Marine Mammals Laboratory Manual

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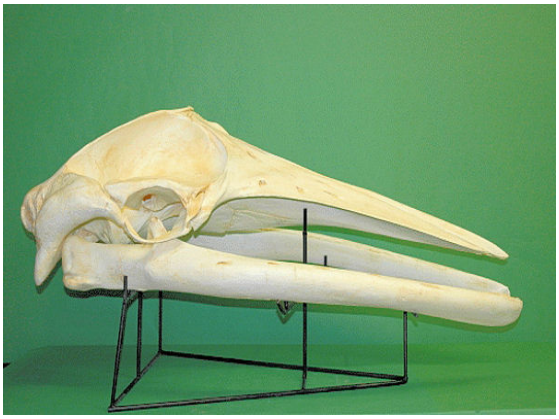


INTRODUCTION

This is a manual specifically produced for students interested in developing their knowledge on marine mammals though hands-on experiences. Through this course we offer students the opportunity to (1) sharpen their knowledge of the diversity of marine mammals, (2) understand the functional anatomy of these creatures, and (3) better comprehend the their evolutionary features. To those ends, students will be exposed to a variety of specimens of marine mammals in the form of casts and actual osteological material, carcasses, dissections, baleens, marine mammal products, pictures, and sounds. By the end of this course students should have mastered the systematics and numerous aspects of the biology of these organisms.

WARNING!

All animal carcasses or parts used in this class come from animals found dead or that have been euthanized for humane reasons. They have been obtained with the appropriate permits issued by the National Marine Fisheries Service and/or the U.S. Fish and Wildlife Service. Those permits are available upon request.

All animal carcasses must be handled with care and students must wear surgical gloves, masks, goggles, and a lab coat or any other clothing-protecting gear at all times during dissections.

Marine mammal carcasses are general safe to handle. Occasionally some animals may have been infected with morbiliviruses or other infectious pathogens. The instructor will notify students if that is the case and students may choose not to participate directly in the exercise. They will not be penalized for making such decision based on health concerns.

All wounds or cuts in the skin of the students must be fully bandaged and examined by the instructor prior the beginning of the exercise. Any cut or injury that occurs during the dissecting procedures must be reported to the instructor immediately and must be cleaned, disinfected, and bandaged at once. The instructor must take note of the incident. If medical attention is required, the provider must be informed that the person in question was exposed to a marine mammal carcass.

Any portion of the skin that was exposed to marine mammal fluid and/or tissue must be thoroughly scrubbed with soap before leaving the lab.

PART 1: Marine Mammal Dissections

1.1. Purpose of this exercise

This exercise is aimed at guiding in the necropsy procedures for studying whole or parts or marine mammals in the Marine Mammal Laboratory Class taught at Arkansas State University. The reason why we do dissections early on in this course is because we want to mount the skeletons of these animals and the whole process of cleaning and mounting is a long one that takes about the entire semester.

Besides the educational value for the students in conducting a necropsy, the main purpose of this kind of exercises is to determine the cause of death of the animal being examined. To that end we will examine the gross anatomy, tissues, and chemicals in the specimen. If the final results are of any scientific value, they will be published with all the students participating in the exercise appearing as co-authors.

1.1.2. Materials

The following material will be needed in order to perform dissections of all marine mammals of small to medium size:

1.1.2.1. Individual materials

1. This manual
2. A notebook
3. Report forms
4. Clipboards
5. Pencils #2
6. Black ink pens
7. Sharpie markers (black ink)
8. Surgical gloves (to avoid direct contact between the student and the carcass)
9. Masks (decomposing carcasses usually generate foul smell, particularly cetaceans)
10. Goggles (To avoid any fluid from the carcass to directly or indirectly contact the eyes of students).
11. Lab coat / apron (clothes that the student does not mind to be stained are also recommended)
12. Biohazard suits in case of dealing with carcasses that might be infected with pathogens that can be dangerous to humans (e.g., morbiliviruses).

1.1.2.2. General equipment

13. First aid kit

14. Dissecting table
15. A plastic pocket to collect refuse materials drained from the dissecting table
16. Large scale full carcass weight
17. Videocamera and tapes to record the procedure
18. Digital camera to record the procedure
19. An identification book for the group of species being examined
20. An anatomy book/papers

1.1.2.3. Collecting material

21. Label tape (www.WVR.com)
22. Packaging tape
23. Blank tags and white Tiveck (www.LSS.com)
24. Laundry tags (www.golps.com) Daily delivery tag, 1-ply fiber
25. 2 x 3, 3 x 5, 4 x 6, 8 x 10, 9 x 13 write-on zip lock bags
26. 4 x 6 whirlpack bags
27. **2** 1.0L bottles filled ¾ with 10% NB formalin for histological samples
28. **1** 250ml – 1000ml bottle filled ¾ with 10% NB formalin for life history samples
29. **1** ½ gallon seal tight container for stomach contents
30. DMSO vial
31. Plastic centrifuge tubes (10-20mL)
32. Syringes (various sizes)
33. Aluminum foil
34. Acetone
35. EtOH and saline
36. Culture swabs (aerobic and anaerobic)
37. Butane flame
38. Cutting board
39. Sharpening steel
40. **5** toothed forceps
41. **3** Stainless steel dissecting scissors
42. 5 sharp stainless steel knives (6” – 9”)
43. Box of scalpel blades
44. **5** scalpel blade handles
45. Scalpel blade remover
46. Flat head screwdriver (tooth extractor)
47. Stryker saw
48. Chisel
49. Hammer
50. Cotton string
51. Small pan scale for tissue weights (mg or g)
52. Tape measure
53. Rules (in cm)
54. Calipers
55. A saw and bone cutters
56. Jars of different sizes with lids

57. Ethanol (70%) to preserve soft tissues
58. Bouin's fixative or formalin

1.1.2.4. Disposal material

59. Heavy duty construction bags to collect materials either for disposal or for storage
60. Paper towel
61. 30-gallon barrel for soft tissue disposal.

Some critical thinking questions

1. Why is it so important to wear protective clothing when handling marine mammals?
2. Why do we videotape dissecting procedures?

1.2. Necropsy Procedures

1.2.1. Necropsy Procedure

1. All specimens received come frozen and are kept that way until the day they are necropsied. These specimens should be de-frozen prior beginning the procedures. The time to do that will vary from specimen to specimen and will depend mostly on the size of the animal. An elephant seal pup or a sea otter will take about 48 hours; a porpoise about 60 hours at room temperature.
2. Begin by identifying the species being examined.
3. Take notes of the basic (stranding) information of the specimen. That includes: ID number, species name, location and date in which the specimen was collected.
4. Determine the gender of the animal. Take pictures of the genital area. Measure the distance between the tip of the animal and the genital and anal orifices.
5. Weight the animal by hanging it from a scale suspended from the ceiling or placed on the floor, whatever is more practical.

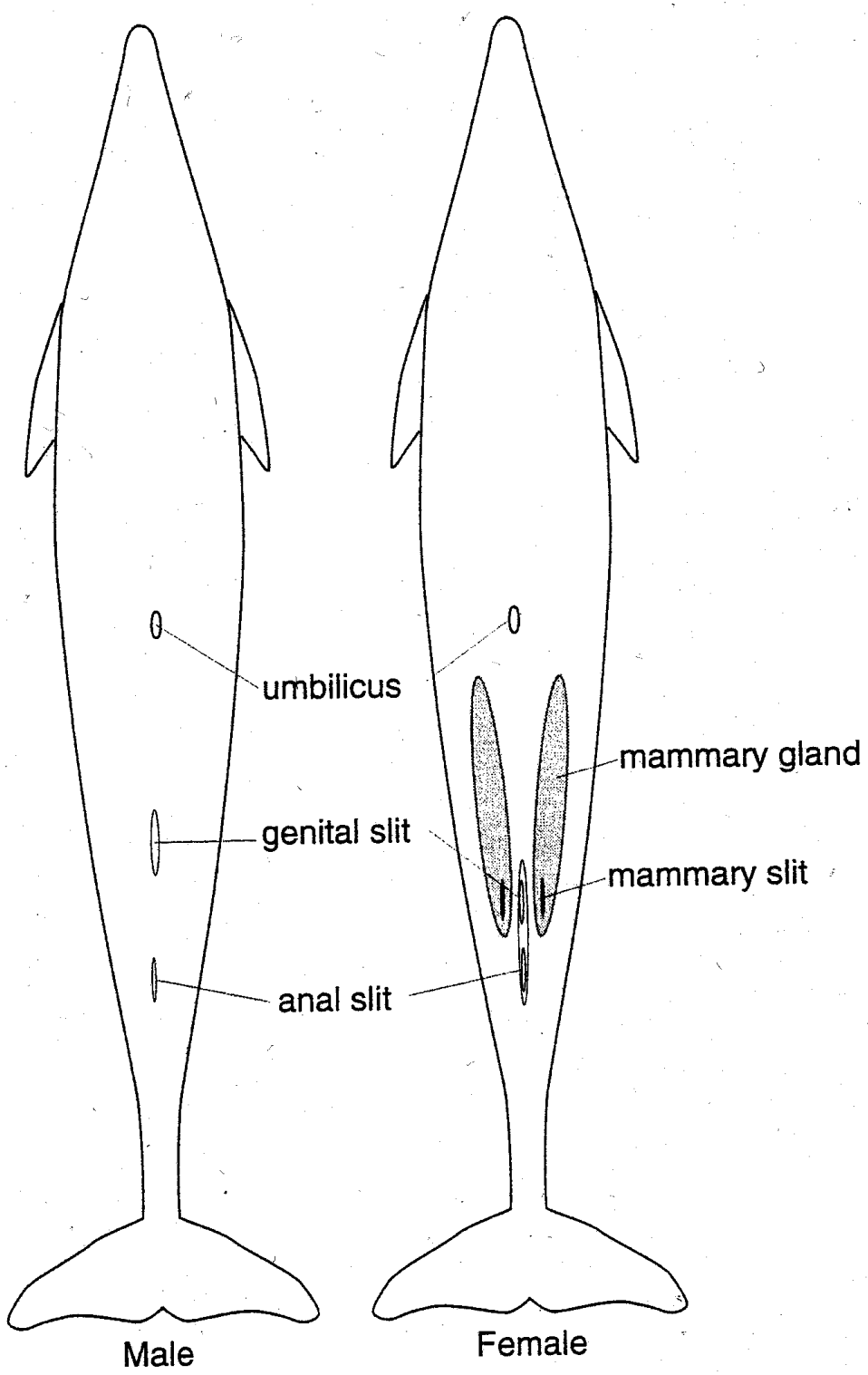


Figure 13.4. External sexual differences of cetaceans.

6. For all animals, place the animal on its left side with its head to the right pectoral fins/feet toward you.
7. Examine body orifices for such abnormalities as discharges or erosions. Examine the eyes for lesions and to assess if the carcass is dehydrated.
8. Examine the skin and hair for abnormalities such as wounds, sores, and abscesses, and signs of bycatch such as rope scars and flippers or flukes cut off noting distribution. Examine the fins/feet. Photograph any abnormality.
9. Note the state of decomposition (fresh, medium, rotten or falling apart).
10. For odontocetes at least four teeth should be collected. For mysteces the ear plug should be collected. Count the rings for each one. Each ring represents one year of age.
11. Any parasite found, either externally or internally, must be identified to at least the levels of being tapeworms, roundworms and the total number estimated. The parasites must be preserved in alcohol (70% ethanol) for further identification at the species level.
12. Elevate the right pectoral fin/front leg and begin a ventral midline skin incision over the sternum and extend it to the chin. Continue the incision caudally to one side of the genitalia. If dealing with a large whale, be careful because if the animal is in severe state of decomposition, gases produced by bacteria may have accumulated internal and could cause the animal to literally explode.
13. For males, examine the penis, examine the scrotal contents and section the testes. In female animals examine and section the mammary glands.
14. Remove the genitalia. The genital tract is cut loose from the behind and first loosened from the body when in females, the ovaries can be identified and collected. Testes are internal and are attached to the dorsal wall of the visceral cavity.
15. Separate the skin from underlying fascia beginning at the chin. Reflect the skin from the thorax and abdomen as far as the dorsal midline. Because it is used for processing, keep the skin/hide intact. The hind limb is reflected preferably by separation of the pubic symphysis or by disarticulation of the coxofemoral joint.
16. Carefully incise the abdominal muscles at the posterior edge of the last rib on the right side without puncturing abdominal viscera. Continue this muscle incision from the xiphoid cartilage to the lateral processes of the lumbar vertebrae, along the ends of these processes, and down to the pelvis. Pull the muscle flap toward you and examine the position and color of the abdominal viscera.
17. Lift the rib cage and incise the diaphragm close to the ribs. The diaphragm should collapse as air enters the thorax indicating that there was negative pressure in the thorax.
18. Cut the costal cartilages close to the sternum with a knife in a young animal, or with bone cutters or a saw in older animals. Cut the ribs close to the vertebrae (an axe will do in the field). Remove the right side of the rib cage.
19. At this time, review the clinical history and survey the exposed viscera. Note major lesions such as peritonitis, pneumonia, malpositioned organs, and hemorrhages. Prepare the specimen bottles and containers that are required. Collect material (e.g. pleural or peritoneal exudates/transudates for microbiology or chemistry before continuing the dissection. Open the pericardial sac. Estimate

volume of any abnormal fluid accumulations. Assess the body condition of the animal on the basis of subcutaneous, intra-abdominal and pericardial fat.

20. Split the mandibular symphysis with a saw or axe and spread apart the ramus. Cut along both sides of the tongue and through the cartilaginous joints of the hyoid bones. With caudal traction, dissect the trachea and esophagus from the neck. Check for jugular or carotid thrombosis at this time. Examine the teeth, tongue, lips, tonsils, retropharangeal lymph nodes and palate for lesions.
21. Remove trachea, esophagus, heart and lungs as a unit by combined caudal traction and mediastinal dissection. Severing the aorta, vena cava and esophagus at the diaphragm will permit the lungs and heart to be removed from the thorax.
22. Examine thyroids, parathyroids and trachea. Open the entire esophagus and examine the mucosa. Open the trachea along its dorsal midline from larynx and continue into large bronchi within lung parenchyma. Note volume and nature of any material within the airways. Make extensive use of palpation to detect focal or diffuse changes in pulmonary texture.
23. Inspect the heart and great vessels for symmetry, position. Transect the ventricles with a single smooth slice at the junction of their middle and lower thirds. Examine the cut surface for ventricular hypertrophy or thinning, for fibrosis, necrosis and thrombosis. Beginning in the posterior vena cava, open the right atrium. Examine the tricuspid valves <u>BEFORE</u> further dissection. Using scissors or a thin knife open the right ventricle along the interventricular septum, starting in the pulmonary artery. Do not cut the pulmonary valves until their structure has been ascertained. The right ventricular wall should be lifted as a U-shaped flap attached dorsally. Open the left atrium, check valve. Open left ventricle along septum. Check left A V valves, open aorta.
24. Examine and remove the omentum.
25. Examine and remove both adrenal glands and spleen.
26. The gastrointestinal tract may be removed *in toto* or it may be partially examined *in situ*. Grasp any portion of small bowel and, with gentle traction, strip it from the mesentery with a sharp knife or scissors. Gradually work proximally and distally until eventually the entire intestine lies rope-like on the table with only the rectum securing it to the carcass. The content and mucosa may then be examined by opening the gut with scissors. Open the stomach along the greater curvature. In ruminants, the small intestine and colon are thrown over the lumbar spine temporarily so that the forestomachs can be removed. A combination of traction and excision will allow the stomachs to roll from the abdomen, remaining connected to the rest of the intestinal tract by the intact duodenum. Open, examine and discard the forestomachs. Strip the remainder of the intestine from mesentery and examine. Open the aorta and follow the major mesenteric branches, checking for patency and parasitic lesions; it is often a lot easier to work back up cecal/colic arteries to cranial mesenteric root. Strip the intestine from the mesentery and examine the mucosa. Be sure to maintain orientation, a major problem in horse intestine.
27. Remove the floor of the pelvis using a saw. Incise the antihilar border of the kidneys and remove the capsule. If required the entire urogenital system may now be removed intact. Otherwise, remove ovaries and uterus, incise and examine.

Check urethra for patency if history warrants it. Incise the urinary bladder and examine it and its contents.

