulations, each country may be subject to other regulations—either national, federal or regional—concerning housing, breeding, care, and transport. Some countries lack regulations because of a lack of institutions housing cetaceans. They should refer to other countries' regulations before establishing their own. In the USA, legislation governing marine mammals is quite complex because of the requirements of the Marine Mammal Protection Act, Endangered Species Act, and the Animal Welfare Act (please refer to Young & Shapiro, 2001, for more details). For other regulations, please contact professional organisations and national legislation websites.

Many institutions housing cetaceans belong to the professional organisations mentioned in the above paragraphs, which promote exchanges of knowledge and strive to ensure better care, improve housing standards, and enhance communications for maintaining genetic diversity through collection management.

In Europe, the EAAM is the professional organisation dedicated to marine mammals. It is responsible for the coordination of breeding programs, population management, and conservation strategies through the European Endangered Species Programme (EEP), the most intensive type, and the European Studbook (ESB), along with other organisations such as the European Association of Zoos and Aquaria (EAZA) (Anonymous, 2005f). The Taxon Advisory Group (TAG)'s task is to develop Regional Collection Plans that describe which species are recommended to be kept, why, and how to manage them. Together with the Species Committee, coordinators make recommendations to member institutions for animal placements to encourage breeding and preserve genetic diversity. It also is the privileged representative interlocutor with regulatory offices in the EU and other European countries, as well as environmental associations or professional organisations. These organisations also promote public awareness of wildlife conservation issues, organise campaigns on conservation of selected species, and reintroduce captive-bred wildlife into restored or secured habitats.

In the USA, similar organisations, such as the American Zoo and Aquarium Association (AZA) (Anonymous, 2005b), manage the equivalent of EEPs and ESBs—the SSPs and Population Management Programs (PMPs). AMMPA is an international association of marine parks, aquaria, zoos, research facilities, and professional organisations dedicated to the highest standards of care for marine mammals and to their conservation in the wild through public education, scientific study, and wildlife presentations (Anonymous, 2005a). AMMPA gives accreditations and serves as a representative in many regulation negotiations.

Literature Cited

Abel, R. S. (1986). Husbandry and training of captive dolphins. In M. M. Bryden & R. H. Harrison (Eds.), *Research on dolphins* (pp. 183-188). Oxford: Clarendon Press.

Allen, M. E. (1996). Introduction: Basic husbandry. In D. G. Kleiman, M. E. Allen, K. V. Thompson, S. Lumpkin, & H. Harris (Eds.), *Wild mammals in captivity: Principles and techniques* (pp. 1-3). Chicago: The University of Chicago Press.

Anonymous. (1979-1984, 1995). *Specifications for the humane handling, care, treatment and transportation of marine mammals* (9 CFR, Subpart E, §3.100-118). Washington, DC: U.S. Department of Agriculture, Animal and Plant Health Inspection Service.

Anonymous. (1990). *Supplement to the Secretary of State standards of modern zoo practice; additional standards for UK Cetacean keeping*. United Kingdom Secretary of State.

Anonymous. (1992). *Guidelines for the care, holding and transport of marine mammals in Queensland* (Draft). Queensland, Australia: Queensland Department of Primary Industries (QDPI). 39 pp.

Anonymous. (2003). *Alliance of Marine Mammal Parks and Aquariums: Standards and guidelines*. Alexandria, VA: AMMPA. 38 pp. Available online: www.ammpa.org. Accessed 18 July 2005.

Anonymous. (2005a). Alliance of Marine Mammal Parks and Aquariums (AAMPA) website. Available online: www.aampa.org. Accessed: 18 July 2005.

Anonymous. (2005b). American Zoo and Aquarium Association (AZA) website. Available online: www.aza.org. Accessed: 18 July 2005.

Anonymous. (2005c). *Aquatic Mammals* website. Available online: www.eaam.org/aquamamm.htm. Accessed: 18 July 2005.

Anonymous. (2005d). Convention of International Trade in Endangered Species of Wild Fauna and Flora (CITES) website. Available online: www.cites.org. Accessed: 18 July 2005.

Anonymous. (2005e). European Association for Aquatic Mammals (EAAM) website. Available online: www.eaam.org. Accessed: 18 July 2005.

Anonymous. (2005f). European Association of Zoos and Aquaria (EAZA) website. Available online: www.eaza.net. Accessed: 18 July 2005.