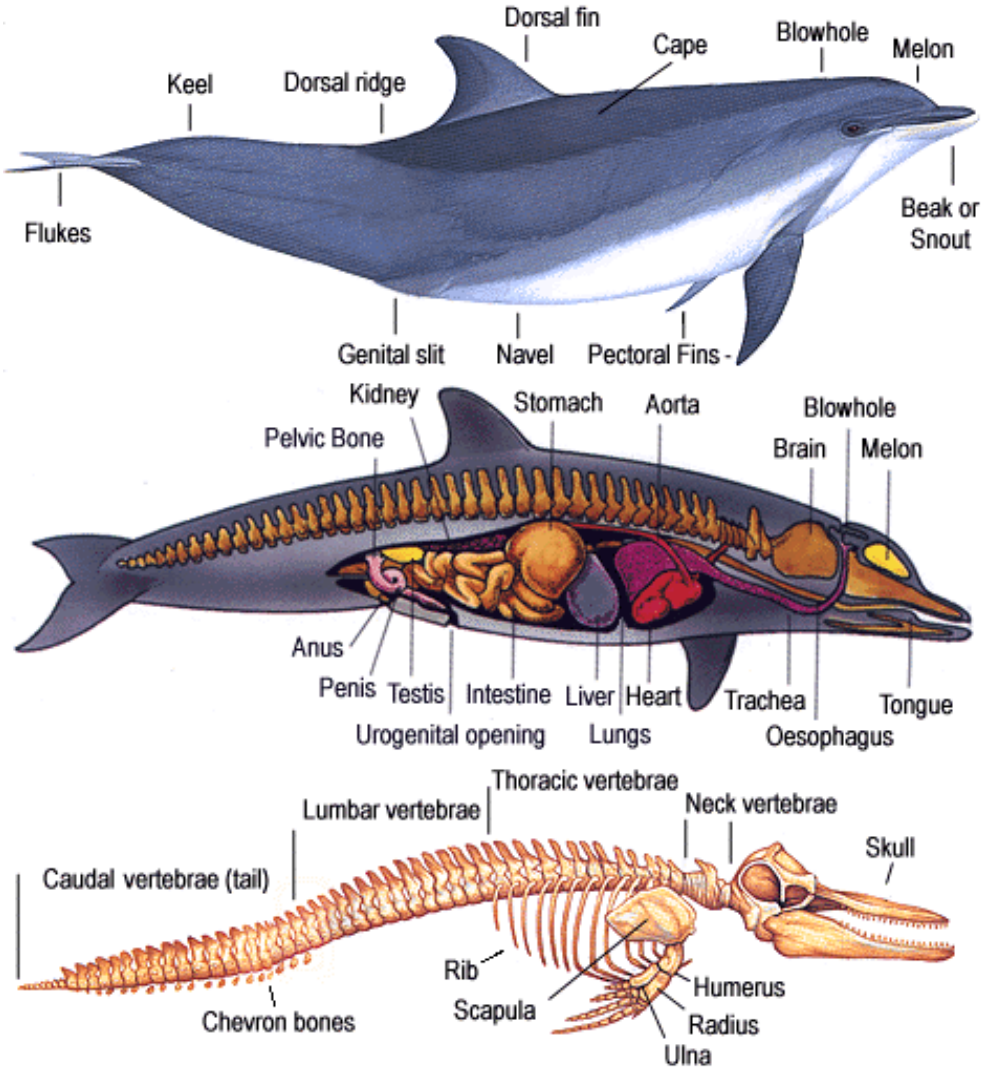
28. Remove and examine the liver. Examine the gall bladder.
29. Skin several joints and open carefully to minimize contamination with bacteria, hair and blood. Evaluate volume and quality of synovial fluid, changes in articular surfaces. Sample for microbiology as required.
30. Remove the head by disarticulation of the atlanto-occipital joint. Remove the skin from the dorsum of the head. Remove the calvarial cap with a handsaw (large species) or bone cutters. Cut transversely across the skull just posterior to the orbits. At right angles to the first cut, make a cut on either side of the calvarium just medial to the orbits and continue each cut posteriorly to join the lateral wall of the foramen magnum at a 45° angle. Pry off the calvarial cap, gently using a chisel if necessary. Remove dura and tentorium cerebelli with scissors.
31. Remove the brain by separating olfactory bulbs from cribriform plate with a spatula (scalpel handle is excellent). Tilt head so that optic nerves can be cut (nose up). Cut infundibulum, branches of carotid artery, and cranial nerve roots to lift brain from the cranial vault.
32. If indicated, eyes are removed by cutting skin away from dorsal and ventral aspects of orbit. Grasping the third eyelid, gently pull forward and ventrally while severing retrobulbar fascia, muscles and optic nerve with curved scissors. Without bruising sclera, remove as much episcleral tissue as possible and drop globe in Bouin's fixative (if fresh) or formalin (if animal have been dead over 4-5 hours.)
33. The spinal cord is removed in small animals by dorsal laminectomy using bone cutters or an electric surgical bone saw. In large animals, a dorsal or lateral laminectomy with a band saw is commonly employed. In practice, a 4-6-cm. segment of cord can be removed with scissors and traction from a spinal segment cut out with a saw or axe. Incise the dura before fixation.
34. Bone and bone marrow may be examined by sawing (or breaking with cutters or by hand) a long bone. Rib tensile strength is subjectively estimated in suspected nutritional diseases.
35. Lymph nodes (e.g. mammary, mesenteric, mediastinal) should be examined with their corresponding systems.
36. Examine several muscle groups.
37. More detailed examination may be warranted to diagnose diseases of some systems, e.g. muscles of respiration for respiratory disease.
38. Tissues collected for toxicology should consist of at least large (e.g., 10 X 10 cm) pieces of blubber, muscle, liver and kidney and then placed in a glass jar rinsed beforehand with hot water and another sample must be stored in a plastic bag.
39. All labeled material should be kept at leas refrigerated if not deep frozen.

Literature consulted

Jauniaux, T.; C. Beans and W. Dabin. 2005. *Stranding, necropsy and sampling: collection data, sampling level & techniques*. Student ECS workshop. 15 pp.

Pugliares, K. R.; A. Bogomolni; K. M. Touhey; S. M. Herzig; C. T. Harry & M. J. Moore. 2007. *Marine mammal necropsy: An introductory guide for stranding responders and field biologists*. Woods Hole Oceanographic Institution Technical Report WHOI-2007-06. 131pp.

Reijnders, P.J.H.; A. Aguilar, and & G.P. Donovan. 1999. Chemical pollutants and cetaceans. *Journal of Cetacean Research and Management* (Special Issue 1). Cambridge, 273pp.



1.3. Skeletal Preparations for Marine Mammals

Although there are many ways to prepare bones for scientific and educational collections, there are few references that outline the various methods. Here we discuss the advantages and disadvantages of different methods in skeletal preparation and articulation. We outline natural and chemical processes, and list contacts or supplies needed for these processes.

1.3.1. Specimen Preparation Techniques

1.3.1.1. Fresh Water Maceration

Maceration is a simple method that is commonly used. Carcasses are soaked in freshwater-filled containers, which accumulate bacteria that aid in flesh decomposition. Be aware that skulls left too long in buckets are subject to bacterial damage. This can be prevented by aerating the water through periodic stirring and addition of more water. Thus, prolonged anaerobic conditions are unfavorable. Choose a container that fits the entire carcass so it can be completely submerged in water. Plastic buckets with lids work best. Some workers add manure to the containers to act as a bacterial step-up. Maceration degreases bones to some extent, however this method has a limited ability to remove oil from large bones, such as cetacean bones. If this oil is not removed, it may leach out over time and result in discolored, odorous bones. Once oily bones have been macerated to remove flesh, they may require further degreasing (see Chemical Maceration section).

Small bones may have their flesh removed quickly, on the scale of a few weeks, while larger bones may take many months for the flesh to decompose away. A skull may soak for a couple of weeks to several months. Cutting away accessible flesh prior to soaking will speed up the process. Interrupting maceration to remove brain and additional flesh accelerates the process. Monitor containers periodically to keep the water level high enough to cover the bones, and to observe the maceration progress. Once the flesh has decomposed, bones should be removed promptly from the maceration water to prevent them from being damaged by bacterial action. Bones of newborn or juvenile animals must be closely monitored, because they tend to fall apart when macerated. Scrubbing and high-pressure hosing can assist with flesh removal. A gentle stream of water may help clean residual tissue from delicate nasal turbinates. After maceration, bones are usually boiled. Care must be taken to not lose teeth and small bones when dumping maceration water or when applying streams of water.

Pros: Natural, no chemicals, inexpensive, less time intensive
Cons: Odorous, may take several months, bones may still need to be degreased

1.3.1.2. Boiling

This method involves cooking bones in boiling water until the flesh falls off. The process works best with bones that were first macerated. Place cleaned skeleton in a detergent solution for approximately 2 weeks to de-grease it, followed by a weak ammonium

hydroxide solution for 1-2 days maximum to bleach the bones. This method requires boiling containers of a size appropriate to the carcass in question. In order to not confuse left and right limb bones, it is best to remove them from the carcass and place them in labeled bags such as stockingettes or other strong boilable flow-through bags. Doing this also helps prevent the loss of tiny bones amongst the debris of boiled flesh. For ease of later assembly, one may also wish to bag other body sections such as the sternum or sections of the vertebral column. Small bones may be boiled in a regular dedicated cooking pot on a hot plate or other heat source in a well-ventilated area (preferably outdoors). Use a lid to keep the water from evaporating during boiling. Keep the water level high enough to cover the bones. Cetacean bones may require a large container such as a 50-gallon barrel and a powerful heat source such as a propane burner. One set up used successfully by Lee Post of Homer Alaska (see additional contacts) has the barrel held up on cement blocks with a propane powered weed burner as the heat source. A metal stovepipe with an elbow bend is used to direct the heat at the bottom of the barrel. In some circumstances, bones too long to fit in the barrel, such as whale jawbones, may be boiled on end and then flipped over to boil the other end. Macerated bones should be boiled until the water no longer has froth. Cooking times vary widely, generally in proportion to the size of the bones. For instance, a medium sized pinniped skull takes 6 to 15 hours of boiling to become clean. Once boiling is complete, bones should have any residual flesh removed by hand and should be allowed to dry thoroughly, ideally in the sun.

Pros: Natural, no chemicals, inexpensive, less time intensive
Cons: Odorous, bones can soften or fall apart especially in young animals, requires very large boiling containers with a sufficient heat source for large cetacean bones

1.3.1.3. Management of the Baleens

Any baleen used for either display or simply for maintaining it in the collection is simply air dried. The rack(s) of baleen must be thorough rinsed with hot water and then set them in front of a fan to make the drying go faster. Heat lamps tend to cause the baleen to warp so they should be avoided. They could also be dried outside by setting a tent of cheesecloth or screening to keep flies and other insects off the baleen. Once the baleens are thoroughly dried one needs to keep an eye out for clothes moths and dermestid beetles. Dried baleen must be kept in museum-style specimen cabinets to protect them from infestation.

1.3.1.4. Saltwater Maceration

Flow-through systems or soaking bones in an open oceanic bay is ideal, but is not an accessible method to all. If access to the ocean is available, clean tissue from skeleton can be placed in shallow waters (attached to point on shore) for 2 to 5 days. The animal parts should be placed in a wire cage with mesh small (e.g., 0.5 cm) enough to capture the smallest bones and staked out to a pier or other stable structure. Marine organisms such as amphipods will consume the flesh and over time (six months to one year) bones

should be void of most flesh. Once the flesh has been eaten away, bones should be dried and bleached in the sun.

Pros: Natural, no chemicals, inexpensive, less time intensive
Cons: Odorous, need to be near saltwater bay or estuary, bones can be lost from enclosures and organisms may consume bones.

1.3.1.5. Dermestid beetles

Dermestid beetle colonies are an efficient method to remove flesh. Several museums with large collections use this method. Beetles ingest flesh and bones. The colony however, must be maintained at 70-80°F in dark, warm conditions and requires numerous specimens to feed them. They can be fed dry dog food in between specimens. Colonies often require a large space (from refrigerator to shipping container size). Beetle colonies are ideal for smaller specimens, but it is difficult to maintain a colony big enough for large cetacean bones.

Before feeding a specimen to a beetle colony, most of the flesh first should be flensed off and the brain and other soft tissues removed. If the specimen is macerated, the brain and soft tissue may be easier to remove. Then, the specimen should be dried. The flesh should have a 'jerky-like' texture. The carcass can be placed in the colony whole or in sections. Place the skeleton in a specially prepared 50 gallon drum containing flesh-cleaning beetles for up to 4 weeks. The colony quickly consumes most of the flesh. For instance, a pinniped skull may take two days and a porpoise skeleton may take two weeks. Smaller specimens should be checked daily; larger specimens can be checked less often. Although beetles will consume the flesh, this method does not degrease the bones. Once the specimen is clean, it should be cleared of remaining larvae either by freezing or by heating.

Pros: Natural, no chemicals, less time intensive, inexpensive (after set up costs)
Cons: Not recommended for large bones, bones still need to be degreased, beetles must be fed when there are no specimens in preparation, set-up costs include obtaining beetles and colony container.

1.3.1.6. Burying

Burying may be the only practical option for large specimens. It is inexpensive (except in labor) and uncomplicated. A large, cumbersome carcass can be buried at the site, deep enough to prevent disturbance and reduce risk to public health, and retrieved months or years later. Burial does have several disadvantages. It is slow (1-10 years), and difficult to monitor (specimens must be dug up periodically to check progress). Small bones, such as flipper or pelvic bones are easily lost. Burying these bones in wire cages or mesh bags may prevent loss. Another option is to remove the flipper bones and prepare them separately. Burial sites must be carefully marked and recorded to prevent losing the entire specimen. Additionally, soil bacteria and chemicals may damage bones. Finally buried bones are quite odorous. Therefore, buried specimens may not be suitable for indoor display.

The general procedure consists in removing tissue from skeleton, placing it in a mesh bag, burying the bag 3 feet underground, and leaving it for 4 weeks

Pros: Inexpensive, ideal for large specimens
Cons: Bones can become lost or decompose, odorous

1.3.1.7. Chemical Maceration

Chemical maceration works quickly, however large tanks and heaters may represent a substantial investment. Specimens prepared by chemical maceration often need no further degreasing or bleaching. Specimens may be completely or partially disarticulated. Removal of specimens from the soak with the intervertebral disks intact greatly facilitates articulation, and produces a more realistic articulation.

Chemical holding tank: Tanks can be salvaged (a discarded bathtub, for example), or built from plywood reinforced at the seams with fiberglass tape and coated with polyester or epoxy resins. Note that tanks will be heavy when filled (800 liters weigh 800 kg); they can be strengthened with 2x4 or 2x6 lumber laid on edge. Tanks can be heated with aquaculture heaters. Heated tanks should be insulated with one inch polystyrene basement insulation (available from lumberyards). Tanks containing corrosive solutions should be fitted with locking covers and warning signs.

Terg-a-zyme is an enzymatic detergent that can be used in a 1% solution (by weight) to strip bones. Terg-a-zyme can be used cold or hot, and can be disposed of down the drain. It is relatively expensive (approximately U.S. $16 for a four-pound container), and relatively aggressive. Specimens must be monitored closely to prevent damage, especially if used hot.

Pros: Can be used cold or hot
Cons: Relatively expensive, can damage bones quickly, especially when used hot

Potassium Hydroxide is available under the name 'caustic potash flake' for approximately U.S. $10 for a 50-pound sack. It should be used at 0.5-1% by weight and kept at approximately 110°F. With the proper heat source, the specimen can be left to macerate. A well-flensed specimen can be degreased and void of flesh in three to five days. Used solution may be neutralized with muriatic (hydrochloric) acid and disposed of down the drain. For all chemicals, please check with local authorities for proper disposal. Finished specimens are rinsed in water and scrubbed in hot soapy water with a nylon bristle brush. Potassium hydroxide slowly decomposes cartilage, therefore sternebrae, intervetertebral disks, and flipper ends are easily preserved. Potassium hydroxide cleans, degreases, and deodorizes bones prepared by other methods, such as burying. Bones from an adult elephant seal soaked in a 0.5% solution subsequent to burying in an attempt to degrease and especially deodorize them showed minor damage after approximately three weeks immersion and were clean enough that indoor mounting was an option.

Pros: One step cleaning, degreasing and bleaching, quick, inexpensive after initial investment
Cons: Requires chemical-resistant tank, heat source

1.3.1.8. Degreasing Agents

The following are chemicals that can be used to remove grease from specimens, in which the flesh has been removed. These chemicals can be used on specimens that were buried, macerated, or placed in a bug colony.

Ammonia is an inexpensive and simple process for degreasing bones. Bones void of flesh can be soaked in a 50% ammonia or ammonia hydroxide and 50% water solution. Soak the specimen for two to seven days. If the solution becomes discolored, change it! Once you remove the specimen, soak it in warm water to flush out the ammonia. Used ammonia can be disposed of down the drain. Bones can also be cooked in ammonia. Simmer bones at low heat in 50/50 ammonia/water solution. Remove fat as it collects on the surface. Allow the bones to dry afterwards.

Pros: Inexpensive, easy disposal, degreases bones
Cons: Caustic

Hydrogen Peroxide soak can whiten and degrease bones, but over time the bones may flake. Use a concentration of 2-5%. Note that most commercial hydrogen peroxide is 35% solution and should be diluted. Simply soak bones void of flesh in hydrogen peroxide for less than one hour. Bones soaked longer than one hour may begin to deteriorate.

Pros: Inexpensive, whitens bones, non toxic
Cons: Bones may flake over time

How are bones stored after being cleaned? either frozen, placed in formalin, or air-dried Skeletons can also be prepared by composting a carcass in manure.

1.3.2. Specimen Articulations

1.3.2.1. Overview and References

When articulating a specimen, a number of decisions must be made prior to beginning the assembly. First, will the specimen be indoors or outdoors? If it is to be outdoors, the bones will need to be coated with epoxy or latex paint for protection from the elements, lest the specimen rapidly decay. Second, will the display be for research purposes, where the bones should be minimally altered, or for educational purposes, where sturdiness might be more important? Third, a pose must be chosen, including whether the specimen will be supported from the top (hanging), the bottom, or the side. It is wise to do a number of sketches of possible poses given the circumstances of the intended display. Look at photographs or watch live animals with the intent of seeing how their skeletal structures move in various positions. Observing emaciated animals can be quite instructive, if the opportunity is available. Examining radiographs can provide valuable information on vertebral spacing and posture, plus unravel the mysteries of wrist and

anklebone placement and spacing. If possible, take radiographs of whole limbs before cleaning the flesh off the bones.

1.3.2.2. Materials

Marine mammal skeletons are generally mounted with a metal rod running from inside the back of the skull, down drilled holes through each vertebra's centrum, to the logical stopping point. In pinnipeds and sea otters, this rod stops at the fused sacral vertebrae; in cetaceans and sirenians it continues until the vertebrae are sufficiently small to merit a change of support rod diameter. The rod may be up to half the diameter of the smallest vertebral centrum through which it will pass. If the spinal column is prepared whole, with vertebral disks left intact, a rod can be covered with polyethylene tubing and run down the neural canal.

Some workers prefer to use threaded stainless steel, with or without a polyethylene tubing covering. Others however, find stainless steel to be hard to bend and drill. Stainless steel can be easily cut with a Dremel cut-off wheel (Dremel Corporation www.dremel.com). Some workers prefer aluminum rods due to their lighter weight and ease of bending. Others however, prefer to not use aluminum because glues do not stick to it well. Some workers prefer galvanized, threaded rod.

A variety of adhesives can be used. Cyanoacrylate adhesives and hot glue are often used to tack bones in place prior to a final application of space-filling material between bones. Aesthetically speaking, a case can be made both for and against using bone-colored space-filling compounds. The skeleton will appear as a more homogeneous whole if bone-colored compounds are used; however, some prefer the more dramatic appearance of a darker material to offset the individual bones. For structurally strong, space-filling materials the authors have used Magic Sculpt (Tap Plastics, www.tapplastics.com) and marine epoxy (West Systems www.westsystems.com). Both materials can be rather expensive in large quantities, but may remain feasible within limited budgets for small to medium sized specimens. Magic Sculpt has the advantage of being the consistency of clay while hardening to a strong plastic overnight. It also smoothes with water while wet and is sandable and paintable after curing. Marine epoxy can be more difficult to work with but is stronger overall. Unthickened, it has the consistency of honey, but it can be thickened with microfibers to the consistency of peanut butter. Thickened, it is roughly bone colored and can be shaped after curing with an abrasive wheel in a Dremel tool. Other workers have used clear or white silicone sealant built up in layers with success.

Five-minute clear epoxy may be useful in some circumstances but is runny prior to setting and tends to yellow with time. Hot glue has been used for a variety of purposes, but is not very strong and appears to loosen its grip over time.

To simulate cartilage, such as the costal cartilages, many materials have been used. Silicone applied in neat layers over heavy gauge wire has worked well, as have various kinds of tubing. Rigid materials tend to not be very successful cartilage replacements. To be thorough when replacing cartilage, remember to consider the cartilaginous extensions of the digits in otariids.

Flipper bones may be pinned together with stainless steel or aluminum wire or small gauge threaded rod segments. Sixteen gauge wire is adequate for small flipper

bones or and for tail vertebrae. If the flipper bones are of sufficient size, 11-gauge aluminum chain-link fencing ties work well and allow easy bone positioning prior to the addition of space-filling material between the bones.

Scapulas are separated from the rib cage by thick, powerful muscles; hence, the limb may be spaced away from the ribs by short sections of clear, stout vinyl tubing or other material. The pectoral flippers are attached by pinning and epoxying the entire front limb together in the desired position, then wiring the scapula to the rib cage at judicious locations, with the tubing sections added as spacers. The pectoral limb may also be mounted to thin, bendable plastic sheet (e.g., polycarbonate), such that the limb arches over the rib cage without touching it. Plastic sheets easily crack, therefore, drill any holes carefully and do not overtighten attachments.

Missing bones may be replaced by a number of methods. If the missing piece is extant but not to be used in the display, a mold can be made of it and a replacement cast. Bondo, Plaster of Paris, Magic Sculpt, silicone rubber, and urethane have been used to make one and two part molds. Silicone rubber or urethane available from Tap Plastics is ideal but may be expensive for large pieces. Plaster of Paris is less expensive but may be more troublesome to work. Replacement material should not be subject to significant shrinkage during curing and should produce a good level of detail. TAP Quik-Cast Polyurethane Casting Resin System (Tap Plastics) works quite well with silicone and urethane molds. The casting material is watery when mixed, flows well into molds, and is ivory colored when cured. This method can also serve to make a plastic resin replacement from a hand sculpted clay or wax model (useful when the original is missing). Keep in mind the need for air bubbles to escape and the method by which you will remove the finished product from the mold. Silicone molds usually require tiny air vents cut at strategic locations. A sharpened three millimeter metal tube works well for removing a tiny core of the mold material at air-trapping locations. One-piece silicone or urethane molds must be judiciously slit to allow removal of first the original and later, the replacement. When slitting these molds, use an Exacto knife and cut in a zigzag fashion to allow for later reapposition of the mold. A straight cut makes for difficulty in proper alignment later.

Skulls are often stabilized and teeth attached by the careful application of white glue. Coat the skull in a 10% white glue mixture for a protective coating. The right and left mandibles may be attached at the symphysis with white glue, silicone or epoxy. The mandible may be attached to the skull by running a wire through the temporomandibular joint by drilling through the chondylar process of the mandible and the mandibular fossa of the temporal bone. Sixteen gauge wire curled into a tight coil at the ends allows the wire to be tightened into the desired position. Round needle-nosed pliers are needed for this. To mount the mandible in an open-mouth position, wire through the joint as above but leave enough play in the wire to allow bending to the desired position. Temporarily brace the mouth open with any material, such as crumpled paper. Tighten the wire as needed. Fix the jaw in position by adding space-filling epoxy to the TM joint. Remove the temporary mouth brace when the epoxy has cured.

A number of methods have been used to attach the skull to the spinal rod. For small skulls, metal pins (such as 3/16 inch threaded steel) may be mounted into the occipital condyles, through the atlas vertebra, and into the axis. The spinal rod, in this case, may stop at the axis or atlas vertebra. For larger skulls, the spinal rod may extend

into the skull cavity itself. The skull cavity will need to be filled with a rigid material such as plaster of Paris or hard-setting insulation spray foam. All perforations in the skull should be masked off before filling the cavity, and the occipital condyles should be masked as well, to prevent the material from adhering to the outside of the skull if it expands beyond the condyles. Once the cavity has been filled, a carefully positioned hole may be drilled to accommodate the rod. Alternatively, a metal support brace or cradle may be custom built to hold the skull in a less altering manner this however requires welding skills and access to equipment. This is a preferred method when working with large specimens.

1.3.2.3. Pinniped and Sea Otter Notes

Pinniped and sea otter preparation techniques are very similar. A realistic spinal posture can be obtained by arranging the vertebrae such that the faces of adjacent vertebrae centra are parallel to each other. The natural bend of the neck comes from the shape of the individual cervical vertebrae, rather than from the spaces between them. Phocids have an extremely deep "S" curve to their neck position. This means that when a seal's head is tucked in, the skull is quite close to the scapulas, nearly forming a "U" shape with the spine curving to the ventral area of the neck. This allows them the ability to quickly dart their head forward to catch prey. Otariids also have a significant curve to the resting neck posture. Pinnipeds and sea otters naturally assume dynamic poses when alive, and have extremely flexible spines. Skeletal articulations will better educate the viewer if they are designed with naturalistic fluid postures.

California sea lions have 15 ribs on each side. The costal cartilages do not merge. Numbers 1-9 attach to the spaces between the sternabrae. Numbers 10-12 reach the ziphoid but are not attached; their tips lie ventral to the ziphoid. Numbers 13-15 float completely and taper in length. On a large animal there may only be 3-4 cm between the end of rib number 15's cartilage and the patella, as the rib cage is quite large overall. Sea lions have a relatively small abdomen.

Vertebral spacing varies from one area of the spine to another. Use radiographs or take careful measurements to maintain accurate spacing, lest the finished skeleton be foreshortened. For example, the California sea lion has much larger spaces between the lumbar vertebrae immediately cranial to the pelvis than between the thoracic vertebrae; this allows the spine to bend to rotate the hips forward for quadrapedal walking. When they do this, the calcaneus (heels) remains very close to the spine, and the trapezoidal shape formed by the four points of the knees and heels pivots as a unit at the lumbar vertebrae.

1.3.2.4. Cetacean Notes

Cetaceans pose several problems. Because cetaceans need to maintain neutral buoyancy their bones are filled with oil, which tends to leak out slowly over time. Preparation by chemical maceration in potassium hydroxide helps remove much of the oil, but bones may still need long soaking in ammonia or other degreasers.

Because their bones are not weight bearing, they are composed almost entirely of cancellous bone, with very thin cortices. For this reason cetacean bones are quite

vulnerable to water intrusion if mounted outdoors, as is often desirable because of their large size. Water intrusion invariably causes rotting, and may increase skeletal weight dangerously. A blue whale skeleton at the Long Marine Lab in Santa Cruz, California was sealed with a shellac-based primer and painted with latex house paint. This system works reasonably well, but requires periodic maintenance. For a gray whale skeletal mount, now in the design phase, they are considering using a clear penetrating epoxy sealer (Smith and Co., www.smithandcompany.org), a layer of marine epoxy, and a linear polyurethane topcoat. However the sealer is expensive (U.S. $75/gl) and requires several coats; it also necessitates using respirators while it is being applied, as the fumes are noxious. This sealer is used at The Marine Mammal Center for cetacean bones displayed outdoors. It protects bones from the elements for several years.

Finally, small cetaceans like dolphins and porpoises have large numbers of almost-identical teeth. A great deal of time can be saved by pulling teeth first and then keeping them in order, for example in modeling clay.

PART 2: SPECIES IDENTIFICATION

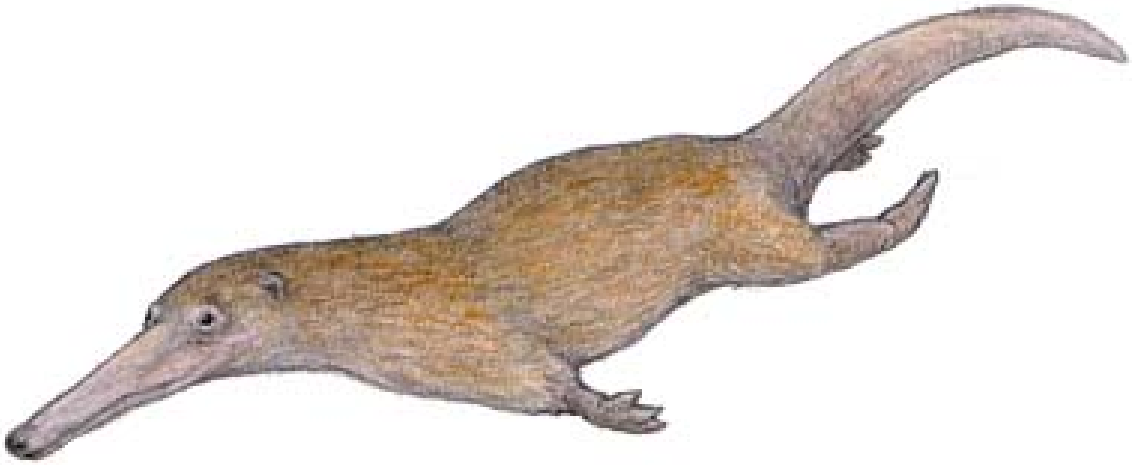
You will be tested on identifying marine mammals for their external characteristics. To that, please go to:
http://ip30.eti.uva.nl/BIS/marine_mammals.php?menuentry=zoeken&id=&selected=wetenschap

Below please find some additional species:

Order **CETACEA Brisson, 1762**
† Suborder **ARCHAEOCETI Flower 1883**
Family **PAKICETIDAE** Gingerich and Russell 1990
Pakicetus inachus Gingerich and Russell 1981

Family **AMBULOCETIDAE** Thewissen *et al*., 1996
Ambulocetus natans Thewissen, Hussain, and Arif 1994

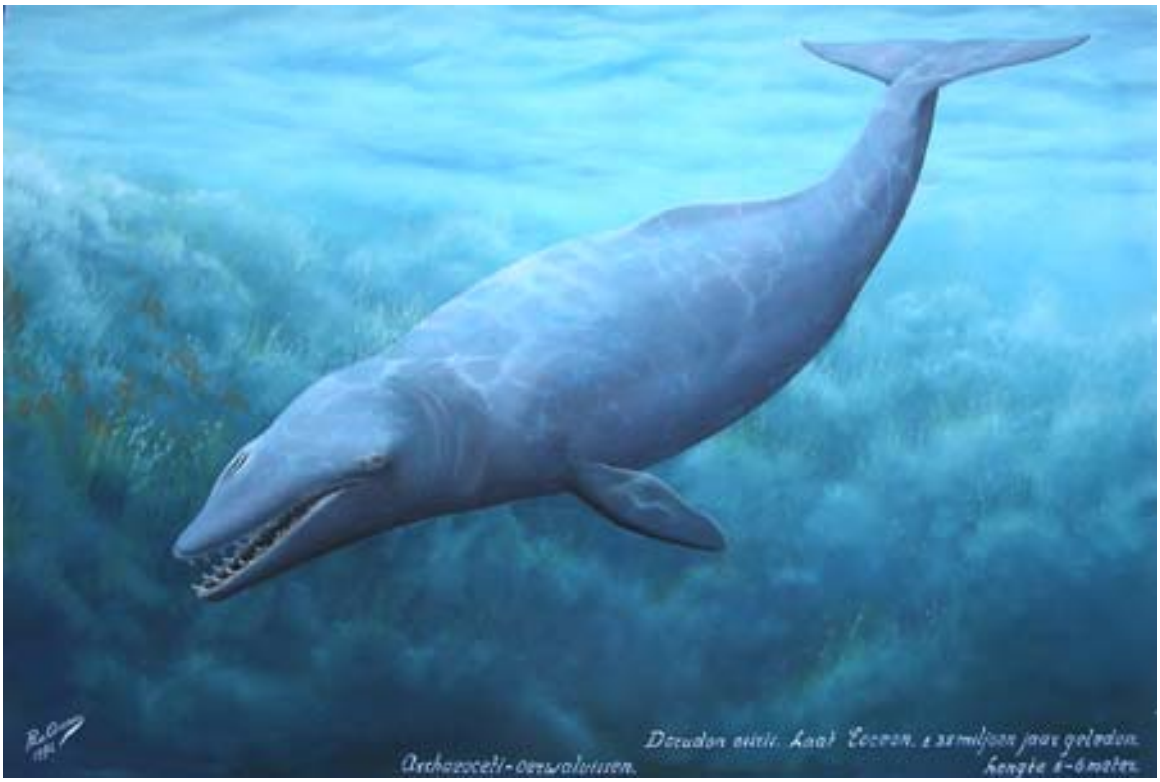
Family **REMINGTONOCETIDAE** Kumar and Sahni 1986
Remingtonocetus domandaensis Gingerich *et al*., 2001



Family **PROTOCETIDAE** Stromer 1908
Protocetus atavus Fraas 1904

Family **BASILOSAURIDAE** Cope 1868
Basilosaurus cetoides (Owen 1839)

Family **DORUDONTINAE** (Miller 1923)
Dorudon atrox (Andrews 1906)



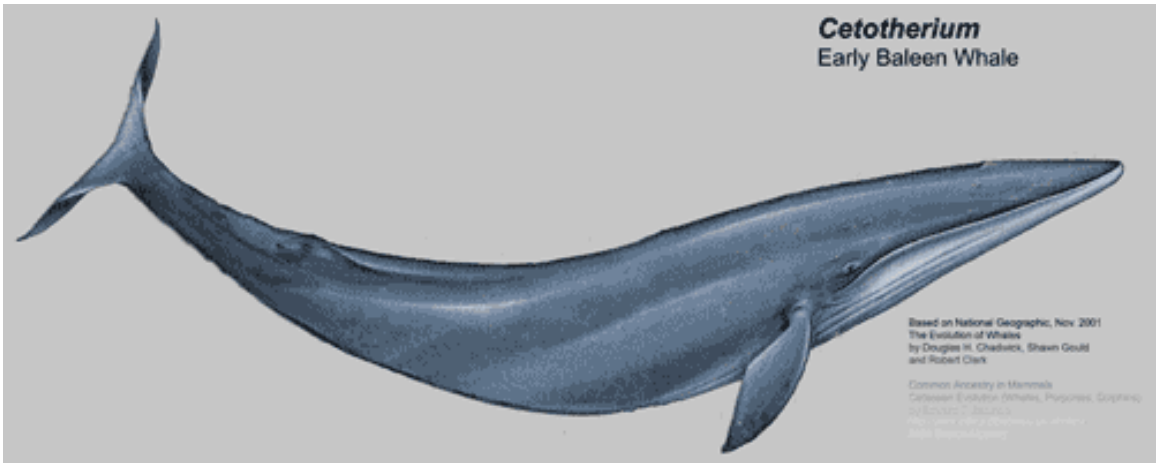
Suborder MYSTICETI Cope 1891

† Family **AETIOCETIDAE**

Mammalodon colliveri Pritchard, 1939 (Fordyce, 1982) Late Oligocene

† Family **CETOTHERIIDAE**

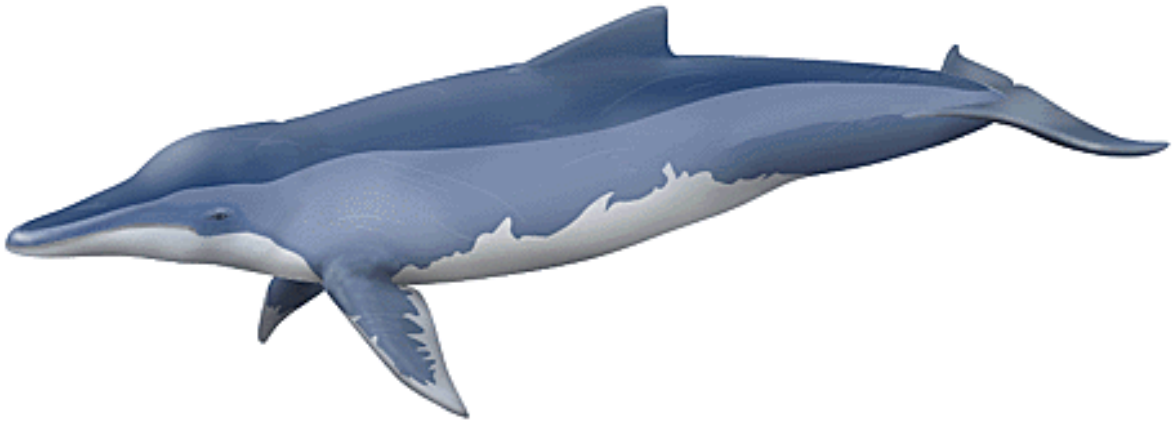
Cetotherium furlongi Kellogg, 1925 (Barnes, 1977) Early Miocene



Suborder ODONTOCETI Flower, 1864

† Family **SQUALODONTIDAE** Brandt, 1872

Squalodon Grateloup, 1840 (Whitmore & Sanders, 1977) Middle - Late Oligocene



Family **PHYSETERIDAE**

† *Brygmophyseter shigensis* (Hirota and Barnes, 1995) Middle Miocene

PART 3: Skulls and Skeletons: Homology, Analogy, and Taxonomy

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OBJECTIVES:

- Introduce students to the detailed study of marine mammal anatomy as represented by close measurement of skulls.
- Encourage students to discover patterns of similarity and difference in the anatomy of a range of marine mammals.
- Show how taxonomists can use these patterns of similarity and difference to elucidate relationships between different taxa. Which animals are more closely related and which less closely related based on skull?

LABORATORY OUTLINE:

FIRST PART: SKULLS

I. Use the provided diagrams and casts to identify the major bones of the skulls.

II. Measure and observe at least three animal skulls from among those provided. Record your observations on the form below.

III. In a class discussion, compare and contrast the anatomy of the skulls of cetaceans, pinnipeds, sirenians the sea otter, and the polar bear.

SECOND PART: SYNTHESIS

I. Review all of your observations plus all of the references and illustrations provided in the lab.

II. Consider how the animals' way of life and their skull characteristics may be related.

III. Consider how the surface features of the bones may give hints of the soft structures that surround them.

IV. Try to guess the taxonomic relationships of these various animals based on the measurements and observations of their skulls and forelimbs. Produce a branching diagram that reflects your assessment of the animals' relatedness.

Check the glossary for this lesson at the bottom of this handout

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QUESTIONS:

Comparative Study of Skulls

For each of the provided skulls, make the requested measurements and answer the questions:

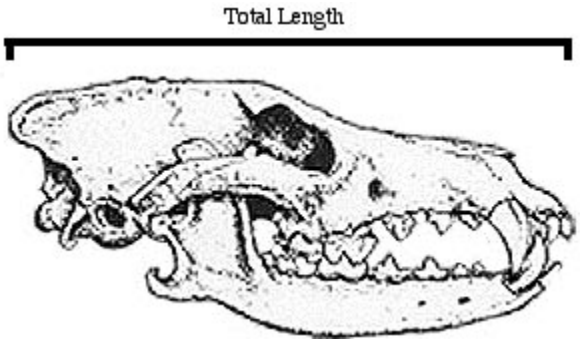
x/1 means divide the answer you get by the skull's total length (measurement #1).