Anonymous. (2005g). European Community and Trade in Wild Fauna and Flora website. Available online: http://europe.eu.int/comm/environment/cites/home_en.htm. Accessed: 18 July 2005.

Anonymous. (2005h). International Association for Aquatic Animal Medicine (IAAAM) website. Available online: www.iaaam.org. Accessed: 18 July 2005.

Anonymous. (2005i). International Marine Animal Trainers Association (IMATA) website. Available online: www.imata.org. Accessed: 18 July 2005.

Anonymous. (2005j). *Marine Mammal Science* website. Available online: www.marinemammalogy.org. Accessed: 18 July 2005.

Anonymous. (2005k). The Society for Marine Mammalogy (SMM) website. Available online: www.smm.org. Accessed: 18 July 2005.

Antrim, J., & McBain, J. (2001). Marine mammal transport. In L. A. Dierauf & F. M. D. Gulland (Eds.), *CRC handbook of marine mammal medicine* (2nd ed.) (pp. 882-891). Boston: CRC Press.

Arnason, U., Spilliaert, R., Palsdottir, A., & Arnason, A. (1991). Molecular identification of hybrids between the two largest whale species, the blue whale (*Balaenoptera musculus*) and the fin whale (*B. physalus*). *Hereditas, 115*, 183-189.

Baird, R. W., Willis, P. M., Guenther, T. J., Wilson, P. J., & White, B. N. (1998). An intergeneric hybrid in the family Phocoenidae. *Canadian Journal of Zoology, 76*, 198-204.

Corkeron, P. (2002). Captivity. In W. F. Perrin, B. Würsig, & J. G. M. Thewissen (Eds.), *Encyclopedia of marine mammals* (pp. 192-197). London: Academic Press.

Cowan, D. F., House, C., & House, J. A. (2001). Public health. In L. A. Dierauf & F. M. D. Gulland (Eds.), *CRC handbook of marine mammal medicine* (2nd ed.) (pp. 767-778). Boston: CRC Press.

Dierauf, L. (Ed.). (1990). *CRC handbook of marine mammal medicine: Health, disease, and rehabilitation*. Boston: CRC Press. 735 pp.

Dierauf, L. A. (2001). The electronic whale. In L. A. Dierauf & F. M. D. Gulland (Eds.), *CRC handbook of marine mammal medicine* (2nd ed.) (pp 117-126). Boston: CRC Press.

Dierauf, L. A., & Gulland, F. M. D. (Eds.). (2001). *CRC handbook of marine mammal medicine* (2nd ed.). Boston: CRC Press. 1,063 pp.

Dierauf, L. A., Frasca, S., Jr., & Mashima, T. (2001). Careers in marine mammal medicine. In L. A. Dierauf & F. M. D. Gulland (Eds.), *CRC handbook of marine mammal medicine* (2nd ed.) (pp. 97-115). Boston: CRC Press.

Dunn, J. L. (1990). Bacterial and mycotic diseases of cetaceans and pinnipeds. In L. Dierauf (Ed.), *CRC handbook of marine mammal medicine: Health, disease, and rehabilitation* (pp. 73-87). Boston: CRC Press.

Geraci, J. R. (1986). Marine mammals: Husbandry. In M. E. Fowler (Ed.), *Zoo & wild animal medicine* (pp. 757-760). Philadelphia: W. B. Saunders Company.

Heide-Jorgensen, M. P., & Reeves, R. R. (1993). Description of an anomalous monodontid skull from West Greenland: A possible hybrid? *Marine Mammal Science, 9,* 258-268.

Herzing, D. L., Moewe, K., & Brunnick, B. J. (2003). Interspecies interactions between Atlantic spotted dolphins, *Stenella frontalis* and bottlenose dolphins, *Tursiops truncatus*, on Great Bahama Bank, Bahamas. *Aquatic Mammals*, *29*(3), 335-341.

Joseph, B. E., Asper, E. D., & Antrim, J. E. (1990). Marine mammal transport. In L. Dierauf (Ed.), *CRC handbook of marine mammal medicine: Health, disease, and rehabilitation* (pp. 543-551). Boston: CRC Press.

Klappenback, S. (1988). The training and implementation of multi-species marine mammal show. *Proceedings of the 16th IMATA Conference* (pp. 104-106), San Antonio, Texas.

Klinowska, M. (1991). *Dolphins, porpoises and whales of the world: The IUCN red data book*. Gland, Switzerland, and Cambridge, UK: IUCN. 429 pp.

Klinowska, M., & Brown, S. (1986). *A review of dolphinaria*. London: Department of the Environment. 247 pp.

Lacave, G., Jenson, A. B., Bossart, G. D., Ducatelle, R., Salbany, A., Ghim, S., Roque, L., Pereira, M., Massei, K., Ova, I., Romao, N., & Lima, C. (2003). Identification of genital papilloma in two female *Tursiops truncatus*, one with pseudorcarcinomatous proprieties, after voluntary biopsies. *31st EAAM Annual Symposium*, Tenerife, Canary Islands, Spain.

McBain, J. F. (2001). Cetacean medicine. In L. A. Dierauf & F. M. D. Gulland (Eds.), *CRC handbook of marine mammal medicine* (2nd ed.) (pp. 895-906). Boston: CRC Press.

Odell, D. K., & McClune, K. M. (1999). False killer whale, *Pseudorca crassidens* (Owen, 1846). In S. H. Ridgway

& R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises* (pp. 213-244). London: Academic Press.

Odell, D. K., & Robeck, T. R. (2002). Captive breeding. In W. F. Perrin, B. Würsig, & J. G. M. Thewissen (Eds.), *Encyclopedia of marine mammals* (pp. 188-192). London: Academic Press.

Ramirez, K. T. (1999). *Animal training: Successful animal management through positive reinforcement*. Chicago: John G. Shedd Aquarium. 578 pp.

Reeves, R. R., Smith, B. D., Crespo, E. A., & Notarbartolo di Sciara, G. (2003). *Dolphins, whales and porpoises: 2002-2010 conservation action plan for the world's cetaceans*. Gland, Switzerland: IUCN Species Survival Commission, Cetacean Specialist Group. 139 pp.

Robeck, T. R., Atkinson, S. K. C., & Brook, F. (2001). Reproduction. In L. A. Dierauf & F. M. D. Gulland (Eds.), *CRC handbook of marine mammal medicine* (2nd ed.) (pp. 193-236). Boston: CRC Press.

St. Aubin, D. J., & Dierauf, L. A. (2001). Stress and marine mammals. In L. A. Dierauf & F. M. D. Gulland (Eds.), *CRC handbook of marine mammal medicine* (2nd ed.) (pp. 253-269). Boston: CRC Press.

Stoskopf, M. K., & Gibbons, E. F. (1994). Quantitative evaluation of the effects of environmental parameters on the physiology, behavior, and health of animals in naturalistic captive environments. In E. F. Gibbons, E. J. Wyers, E. Waters, & E. W. Menzel (Eds.), *Naturalistic environments in captivity for animal behavior research* (pp. 140-160). Albany: State University of New York Press.

Sweeney, J. C. (1986). Infectious diseases. In M. E. Fowler (Ed.), *Zoo & wild animal medicine* (pp. 777-781). Philadelphia: W. B. Saunders Company.

Sweeney, J. C. (1990). Marine mammal behavioral diagnostics. In L. Dierauf (Ed.), *CRC handbook of marine mammal medicine: Health, disease, and rehabilitation* (pp. 53-72). Boston: CRC Press.

Sweeney, J. C. (1993). *Dolphin Quest protocol for veterinary health care management of* Tursiops *&* Steno. San Diego: Dolphin Quest.

Sweeney, J. C., & Reddy, M. L. (2001). Cetacean cytology. In L. A. Dierauf & F. M. D. Gulland (Eds.), *CRC Handbook of marine mammal medicine* (2nd ed.) (pp. 437-448). Boston: CRC Press.

Sweeney, J. C., & Samansky, T. (1995). Elements of successful facility design: Marine mammals. In E. F. Gibbons, Jr., B. S. Durrant, & J. Demarest (Eds.), *Conservation of endangered species in captivity: An interdisciplinary approach* (pp. 465-477). Albany: State University of New York Press.

Sylvestre, J. P., & Tasaka, S. (1985). On the intergeneric hybrids in cetaceans. *Aquatic Mammals, 11*(3), 101-108.

Townsend, F. I., & Gage, L. J. (2001). Hand-rearing and artificial milk formulas. In L. A. Dierauf & F. M. D. Gulland (Eds.), *CRC handbook of marine mammal medicine* (2nd ed.) (pp. 829-849). Boston: CRC Press.