#	MEASUREMENT (click to view an illustration of each measurement)	VALUE
1.	**Total length (rostrum to occipit).** This is the distance from the very front of the skull where the upper teeth join the skull, to the rim of the hole in the back of the skull (the occipit)	
2.	**Width of the orbit; x/1** This is the average diameter of the eye hole. Take several measurements and average.	
3.	**Rostrum to orbit; x/1** This is the distance from the rostrum to the closest point on the orbit of the eye.	
4.	**Orbit occipit length; x/1** This is the distance from the occipit to the closest point on the orbit of the eye.	
5.	**The ratio between rostrum to orbit length and orbit to occipit length; 3/4** The ratio between the distance from the anterior end of the skull to the eye, and the posterior end of the skull to the eye.	
6.	**Interorbital distance; x/1** The shortest distance between the orbits.	
7.	**Angle of the facial plane, in degrees** This is the angle between a line parallel to the hard palate and a line from the nasal bone to the frontal suture.	

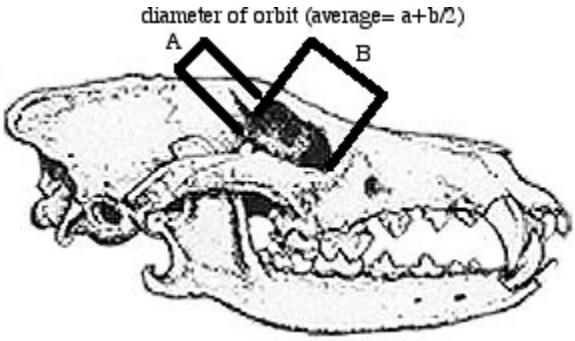
8.	**Cranial width; x/1** This is the width of the brain case at it's widest point.	
9.	**Height of sagittal crest; x/1** This is the greatest distance between the top of the sagittal crest and the cranium	
10.	**Mandible length; x/1** The distance between the articulation of the incisors at the midline and the jaw articulation.	
11.	**How many bones make up the mandible?**	
12.	**Length of the zygomatic arch; x/1**	
13.	**How many pairs of fenestrae (fossae) are there?**	
14.	A**verage diameter of the fenestrae; x/1** Average the widest and narrowest dimension.	
15.	**Is the orbit separated by bone from the fenestrae?**	
16.	**Approximately how many distinguishable separate bones make up this skull?**	

17.	**Identify and sketch the large flat bones of the upper side of the skull.** Pay special attention to which bones touch which other bones. Sketch:
18.	**What kind(s) of teeth does this animal have?** Are they scissorlike for slicing food? Daggerlike for stabbing? Flat and blunt for grinding food?
19.	**How do all of the above observations relate to a) the type of animal; b) the animal's way of life?**

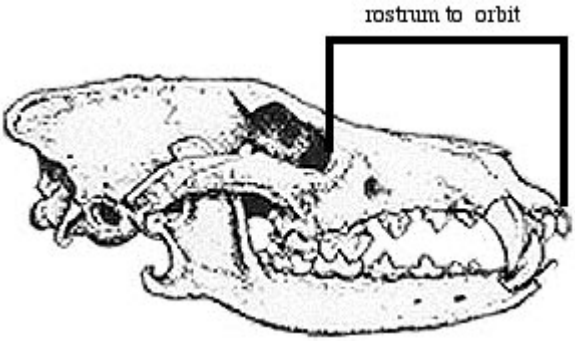
1.


Total Length

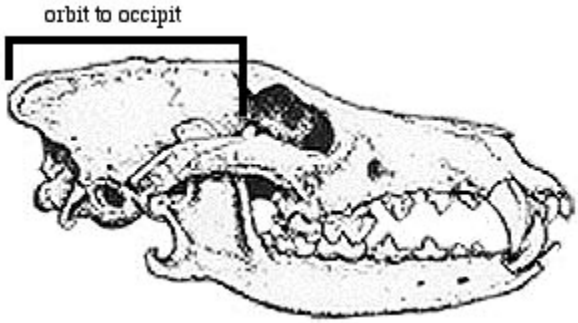
2.


diameter of orbit (average= a+b/2)
A
B

3.

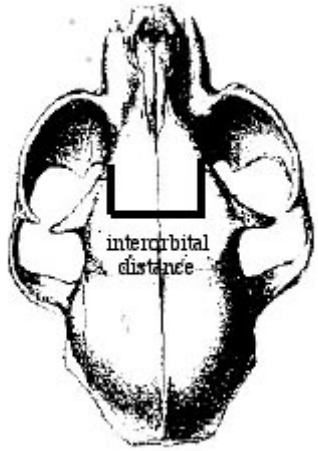

rostrum to orbit

4.

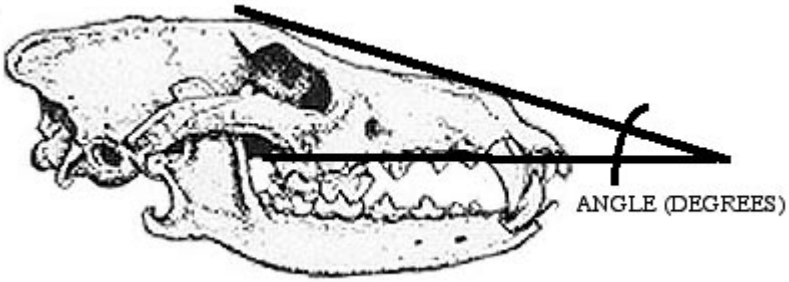


(5. n/a)

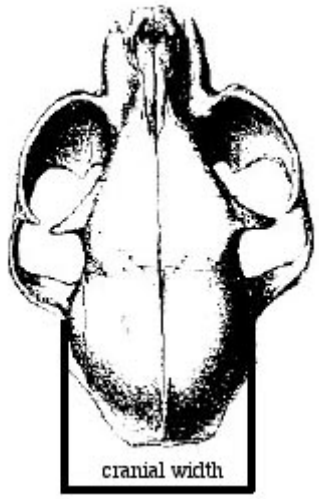
6.



7.

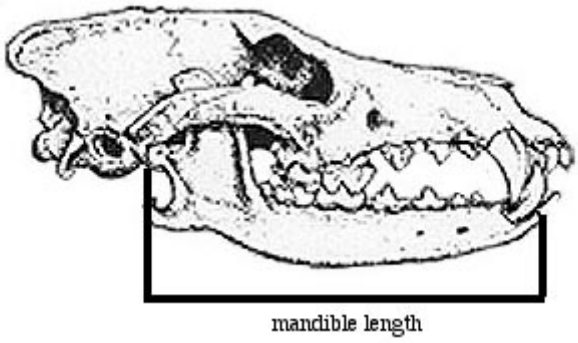


8.

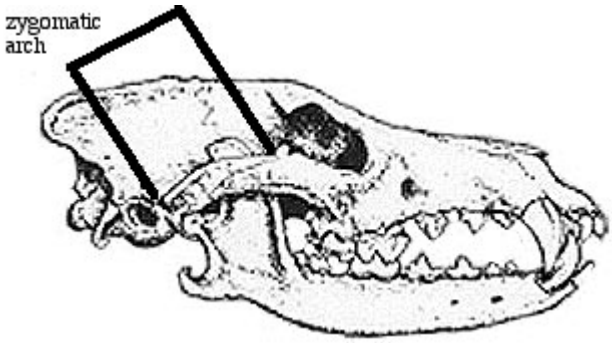


9.

10.



12.



GLOSSARY

Homologous structures:

Structures that have the same origin. They come from the same embryonic source tissue and are found in organisms who share the homologous structure because of descent from a common ancestor. Homologous structures do not necessarily have the same function in different organisms.

Example: Even though seemingly different in anatomy and used for different purposes, a bird's wing and a human's arm are both derived from the same ancestral structure and the same embryonic source tissue.

Primitive Structure:

A structure which is NOT exclusively shared only by all members of a group by virtue of descent from a common ancestor. A primitive structure cannot be used to distinguish between members of a group because all actually or ancestrally possess the structure.

Example: Vertebrae are possessed by all birds but also by all other vertebrates. Possession of vertebrae cannot be used to distinguish the birds from other vertebrates since all vertebrates including birds share that trait.

Shared, Derived Structure:

A structure which IS exclusively shared ONLY by all members of a group by virtue of descent from a common ancestor. A derived structure CAN be used to distinguish members of a group from nonmembers of the group. All members actually or ancestrally possess the structure.

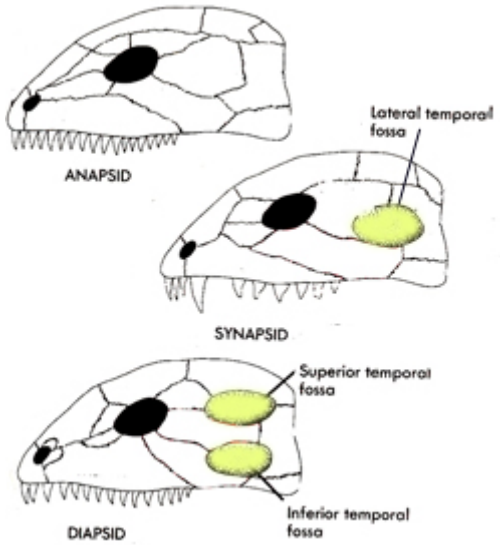
Example: Among vertebrates, feathers are possessed by all birds and by NO OTHER VERTEBRATE. Possession of feathers CAN be used to distinguish the birds from other vertebrates since no vertebrates except birds share that trait.

Types of Vertebrate Skulls:

Anapsids

Vertebrates that possess skulls with no major fenestrae.

Example: Turtles are anapsids



Diapsids

Vertebrates that possess skulls with two major fenestrae.

Example: Snakes, lizards, crocodilians, birds, and dinosaurs are diapsids

Synapsids

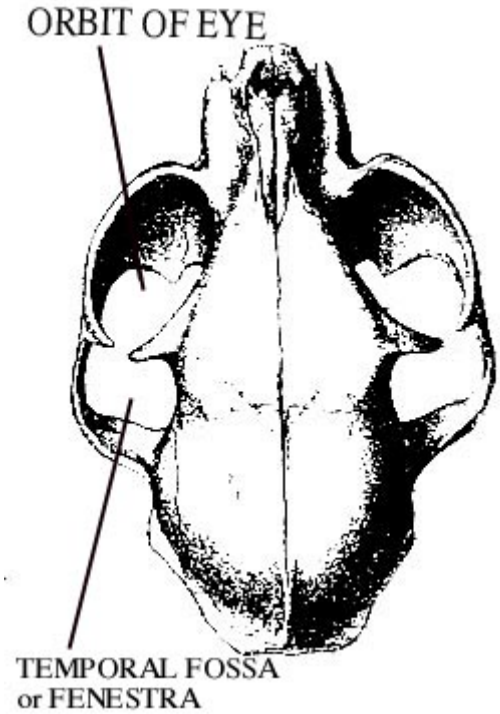
Vertebrates that possess skulls with one major fenestra in the region of the temporal bone.

Example: Mammals are synapsids

Skull Surface Features:

1. Fossae and fenestrae: Fossa means hole. Fenestra means window. Fossae and fenestrae are holes and windows in the skull.

An example is the temporal fossa/fenestra in synapsid skulls, pictured at right. In cats, unlike many animals including humans there is no complete bony separation between the orbit of the eye and the temporal fossa. A cat, along with all other mammals, is a synapsid.

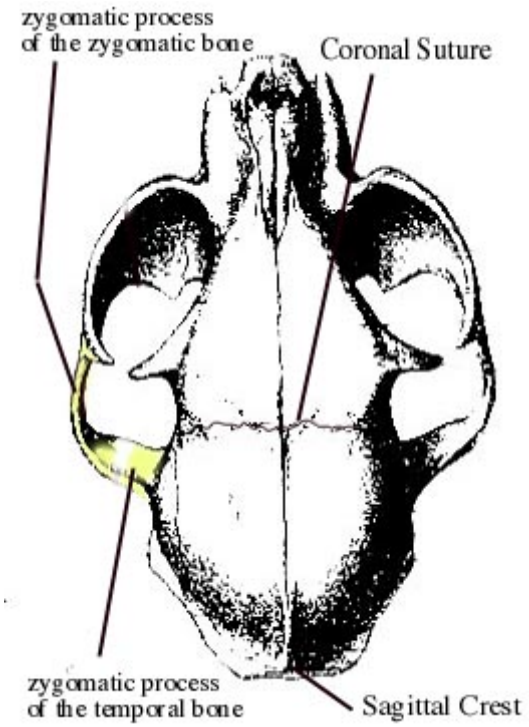


2. Zygomatic Arch: The zygomatic arch is an arch of bone composed of the zygomatic process of the temporal bone and the zygomatic process of the zygomatic bone. It defines the temporal fossa.

3. Process: A process is an extension of a bone.

4. Suture: A suture is an inflexible joint between two flat bones. It appears as a thin line on the surface of the skull.

5. Sagittal Crest: A ridge of bone on the posterior dorsal midline of the skull of many mammals. It is the origin (attachment point) of many muscles responsible for closing the jaw.

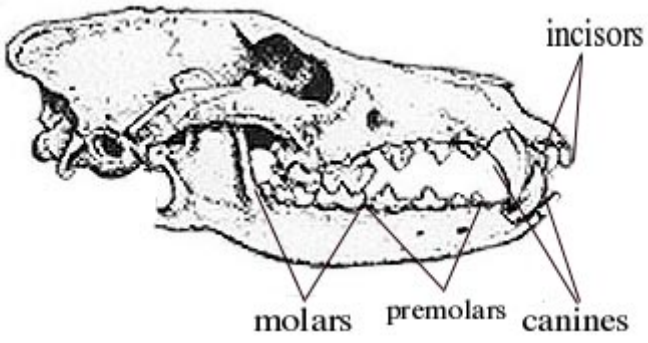


6. Teeth: A) Incisors: The anterior-most teeth, used for shearing and nibbling.

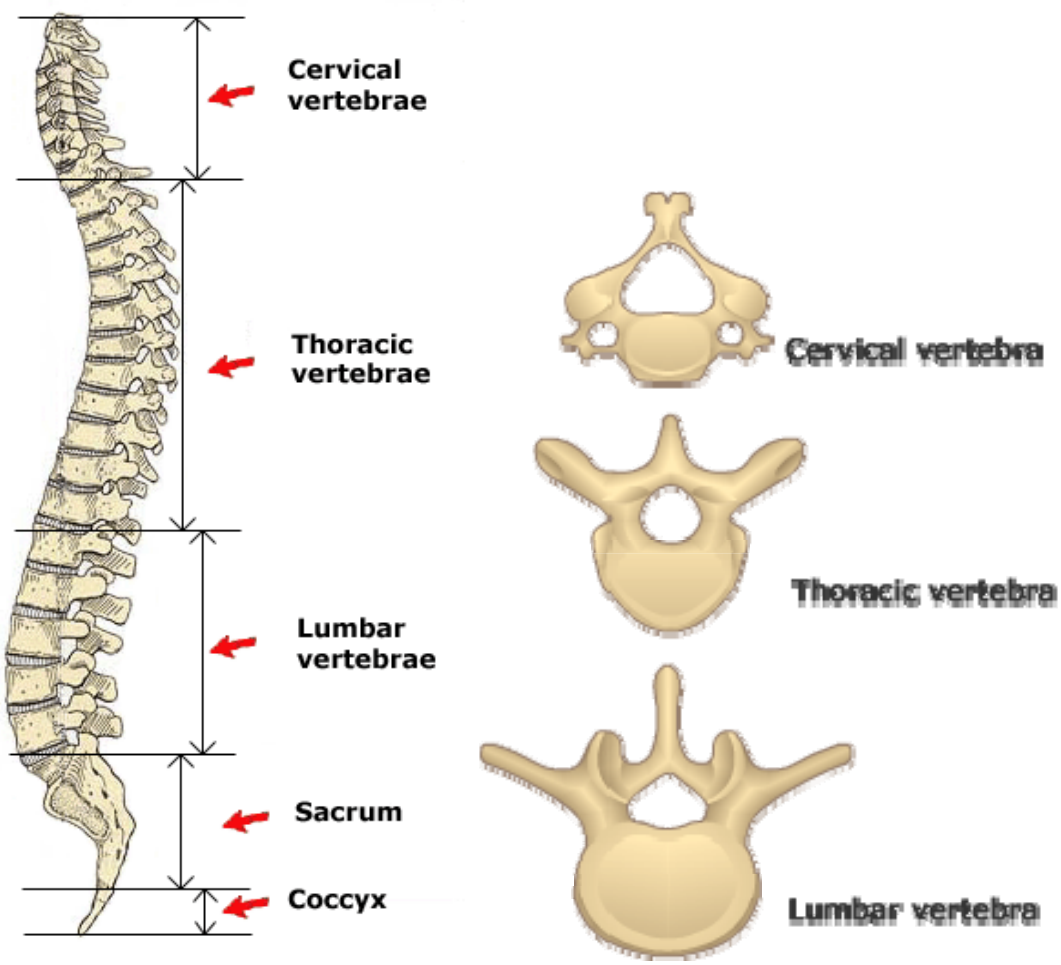
B) Canines: Daggar-like teeth, used for stabbing and holding.

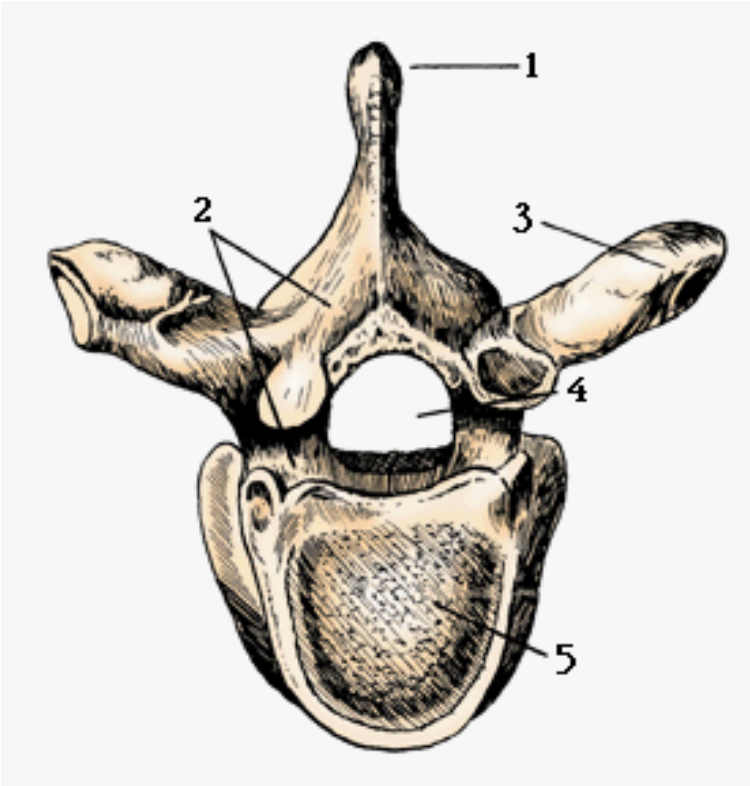
C) Premolars and molars: The posterior-most teeth, used for grinding.