Young, N. M., & Shapiro, S. L. (2001). U.S. federal legislation governing marine mammals. In L. A. Dierauf & F. M. D. Gulland (Eds.), *CRC handbook of marine mammal medicine* (2nd ed.) (pp. 741-766). Boston: CRC Press.

Wells, R. S., & Scott, M. D. (1999). Bottlenose dolphin, *Tursiops truncatus* (Montagu, 1821). In S. H. Ridgway & R. J. Harrison (Eds.), *Handbook of marine mammals. Vol. 6: The second book of dolphins and porpoises* (pp. 137-182). London: Academic Press.

Zornetzer, H. R., & Duffield, D. A. (2003). Captive-born bottlenose dolphin × common dolphin (*Tursiops truncatus* × *Delphinus capensis*) intergeneric hybrids. *Canadian Journal of Zoology/Revue Canadienne de Zoologie*, *81*(10), 1755-1762.

Aquatic Mammals 2005, *31*(3), 382-385, DOI 10.1578/AM.31.3.2005.382

Appendix

List of cetacean facilities worldwide in 2005

Continent/region	Country	Facility name	Number of facility
Europe			Subtotal 50
	Belgium	Boudewijn Seapark	1
	Bulgaria	Dolphinarium Varna Firm Lazur	1
	Denmark	Fjord & Belt Centret	1
	Finland	Särkänniemi Delfinaario	1
	France	Marineland Antibes	2
		Parc Asterix	
	Germany	Allwetterzoo Munster	4
		Heidepark Soltau	
		Tiergarten Nürnberg	
		Zoo Duisburg	
	Italy	Aquario di Genova	6
		Delfinario Rimini	
		Gardaland	
		Oltremare	
		Zoomarine Italy	
		Zoosafari Delfinario Fasano	
	Lithuania	Center of Marine Culture – Klaipeda	1
	Malta	Marineland Ltd.	1
	Portugal	Jardim Zoologico de Lisboa	2
		Zoomarine Mundo Aquatico	
	Romania	Constantza Dolphinarium	1
	Russia	Bolshoy Utrish – Anapa	8
		Dolphinarium St-Petersburg	
		Gelendzhik Dolphinarium	
		Maly Utrish – Novorossiysk	
		Rostov-na-Donu Dolphinarium	
		Utrish Dolphinarium – Moscow	
		Utrish Dolphinarium Aquatheater – Sochi	
		Yessentuki Dolphinarium	
	Spain	Aquapolis, Delfinario Vila Seca	10
		Loro Parque	
		Marineland Cataluna	
		Marineland Majorca	
		Marineland Tenerife	
		L'Oceanografic de Valencia	
		Mundomar	
		Parque Zoologico de Barcelona	
		Selwo Marina	
		Zoo Aquarium de La Casa del Campo	
	Sweden	Kolmårdens Djurpark	1
	Switzerland	Conny Land	1
	The Netherlands	Dolfinarium Harderwijk	1
	Ukraine	Aquamarine – Sevastopol	8
		Dolphinarium Odessa	
		Dolphinarium Partenit – Alushta	

		Dolphinarium Yalta	
		Dolphinarium Yevpatoria 1	
		Dolphinarium Yevpatoria 2	
		Karadag Biostation – Sudak	
		State Oceanarium of Ukraine – Sevastopol	
North America			Subtotal 41
	Canada	Marineland of Canada	2
		Vancouver Aquarium	
	USA	Aquarium for Wildlife Conservation – New York	39
		Brookfield Zoo	
		Clearwater Marine Aquarium	
		Dolphin Cove	
		Dolphin Quest Hawaii, Oahu, Kahala Mandarin Oriental	
		Dolphin Quest Hawaii, Big Island, Hilton Waikoloa	
		Dolphin Research Center	
		Dolphins Plus	
		Epcot – Walt Disney World – The Living Seas	
		Gulf World	
		Gulfarium	
		Hawaii Institute of Marine Biology	
		Hershey Park	
		Indianapolis Zoo	
		John G. Shedd Aquarium	
		Knott's Berry Farm	
		Long Marine Laboratory	
		Marine Life Oceanarium (Marine Animal Productions)	
		Marineland of Florida	
		Miami Seaquarium	
		Minnesota Zoo	
		Mote Marine Laboratory	
		Mystic Marinelife Aquarium	
		National Aquarium in Baltimore	
		Pittsburgh Zoo	
		Point Defiance Zoo Aquarium	
		Sea Life Park – Dolphin Discovery	
		SeaWorld – California	
		SeaWorld – Florida	
		SeaWorld – Discovery Cove	
		SeaWorld – Texas	
		Six Flags Marine World – California	
		Six Flags World of Adventure – Ohio	
		Texas State Aquarium	
		Theater of the Sea	
		The Dolphin Connection	
		The Dolphin Institute – Kewalo Bassin	
		The Mirage Dolphin Habitat	
		U.S. Navy Marine Mammal Program – Spawar San Diego	
Central and South Americas, and the Carribean Islands			Subtotal 43
	Anguilla	Dolphin Fantaseas	1
	Antigua	Dolphin Fantaseas	1
	Argentina	Mar del Plata	2

		Mundo Marino	
	Bermuda	Dolphin Quest Bermuda	1
	Colombia	Acuario y Museo de Mar – El Rodadero en Santa Marta	2
		Islas del Rosario, Cartagena de Indias	
	Cuba	Acuario de Baconao	4
		Acuario de Bahia de Naranjo – Holguin	
		Aquario Nacional de Cuba	
		Delfinario de Cienfuegos	
	Dominican Republic	Manati Park	1
	Honduras	Roatan Institute for Marine Sciences – Anthony's Key	1
	Jamaica	Dolphin Cove	1
	Mexico	Acuario Aragon Convimar	21
		Acuario de Veracruz	
		Akua'n Kay	
		Aletta Bay Dolphin Interactive Facility	
		Amigos del Mar	
		Atlantis	
		Delfiniti Ixtapa	
		Dolphin Adventure	
		Dolphin Discovery Cozumel	
		Dolphin Discovery Isla Mujeres	
		Dolphin Discovery Puerto Aventuras	
		Jungla Magica – Feria Chapultepec	
		La Isla Interactive Aquarium	
		La Paz Interactive Dolphin Parque	
		Parque Acuatico Cici Acapulco	
		Parque Marino Atlantis	
		Parque Nizuc, Wet and Wild	
		Reino Aventura	
		Selva Magica	
		Via Delphi Xcaret	
		Xel-Ha	
	Peru	Cilde – Hotel Los Delfines	1
	The Bahamas	Dolphin Encounters	2
		The Dolphin Experience	
	Tortolla	Prospect Reef Resort	1
	Venezuela	Aquarium de Valencia J.V. Seijas	4
		Diver Land – Margarita Island	
		Mundo Marino	
		Parque Zoologico "El Pinar"	
Asia/Pacific			Subtotal 59
	Australia	Pet Porpoise Pool	2
		SeaWorld Australia	
	China and Hong Kong	Baiji Dolphin Aquarium	7
		Beijing Landa Aquarium	
		Qingdao Ocean Park Dolphinarium	
		Guangzhou Dolphinarium	
		Ocean Park – Hong Kong	
		Ocean World Guangzhou	
		Xiangtan Heping Park Dolphinarium – Shanghai	
	French Polynesia	Moorea Dolphin Center	1
	Indonesia	Dolphin Lodge – Batam	2
		Gelangang Samudra Jaya Ancol	
	Japan	Amakusa Aquarium	39