D) carnassials: Specialized scissor-like premolars and molars. Found in meat eaters and used to shear and slice meat.



7. Vertebra


Cervical vertebrae
Thoracic vertebrae
Lumbar vertebrae
Sacrum
Coccyx
Cervical vertebra
Thoracic vertebra
Lumbar vertebra



Example of a thoracic vertebra:

1. Neural spine
2. Neural arch
3. Transverse process
4. Vertebral foramen
5. Centrum (body)

LIST OF LAB STUDY MATERIAL

CETACEA

1. Family: Ambulocetidae. *Ambulocetus natans*, walking whale (skull)
2. Family: Balaenopteridae. *Megaptera novaeangliae*, humpback whale (baleens)
3. Family: Balaenopteridae. *Megaptera novaeangliae*, humpback whale (vertebra)
4. Family: Balaenopteridae. *Balaenoptera acutorostrata*, northern minke whale (skull)
5. Family: Physeteridae. *Brygmophyseter shigensis*, biting sperm whale (fossil tooth)
6. Family: Physeteridae. *Physeter macrocephalus*, sperm whale (tooth)
7. Family: Physeteridae. *Physeter macrocephalus*, sperm whale (ambergris)
8. Family: Physeteridae. *Kogia breviceps*, pygmy sperm whale (skeleton)
9. Family: Physeteridae. *Kogia breviceps*, pygmy sperm whale (skull)
10. Family: Physeteridae. *Kogia sima*, dwarf sperm whale (skull)
11. Family: Ziphiidae. *Ziphius cavirostris*, goosebeak (Cuvier's beaked) whale (skull, teeth)
12. Family: Ziphiidae. *Mesoplodon carlhubbsi*, Hubbs' beaked whale (skull, teeth)
13. Family: Platanistidae. *Platanista gangetica minor*, Indus River dolphin (skull)
14. Family: Platanistidae. *Platanista gangetica gangetica*, Ganges River dolphin (skull)
15. Family: Iniidae. *Inia geoffrensis*, Amazon River dolphin (skull)
16. Family: Pontoporiidae. *Pontoporia blainvillei*, La Plata dolphin, Franciscana (skull)
17. Family: Monodontidae. *Delphinapterus leucas*, beluga (skeleton)
18. Family: Monodontidae. *Delphinapterus leucas*, beluga (skull and ossicles)
19. Family: Monodontidae. *Monodon monoceros*, narwhal (skull and tusk)
20. Family: Delphinidae. *Steno bredanensis*, rough-toothed dolphin (skull)
21. Family: Delphinidae. *Sotalia fluviatilis*, tucuxi (skull)
22. Family: Delphinidae. *Tursiops truncatus*, bottlenose dolphin (skull)
23. Family: Delphinidae. *Stenella attenuata*, pantropical spotted dolphin (skull)
24. Family: Delphinidae. *Delphinus delphis*, shortbeaked common dolphin (skull)
25. Family: Delphinidae. *Delphinus capensis*, longbeaked common dolphin (skull)
26. Family: Delphinidae. *Lagenorhynchus obliquidens,* Pacific whitesided dolphin (skull)
27. Family: Delphinidae. *Lissodelphis borealis*, northern right-whale dolphin (skull)
28. Family: Delphinidae. *Grampus griseus*, grampus, Risso's dolphin (skull)
29. Family: Delphinidae. *Peponocephala electra*, melonheaded whale (skull and ossicles)
30. Family: Delphinidae. *Pseudorca crassidens*, false killer whale (skull and ossiciles)
31. Family: Delphinidae. *Orcinus orca*, orca, killer whale (skull)
32. Family: Delphinidae. *Globicephala macrorhynchus*, shortfinned pilot whale (skull)

33. Family: Phocoenidae. *Phocoena phocoena*, harbor porpoise (skull)
34. Family: Phocoenidae. *Phocoena phocoena*, harbor porpoise (skeleton)
35. Family: Phocoenidae. *Phocoenoides dalli*, Dall's porpoise (skull)

PINNIPEDIA

36. Family: Otariidae. *Arctocephalus tropicalis*, subantarctic fur-seal (skull)
37. Family: Otariidae. *Callorhinus ursinus*, northern fur-seal (2 skulls, ♂, ♀, and baculum)
38. Family: Otariidae. *Zalophus californianus*, California sea-lion (2 skulls, ♂, ♀)
39. Family: Otariidae. *Eumetopias jubatus*, northern sea-lion (2 skulls, ♂, ♀, and baculum)
40. Family: Otariidae. *Phocarctos hookeri*, Auckland sea-lion (skull)
41. Family: Otariidae. *Otaria flavescens*, South American sea-lion (2 skulls, ♂, ♀)
42. Family: Odobenidae. *Odobenus rosmarus*, walrus (skull, deformed and healthy baculum)
43. Family: Phocidae. *Erignathus barbatus*, bearded seal (skull)
44. Family: Phocidae. *Phoca vitulina*, harbor seal (skull)
45. Family: Phocidae. *Phoca largha*, spotted seal (skull)
46. Family: Phocidae. *Pusa hispida*, ringed deal (skull)
47. Family: Phocidae. *Pusa sibirica*, Baikal seal (skull)
48. Family: Phocidae. *Halichoerus grypus*, gray seal (skull)
49. Family: Phocidae. *Histriophoca fasciata*, ribbon seal (skull)
50. Family: Phocidae. *Pagophilus groenlandicus*, harp seal (skull)
51. Family: Phocidae. *Cystophora cristata*, hooded seal (skull)
52. Family: Phocidae. *Monachus tropicalis*, Caribbean monk seal (skull)
53. Family: Phocidae. *Monachus monachus*, Mediterranean monk seal (skull)
54. Family: Phocidae. *Monachus schauinslandi*, Hawaiian monk seal (skull)
55. Family: Phocidae. *Mirounga angustirostris*, northern elephant seal (2 skulls, ♂, ♀, and baculum)
56. Family: Phocidae. *Mirounga angustirostris*, northern elephant seal (skeleton)
57. Family: Phocidae. *Leptonychotes weddellii*, Weddell seal (skull)
58. Family: Phocidae. *Lobodon carcinophaga*, crabeater seal (skull, ♂)
59. Family: Phocidae. *Hydrurga leptonyx*, leopard seal (skull)

SIRENIA

60. Family: Trichechidae. *Trichechus manatus*, Caribbean (West Indies) manatee (skeleton)
61. Family: Trichechidae. *Trichechus manatus*, Caribbean (West Indies) manatee (skull, scapula, humerus, hand, rib)
62. Family: Trichechidae. *Trichechus inunguis*, Amazon manatee (skull)
63. Family: Dugongidae. *Dugong dugon*, dugong (skull)
64. Family: Dugongidae. *Hydrodamalis gigas*, Steller's sea-cow (skull)

CARNIVORA

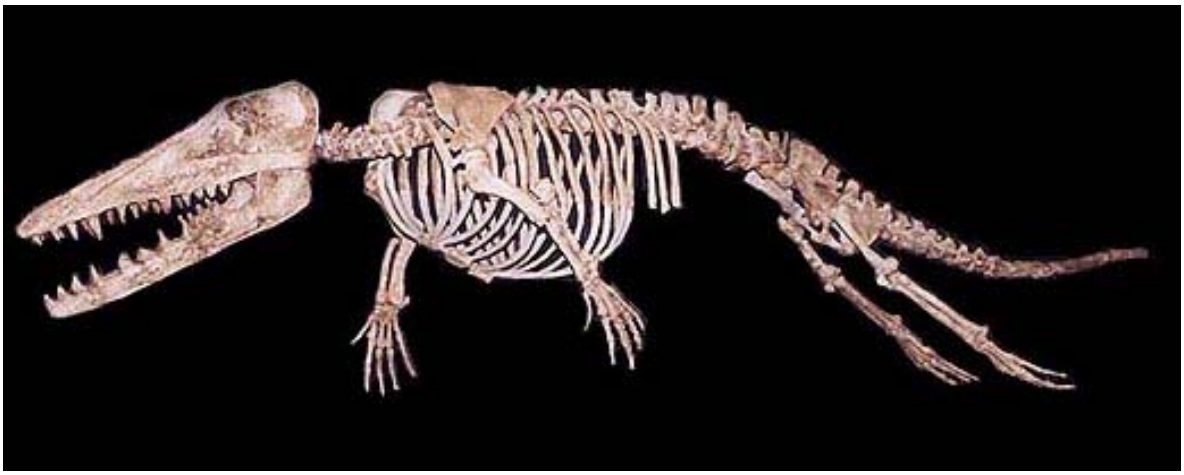
65. Family: Ursidae. *Ursus maritimus*, polar bear (skull, baculum, claw)
66. Family: Ursidae. *Ursus maritimus*, polar bear (hide)
67. Family: Mustelidae. *Enhydra lutris* (skeleton)
68. Family: Mustelidae. *Enhydra lutris*, sea otter (skull, baculum)
69. Family: Mustelidae. *Enhydra lutris* (pelt).

DESCRIPTION LAB STUDY MATERIAL

Below find the description of the material we provide you in the lab in the form of casts and other types of derives materials. Each description includes the species name, the English name, the type of material, family, size of the material and some additional remarks that are relevant for you to understand when examining that material. I have also included a picture of each one of them.

1. *Ambulocetus natans* **walking whale** skull AMBULOCETIDAE
Average Size: 3 m

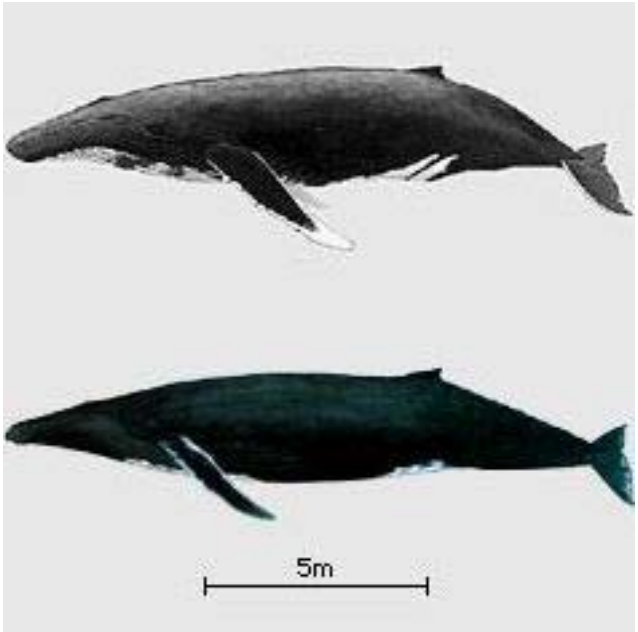
Ambulocetus's skull had a long muzzle, teeth that were very similar to later archaeocetes, a reduced zygomatic arch, and a tympanic bulla (which supports the eardrum) that was poorly attached to the skull. Although *Ambulocetus* apparently lacked a blowhole, the other skull features qualify *Ambulocetus* as a cetacean. The post-cranial features are clearly in transitional adaptation to the aquatic environment. Thus *Ambulocetus* is best described as an amphibious, sea-lion-sized fish-eater that was not yet totally disconnected from the terrestrial life of its ancestors.




research casting international

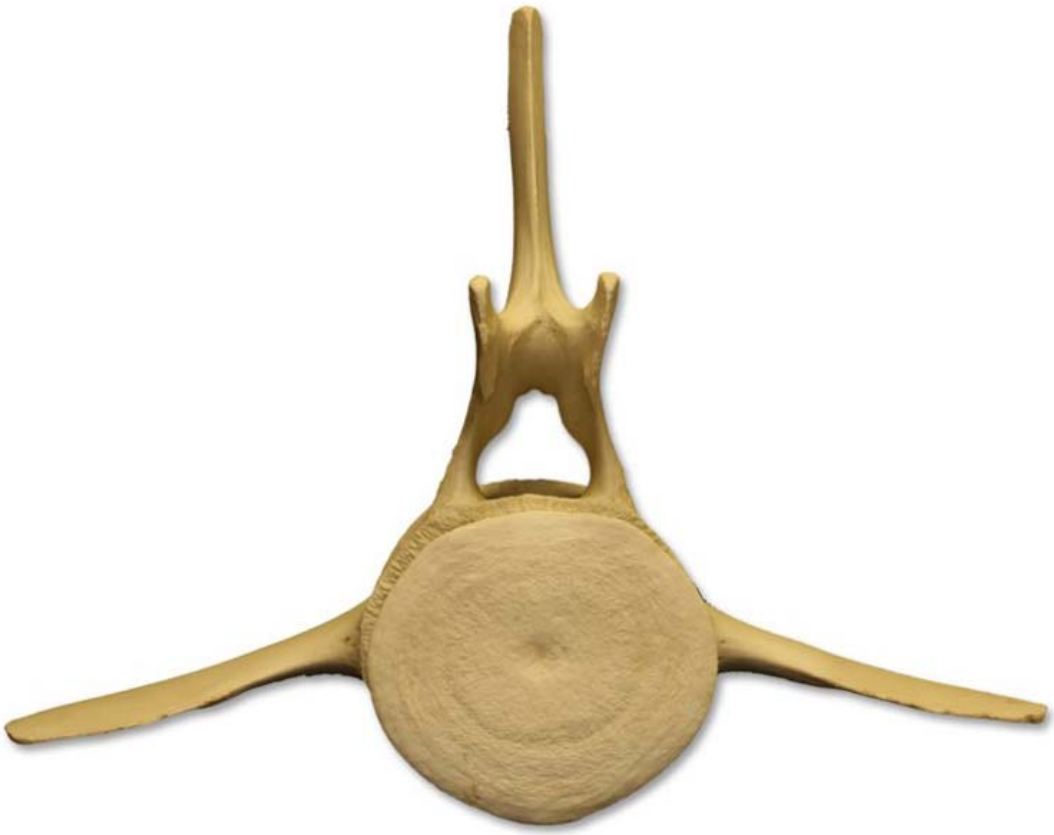
2. *Megaptera novaeangliae* **humpback whale** baleens BALAENOPTERIDAE

The humpback whale has about 330 pairs of dark gray baleen plates with coarse gray bristles hanging from the jaws. They are about 0.6 m (25 inches) long and 34 cm (13.5 inches).



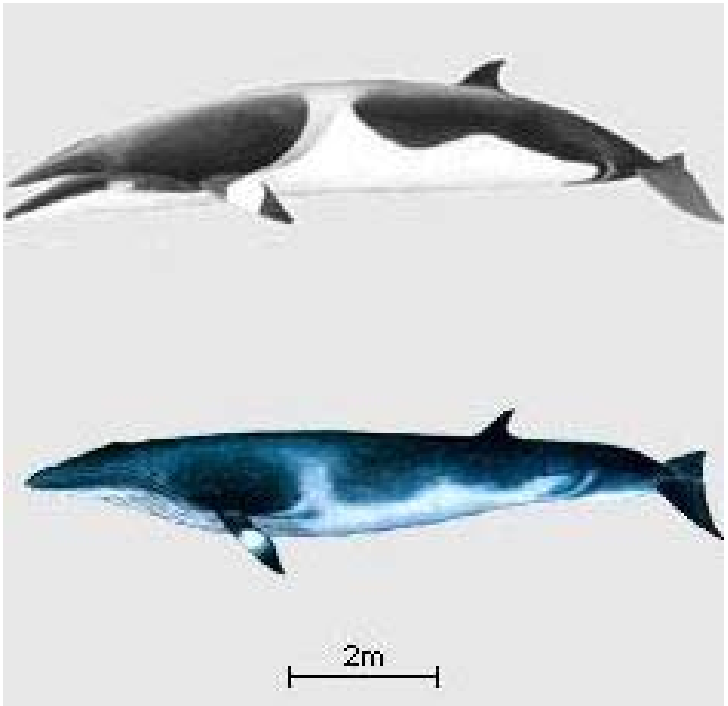
3. *Megaptera novaeangliae* **humpback whale** vertebra BALAENOPTERIDAE

This vertebra was cast from a 42-foot long female that washed ashore in Massachusetts. It measures 66 cm (26 inches) high and 84 cm (33 inches) wide. The individual epiphyseal disks are cast separately and removable. This specimen was cast to retain the natural weight and feel of the original specimen.



4. *Balaenoptera acutorostrata* **northern minke whale** skull BALAENOPTERIDAE
Average Skull Length: 152 cm

The minke whale, one of the smaller baleen whales, only reaches a length of 10.6m. Ranging throughout the cooler waters of the Atlantic, Pacific, Indian and Arctic oceans, the minke whale feeds on krill and small schooling fish.

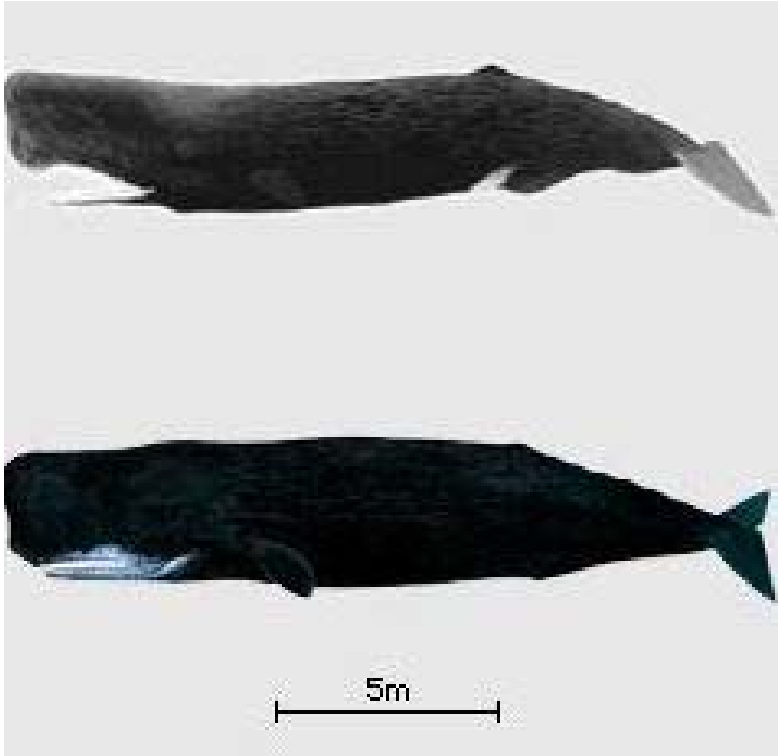


5. *Brygmophyseter shigensis* **biting sperm whale** fossil tooth
PHYSETERIDAE

Average Length: 29 cm 12-12/12-12

This is a cast of a tooth from a biting sperm whale from the Middle Miocene (about 15 MYA) from Japan. This animal reached 7 meters in length. Unlike the modern sperm whales they had teeth on both jaws which suggests they were predators of large marine animals.