		Aomori Prefectural Asamushi Aquarium	
		Awashima Marine Park	
		Dolphin Base	
		Echizen Matsushima Aquarium	
		Enoshima Aquarium	
		Futami Sea Paradise	
		Inubosaki Marine Park	
		Irukajima/Ise-Shima	
		Izu-Mito Sea Paradise	
		Kamogawa SeaWorld	
		Katsurahama Aquarium	
		Keikyo Aburatsubo Marine Park	
		Kinosaki Marine World	
		Marine World Uminonakamichi	
		Marinepia Matsushima Aquarium	
		Minamichita Beach Land Aquarium	
		Misaki Amusement Park Aquarium	
		Miyajima Public Aquarium	
		Nanki Shirahama Adventure World	
		Niigata City Aquarium – Marinepia Nihonkai	
		Noboribetsu Marine Park Nixe	
		Notojima Aquarium	
		Oarai Aquarium	
		Okinawa Expo Aquarium	
		Okinawa Marine Research Center	
		Osaka Kaiyukan Aquarium	
		Otaru Aquarium	
		Shima Marineland	
		Shimoda Aquarium	
		Shimonoseki Municipal Aquarium	
		Shinagawa Aquarium	
		Suma Aqualife Park	
		Sunshine International Aquarium	
		Taiji Whale Museum	
		Toba Aquarium	
		Yashima Sea Palace	
		Yokohama Hakkeijima Sea Paradise	
		World Dolphin Resort	
	Korea	Seoul Grand Park	1
	New Zealand	Marineland of New Zealand	1
	Palau	Dolphins Pacific	1
	The Philippines	Ocean Adventures Park	1
	Singapore	Underwater World	1
	Taiwan	Ocean World	1
	Thailand	Oasis Sea World	2
		Safari World	
Africa			Subtotal 4
	Egypt	Dolphinella (Utrish) – Sharm el Sheikh	2
		Media Production City – Cairo Dolphinarium	
	South Africa	Bayworld Complex (formerly Port Elizabeth Oceanarium)	2
		uShaka Marine World (formerly SeaWorld – Durban)	
Middle East			Subtotal 2
	Bahrein	Dolphin Park	1
	Israel	Dolphin Reef Eilat	1
			TOTAL 199

Subscription Rates for

Aquatic Mammals

Effective June 2005 – May 2006

Circle type of subscription desired.

Printed version, 4 issues per year (approximately 375 pages):
Library: $230 USD
Agency: $215 USD
Individual, student, or retired colleague: $115 USD

Online access through Ingenta's Web page, 4 issues per year (Articles are searchable and printable from the Web page.):
Library: $150 USD
Agency: $130 USD
Individual: $50 USD
Student or retired colleague: $35 USD

CD Version, 4 CDs per year:
Library: $150 USD
Agency: $130 USD
Individual: $60 USD
Student or retired colleague: $40 USD

Printed and online versions, 4 issues per year:
Library: $350 USD
Agency: $300 USD
Individual: $140 USD
Student or retired colleague: $125 USD
(Students: Include a copy of current student ID with order.)

Sold to:

____
____
____
____

Ship to:

____
____
____
____

Submit payment as check in U.S. dollars or by Visa or MasterCard to . . .

Aquatic Mammals
Horrabin Hall 59
Western Illinois University
Macomb, Illinois 61455, USA
E-mail: GR-Colley@wiu.edu
Telephone: (309) 298-1917
Fax: (309) 298-2869

Subscription cost:	$_____USD
Shipment outside N. Am., add	$_____15 USD
Payment by wire transfer, add	$_____25 USD
Total Charge:	$_____USD

Credit Card Number: __ __ __ __ -__ __ __ __-__ __ __ __   Expiration: _____/_____

Name on Credit Card: ____ (Circle one.)

Receipt available upon request.

Back Issues of *Aquatic Mammals* (Volume 21 to Present)

Now Available on CD, with 3 Issues per Volume

Volume(s) Ordered:

____ 21 (1995)
____ 22 (1996)
____ 23(1997)
____ 24 (1998)
____ 25 (1999)
____ 26 (2000)
____ 27 (2001)
____ 28 (2002)
____ 29 (2003)
____ 30 (2004)

Printed Version (72 USD) × ___ volumes (3 issues per volume)	________	USD
CD Version (36 USD) × ___ volumes (3 issues per CD)	________	USD
Discount for multiple printed versions (-12 USD per volume)	________	USD
Discount for multiple CD versions (-6 USD per CD)	________	USD
Shipment within North America (3 USD × __ volumes)	________	USD
Shipment outside North America (4 USD × __volumes)	________	USD
Total	________	**USD**

Mail, Fax, or E-mail Payment to . . .

Aquatic Mammals
c/o Gina Colley
Document and Publication Services
Horrabin Hall 59
Western Illinois University
Macomb, Illinois 61455 USA
E-mail: GR-Colley@wiu.edu
Telephone: (309) 298-1917
Fax: (309) 298-2869

Credit Card Number: __ __ __ __ -__ __ __ __-__ __ __ __ Expiration: _____/_____

Name on Credit Card: ____ (Circle one.)

Sold to: ________________________________ Ship to: ________________________________

________________________________ ________________________________

________________________________ ________________________________

________________________________ ________________________________

E-mail: ________________________________

Receipt available upon request.