6. *Physeter macrocephalus* **sperm whale** tooth PHYSETERIDAE
Average Length: 170 mm 0-0/16-30 X2 = 32-60

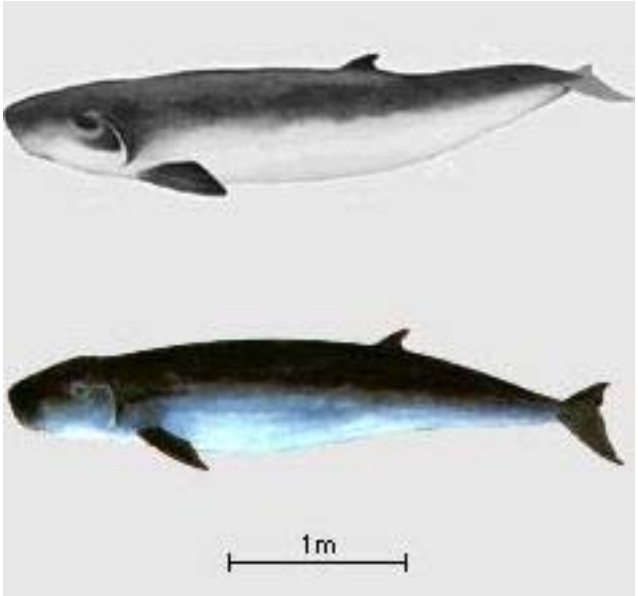


7. *Physeter macrocephalus* **ambergris** bottle PHYSETERIDAE

Ambergris is a waxy substance produced by in the sperm whales' stomachs and intestines, probably as the result of irritation caused by the beaks of cuttlefish (*Sepia medicinales*) these cetaceans consume. A single chunk of ambergris can weight more than 400 kilograms (ca. 882 pounds). Because it is less dense than seawater, ambergris is found either floating in the ocean or washed up on beaches after being excreted by the sperm whales. Ambergris has been used since antiquity for medicinal purposes by many cultures around the world. Ambergris most common use since the Middle Ages was as a key ingredient in the manufacturing of perfumes by the Arabs in order to slow down the evaporation rate of the perfume and add fragrance to it. Because of its rarity, natural ambergris has always commanded a high price. Today the ambergris that is used in perfumery is largely synthetic. Because of this, and the prohibition by the United States (through its *Marine Mammal Protection Act* of 1972) against trading any marine mammal product, natural ambergris has decreased in value and when sold elsewhere, the price fluctuates around US $20 per gram, depending upon its quality.

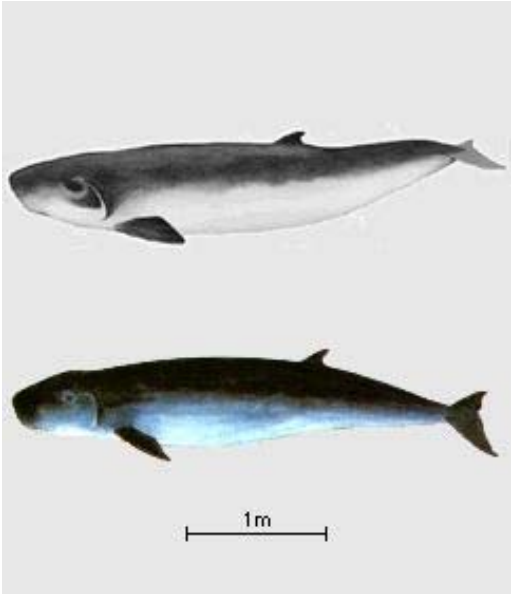
8. *Kogia breviceps* **pygmy sperm whale** skeleton PHYSETERIDAE
Average Body Length: 3.8 m

The pygmy sperm whale is found throughout much of the worlds tropical and temperate oceans. This species, as well as the sperm whale and the dwarf sperm whale, possesses a spermaceti organ from which it gets its name. This species feeds on squid crustaceans and small fish.



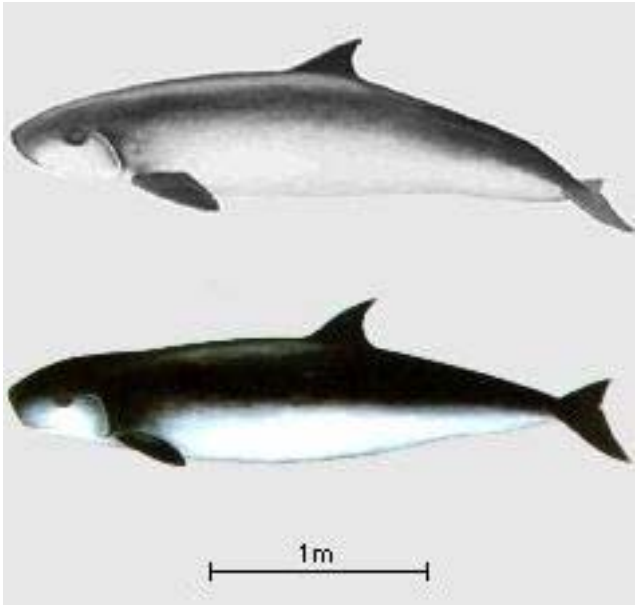
9. *Kogia breviceps* **pygmy sperm whale** skull PHYSETERIDAE
Average Skull Length: 44 cm 0-0/8-11 X2 = 16-22

The pygmy sperm whale feeds on squid, crustaceans, and small fish.



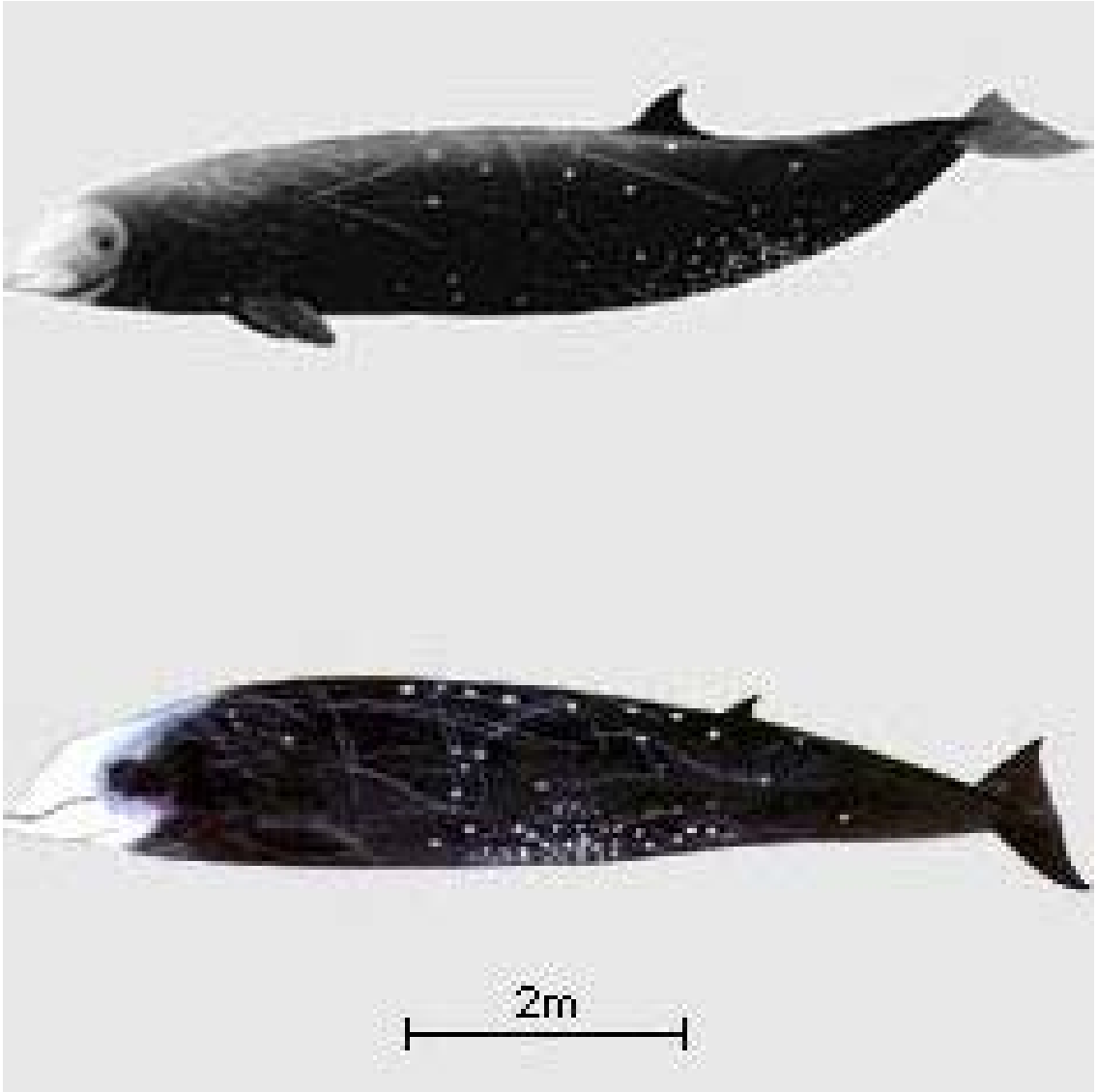
10. *Kogia sima* **dwarf sperm whale** skull PHYSETERIDAE
Average Skull Length: 26 cm 0-0/12-16 X2 = 24-32

This species feeds on squid, crustaceans, and small fish.



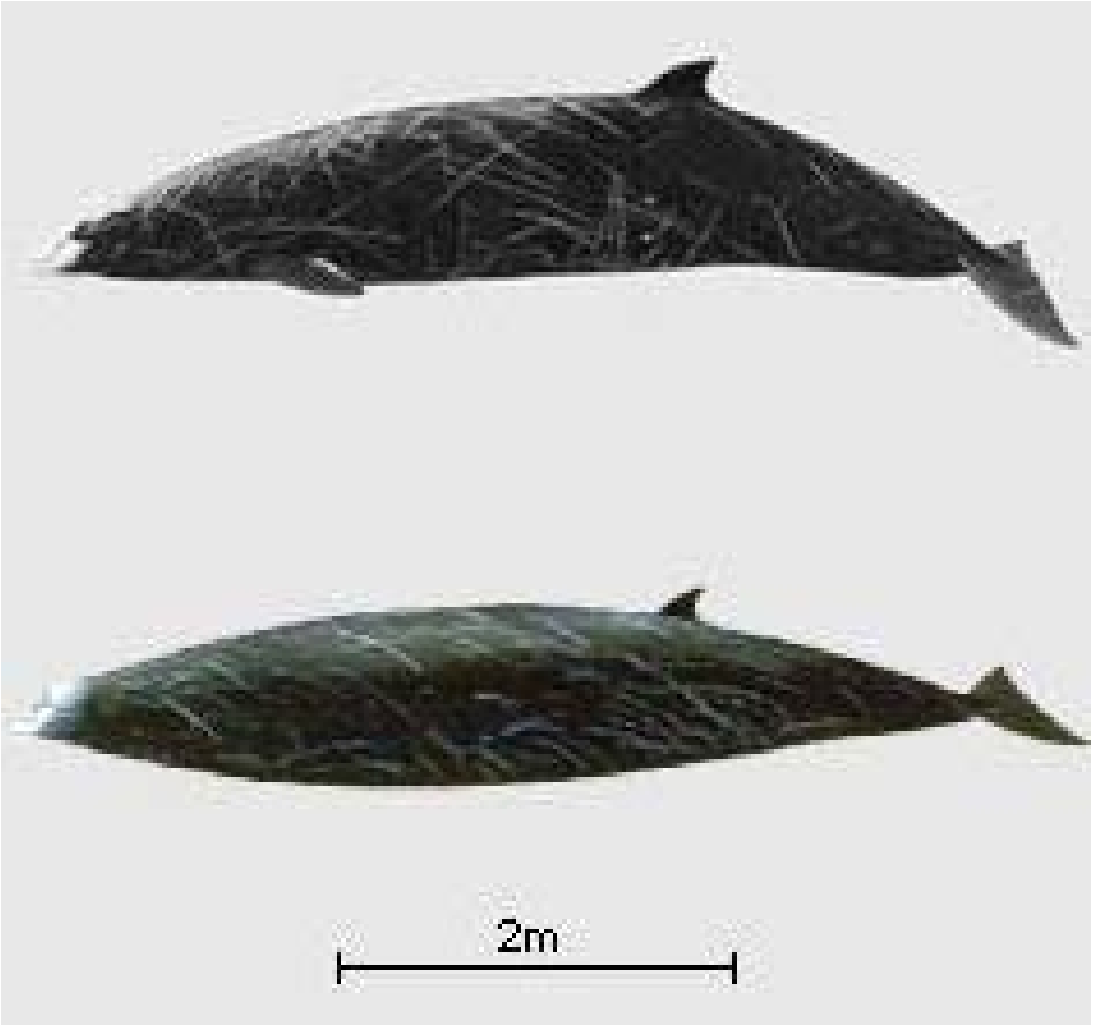
11. *Ziphius cavirostris* **goosebeak (Cuvier's beaked) whale** skull and teeth ZIPHIIDAE
Skull Length: 95 cm 0/1 X 2 = 2

They feed mostly on deep sea squid, but also take fish and some crustaceans.


2m

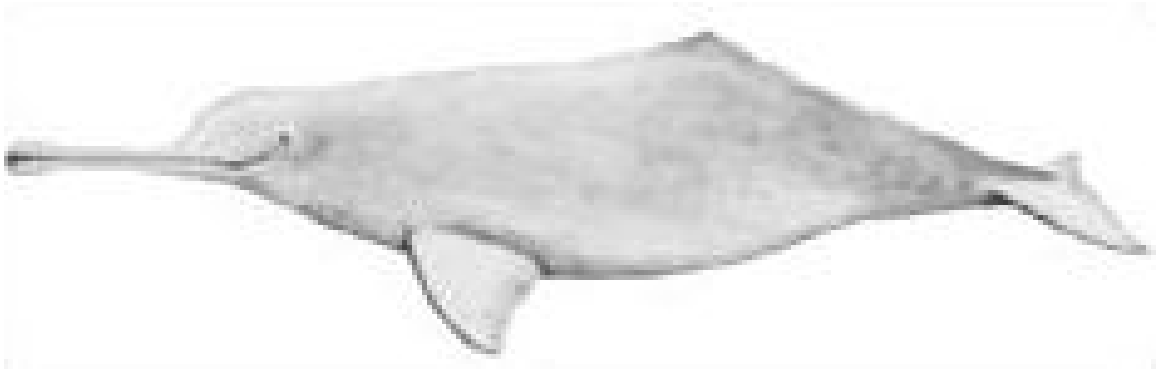
12. *Mesoplodon carlhubbsi* **Hubbs' beaked whale** skull and teeth ZIPHIIDAE
Average Skull Length: 79.5 cm 1-1 X2 = 2

All beaked whales have a long rostrum or "beak". Most beaked whales possess only two teeth that only erupt in mature males. Males use these teeth for fighting, which generally results in many scars, but rarely death.


2m

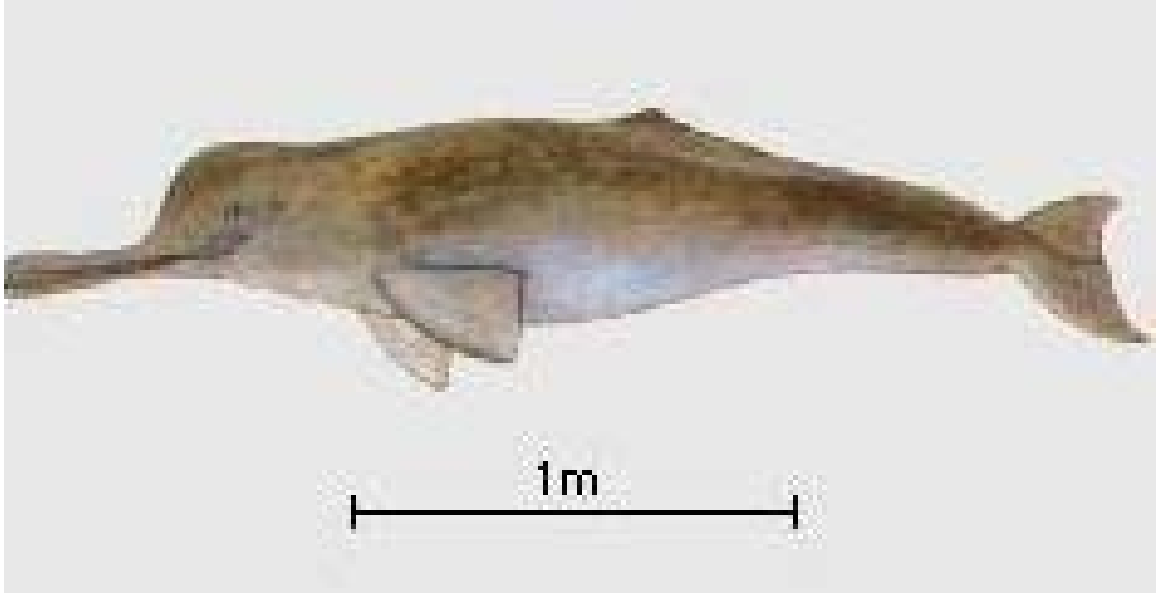
13. *Platanista gangetica minor* **Indus River dolphin** skull PLATANISTIDAE
Average Skull Length: 36.83 cm 26-29/26-29 X 2 = 104-156

They use their echolocation abilities combined with their highly toothed, long snouts to forage for many bottom-dwelling animals including fish and invertebrates.



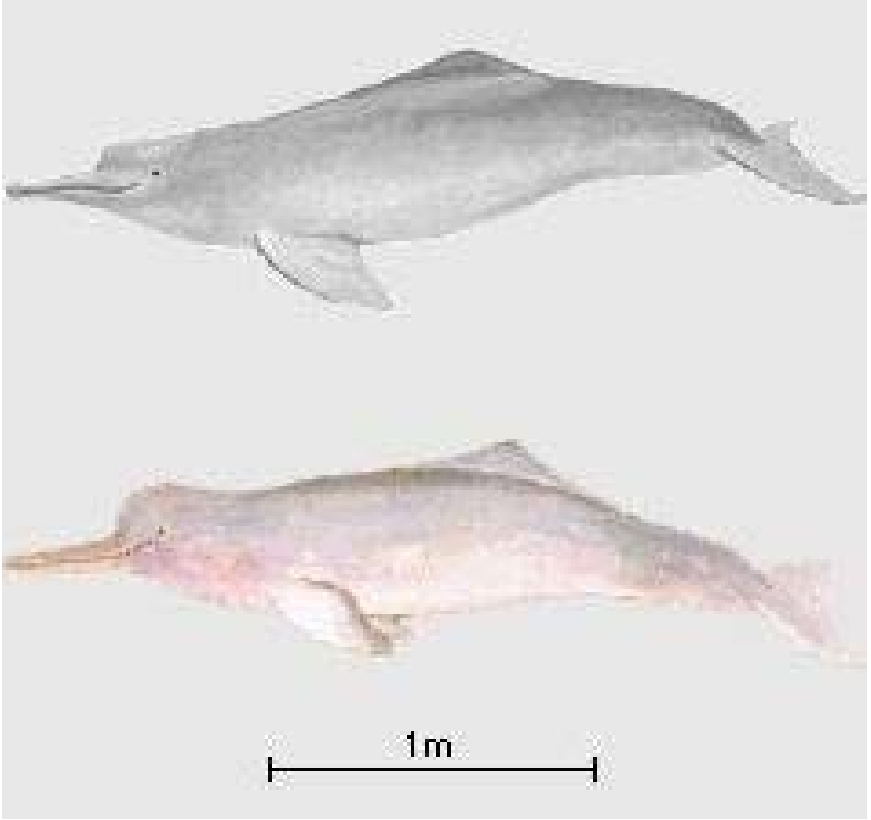
14. *Platanista gangetica gangetica* **Ganges River dolphin** skull PLATANISTIDAE
Average Skull Length: 44.45 cm 26-39/26-39 X2 = 104-156

They feed on several species of fishes, invertebrates, and possibly turtles and birds. They do much of their feeding at or near the bottom, echolocating and swimming on one side



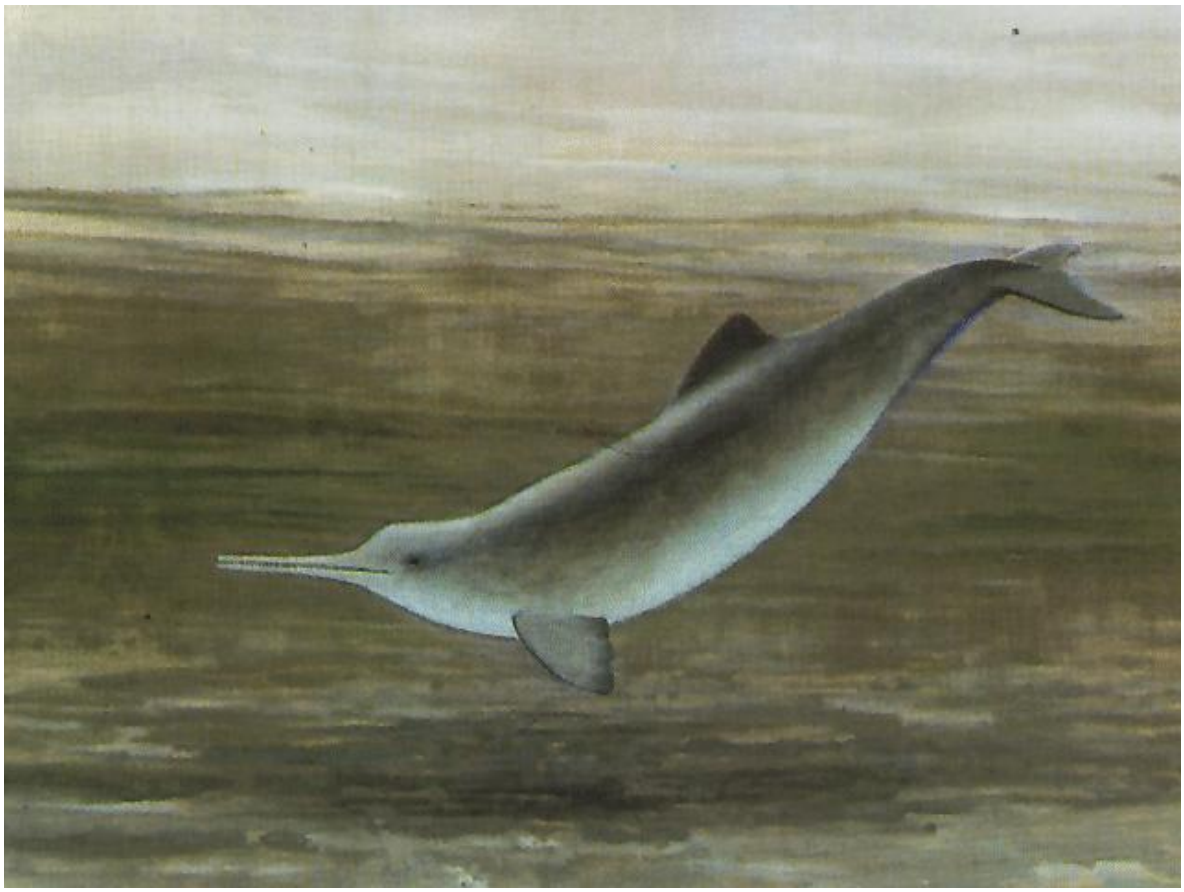
15. *Inia geoffrensis* **Amazon River Dolphin** skull INIIDAE
Average Skull Length: 48 cm 33-34/33-34 X2 = 132-136

The Amazon River dolphin is found throughout the Amazon and the Orinoco basins. Note that the dentition is typical of small fish-eater creatures like other freshwater dolphins and crocodiles, for example.

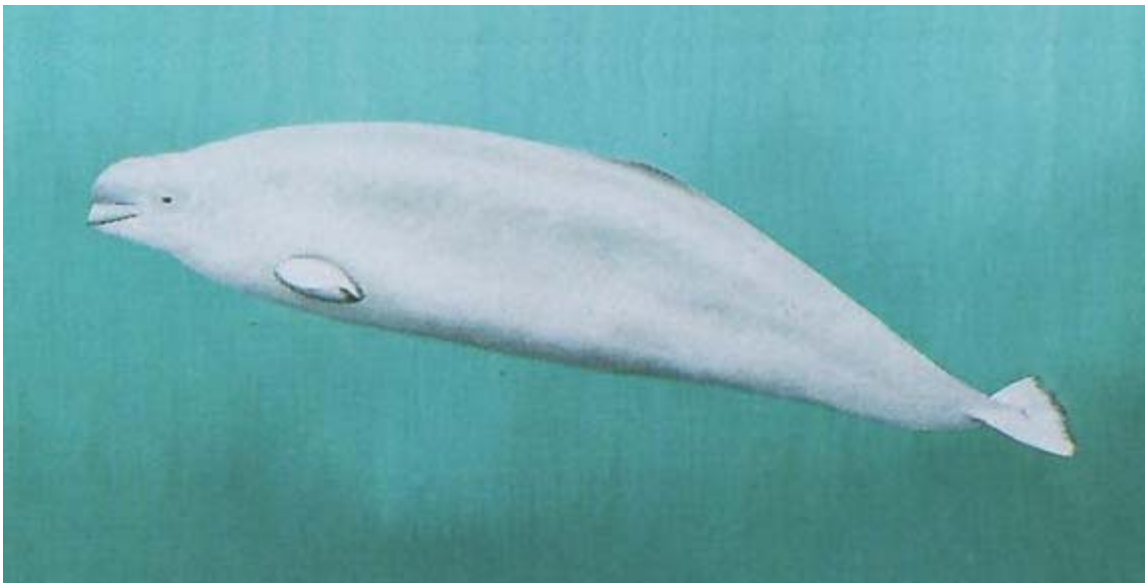


16. *Pontoporia blainvillei* **La Plata dolphin, Franciscana** skull PONTOPORIIDAE
Average Skull Length: 38.1 cm 48-62/48-61 X2 = 192-244

They feed on fish, squid, and shrimp.



17. *Delphinapterus leucas* **Beluga** skeleton MONODONTIDAE
Length: 3.2m

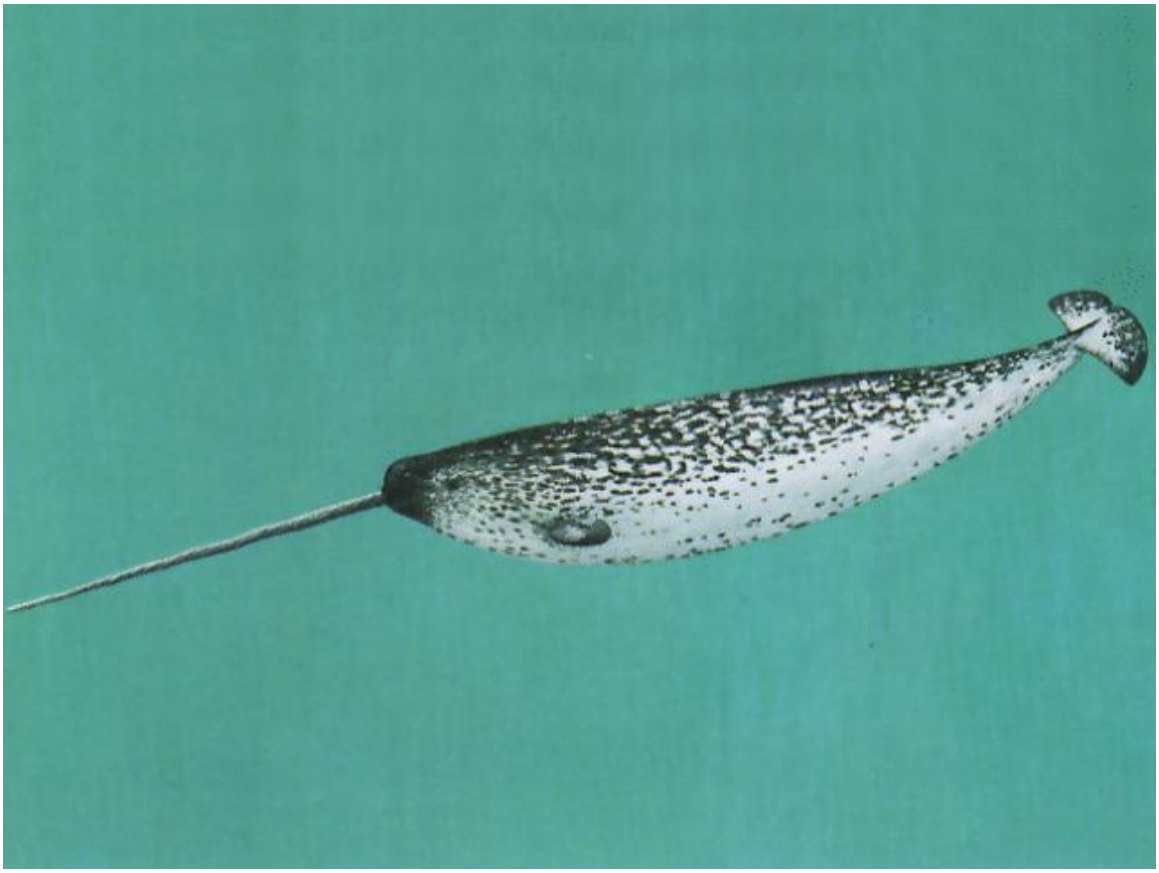


18. *Delphinapterus leucas* **Beluga** skull MONODONTIDAE
Average Skull Length: 50 cm 8-10/8-9 X2 = 34-38

They eat many fish species as well as invertebrates such as octopus, squid, crab, and snails. Feeding dives last from 3-5 minutes, although one individual was seen to remain underwater for 20 minutes and dove to a depth of 647 m. Due to the flexibility of the lips, a characteristic only shared by the Irrawaddy dolphin, it has been hypothesized that this species may use a "sucking" motion in order to draw prey into the mouth.

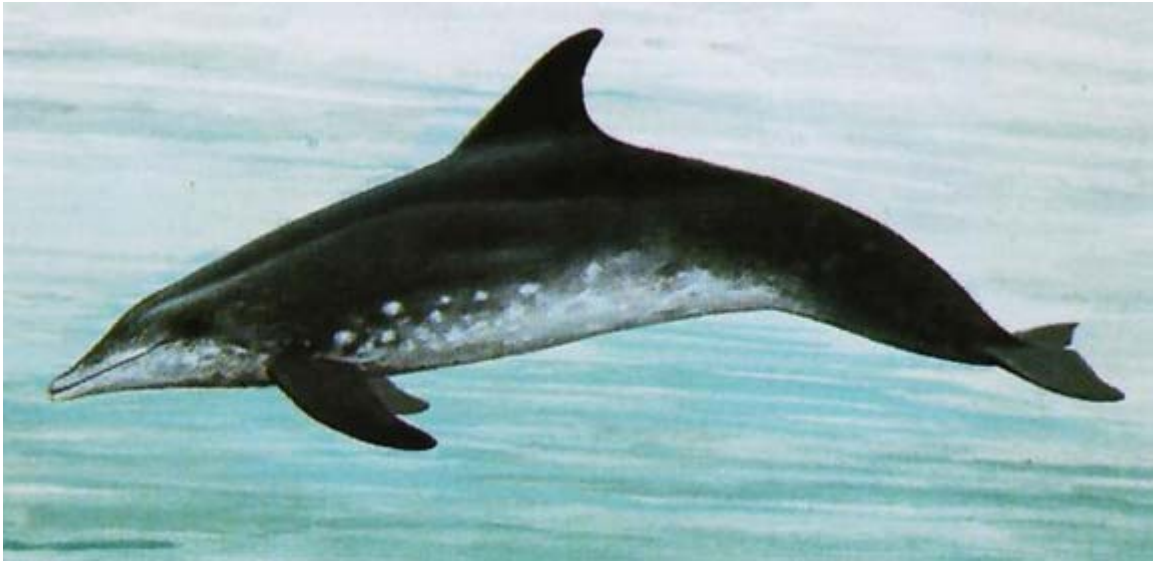
19. *Monodon monoceros* **Narwhal** skull and tusk MONODONTIDAE
Skull Length: 56 cm, Tusk Length: 155 cm 1-2/2 X 2 = 1-2

This species possess a long spiral tooth that projects like a horn and can reach a length of 3m in males. During the middle ages, the tusk of the narwhal was considered proof of the existence of the unicorn. This species lacks functional teeth for feeding but uses a suck and gulp technique to dine on shrimp, squid and fish.



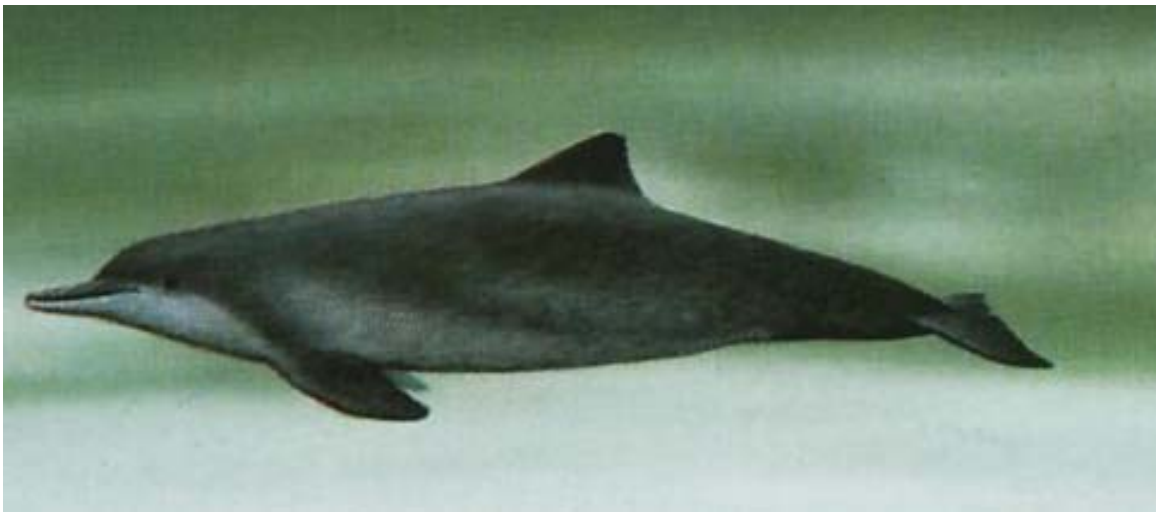
20. *Steno bredanensis* **rough-toothed dolphin** skull DELPHINIDAE
Average Skull Length: 51 cm 20-28/20-28 X2 = 80-112

The rough-toothed dolphin feeds on a variety of fish and squid utilizing its “rough” teeth.



21. *Sotalia fluviatilis* **tucuxi** skull DELPHINIDAE
Skull Length: 33 cm 26-36/26-36 X 2 = 104-144

They feed on fish and cephalopods.

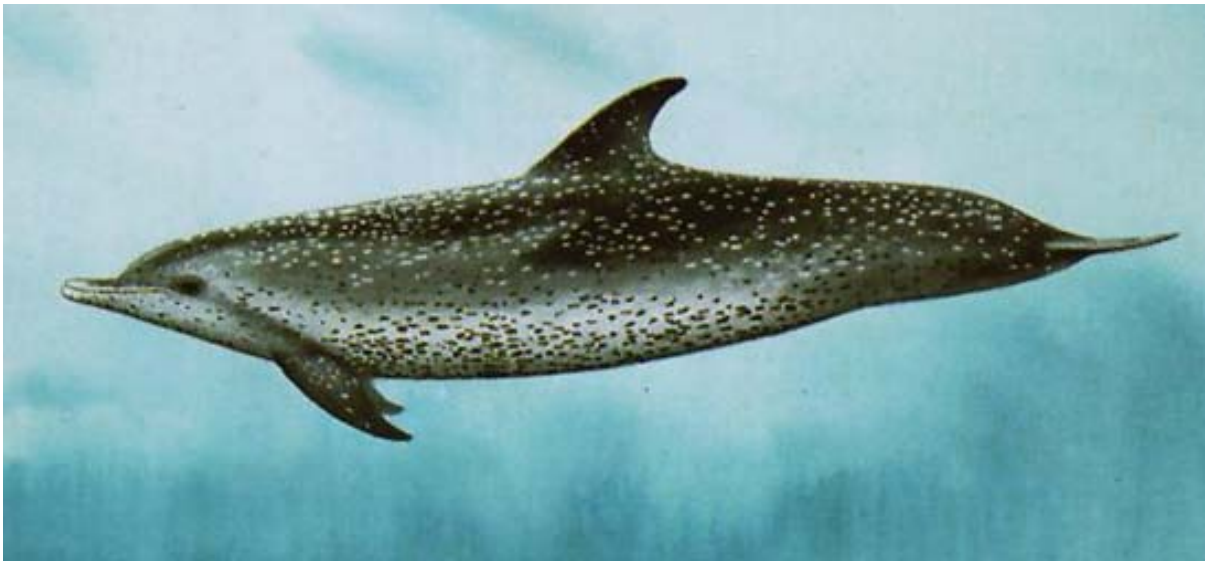


22. *Tursiops truncatus* **Bottle-nosed dolphin** skull DELPHINIDAE
Average Skull Length: 49 cm 20-28/20-28 X2 = 80-112

The bottle-nosed dolphin is probably the best known cetacean.

23. *Stenella attenuata* **pantropical spotted dolphin** skull DELPHINIDAE
Skull Length: 42 cm 35-48/34-47 X 2 = 138-190

They feed on fish, squids, and crustaceans.



24. *Delphinus delphis* **shortbeaked common dolphin** skull DELPHINDAE
Skull Length: 39 cm 41-54/41-54 X 2 = 164-216

They feed on fish and squids.

25. *Delphinus capensis* **longbeaked common dolphin** skull DELPHINIDAE
Average Skull Length: 47 cm 47-67/47-67 X2 = 188-268

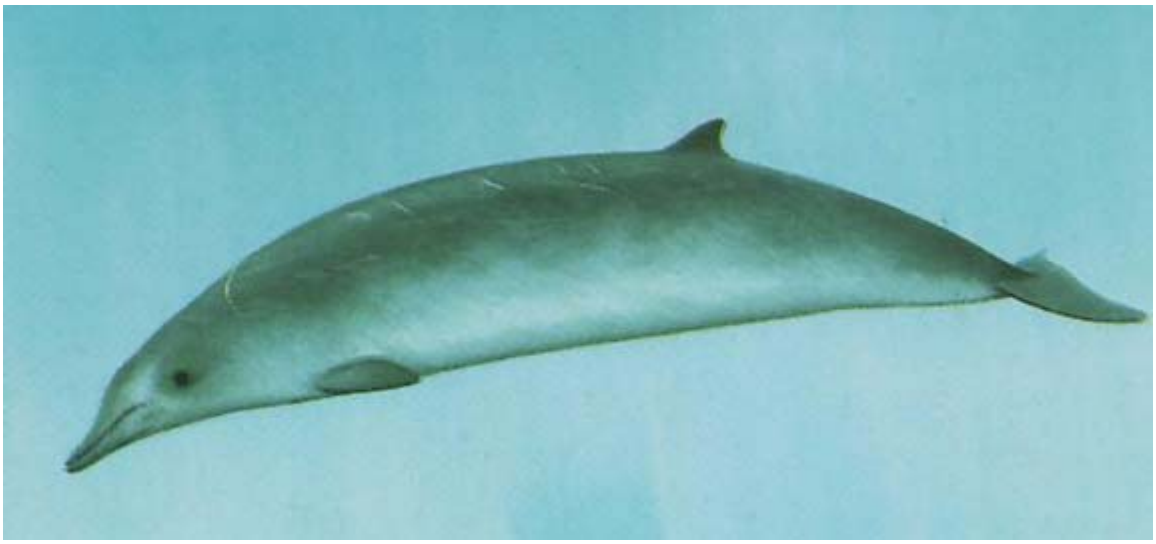
They feed on fish and squids.

26. *Lagenorhynchus obliquidens* **Pacific whitesided dolphin** skull DELPHINIDAE
Average Skull Length: 39 cm 23-36/23-36 X2 = 92-144

They feed on fish, squids, and crustaceans.

27. *Lissodelphis borealis* **northern right-whale dolphin** skull DELPHINIDAE
Skull Length: 45 cm 37-54/37-54 X 2 = 148-216

Right whale dolphins feed on squid and fish, particularly lanternfish.



28. *Grampus griseus* **grampus, Risso's dolphin** skull DELPHINIDAE
Average Skull Length: 56 cm 0/4-5 X2 = 8-10

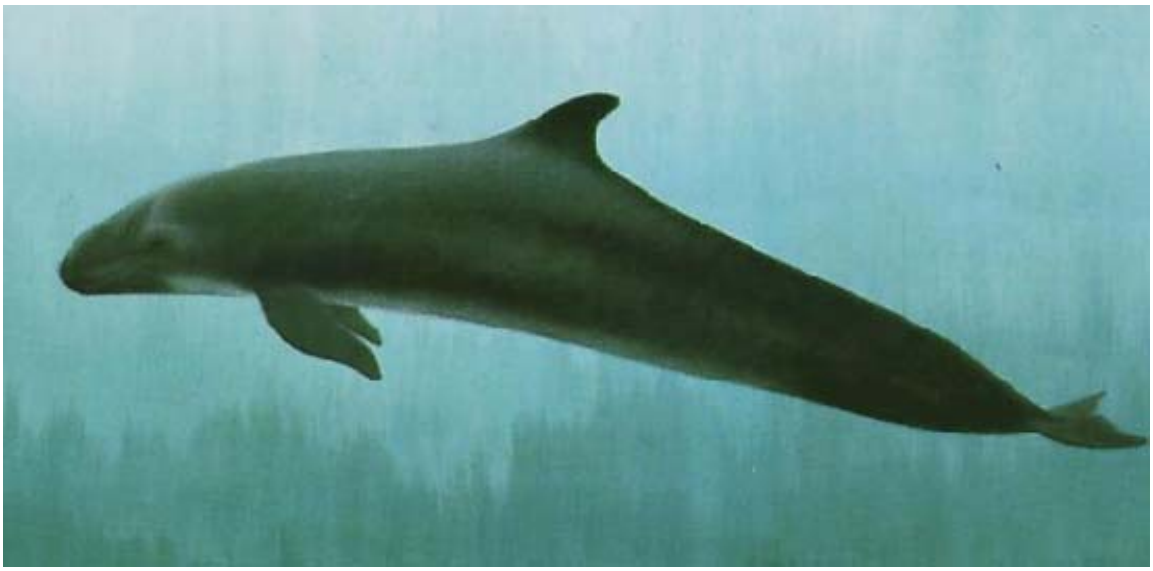
They feed primarily on squid and on a variety of fish.

29. *Peponocephala electra* **melonheaded whale** skull DELPHINIDAE
Average Skull Length: 46 cm 21-25/21-25 X2 = 84-100

This species feeds on pelagic fish, squid and crustaceans.

30. *Pseudorca crassidens* **false killer whale** skull DELPHINIDAE
Average Skull Length: 59 cm 8-11/8-11 X2 = 32-44

They normally feed on fish and cephalopods but occasionally may prey on other cetaceans as big as a humpback whale.

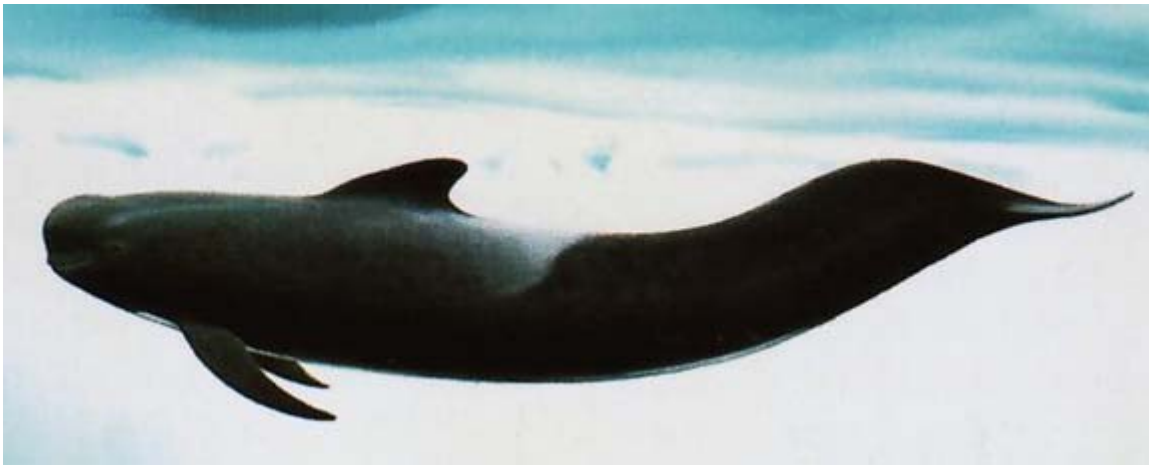


31. *Orcinus orca* **orca, killer whale** skull DELPHINIDAE
Average Skull Length: 87 cm 10-10/14-14 X2 = 40-56

Orcas feed on seals, fish, sea turtles, and large baleen whales.

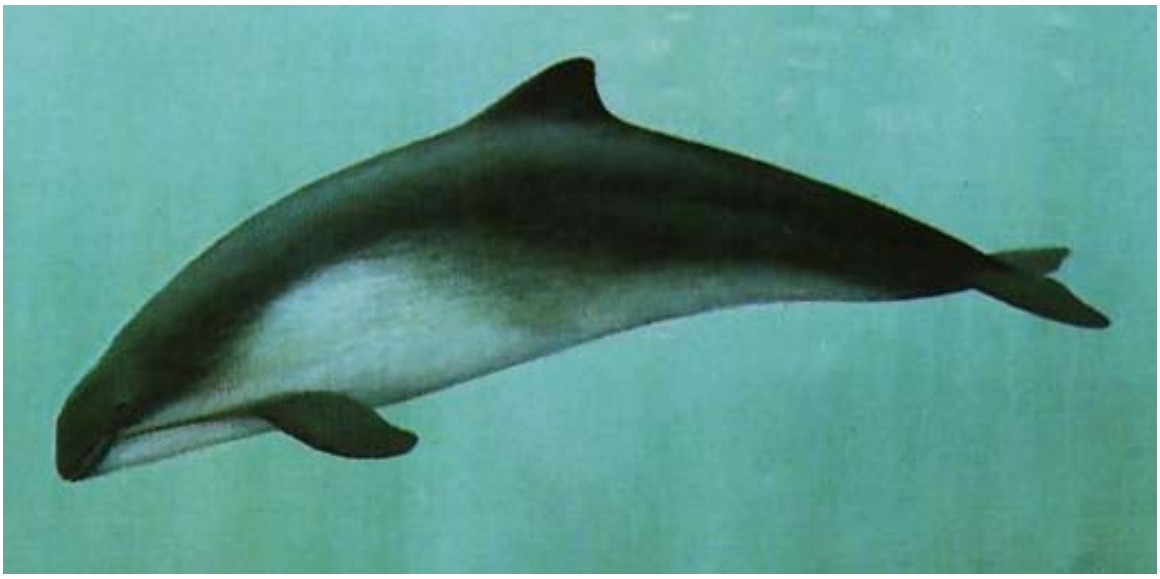
32. *Globicephala macrorhynchus* **shortfinned pilot whale** skull DELPHINIDAE
Average Skull Length: 56 cm 8-10/8-10 X2 = 32-40

They feed on squid and other cephalopods and small fish.

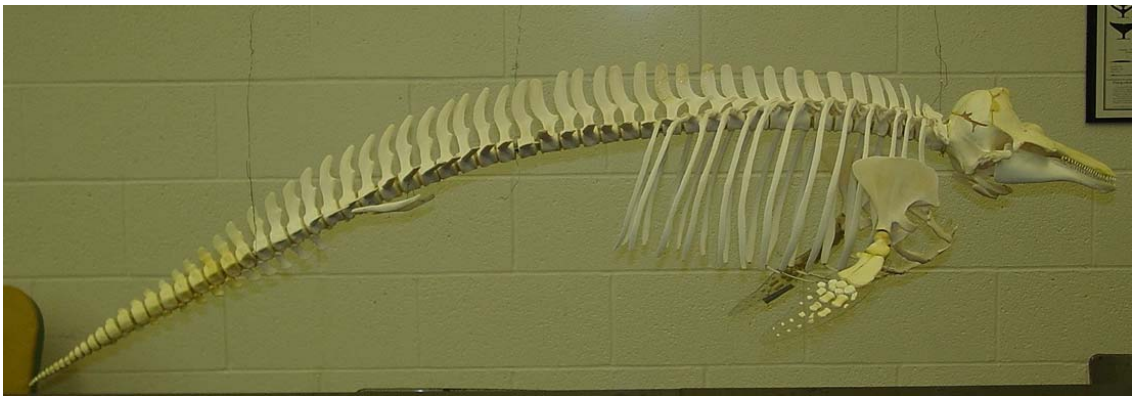


33. *Phocoena phocoena* **harbor porpoise** skull PHOCOENIDAE
Average Skull Length: 310mm 16-28/16-28 X2 = 64-112

This species uses its characteristic spade-like teeth when foraging for fish, squid, and crustaceans.



34. *Phocoena phocoena* **harbor porpoise** skeleton PHOCOENIDAE



35. *Phocoenoides dalli* **Dall's porpoise** skull PHOCOENIDAE
Average Skull Length: 29.5 cm 15-30/15-30 X2 = 60-120

Using its small spade-like teeth, it feeds mainly on fish and small squid.

36. *Arctocephalus tropicalis* **subantarctic fur-seal** skull OTARIIDAE
Skull Length 17 cm I 3/2, C 1/1, PC 6/5 X 2 = 36

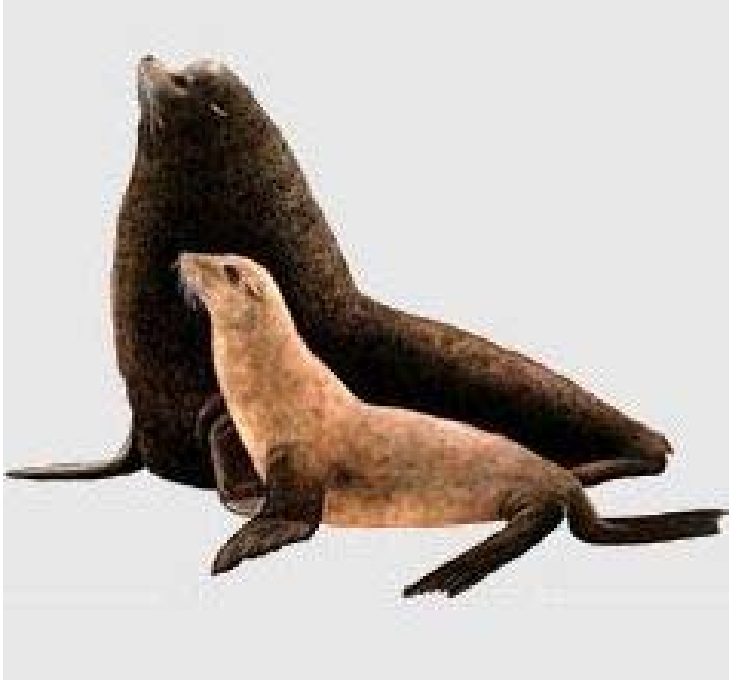
They feed on fish and squids.

37. *Callorhinus ursinus* **northern fur-seal** skull OTARIIDAE
Average Skull Length: 24 cm I 3/2; C 1/1; PC 6/5 X 2 =36

They are opportunistic feeders, eating the most abundant species available, depending on the season and their location during migration. Organisms commonly consumed include squid, herring, pollock, rockfishes and lantern fish.

38. *Zalophus californianus* **California sea-lion** skull OTARIDAE
Average skull length: 30 cm (♂); 24.5 cm (♀) I 3/2; C 1/1; PC 5-6/5-6 X 2 = 34-38

They feed on fish and squid near the ocean surface down to 30 meters but can dive to depths greater than 200 meters.



39. *Eumetopias jubatus* **northern sea lion** skull, baculum OTARIIDAE
Average skull length: 37 cm (♂); 28 cm (♀) I 3/2; C 1/1; PC 5-6/5-6; X 2 = 34-38

They normally feed on fish, squids and octopus, and occasionally on northern fur seal pups, harbor seals, and ringed seals.

40. *Phocarctos hookeri* **Auckland sea-lion** skull OTARIIDAE
Average skull length: 25 cm I 3/2; C 1/1; PC 6/5; X 2 = 32

They are opportunistic feeders with a diet of cephalopods, crabs, crayfish, and fish. They may eat penguins, fur seals, and sea lion pups on occasion.

41. *Otaria flavescens* **South American sea-lion** skull OTARIIDAE
Average skull length 33 cm I 3/2; C 1/1; p 4/4; M 1-2/1 X 2 = 34-36

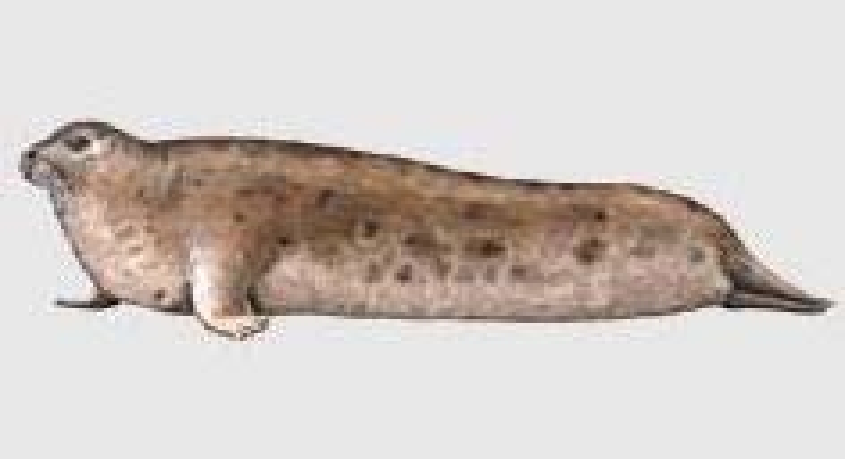
They are opportunistic feeders with a diet of demersal, pelagic, and bottom species including fish, cephalopods, and crustaceans. Females feed in coastal areas and on a larger variety of prey than the males.

42. *Odobenus rosmarus* **walrus** skull, pathological and normal baculum
ODOBENIDAE
Average skull length: 44 cm I 1/0; C 1/1; PC 3/3; X 2 = 18

Walruses feed mainly on benthic invertebrates, such as mollusks, echinoderms and crustaceans. They eat up to 45 kg per day.

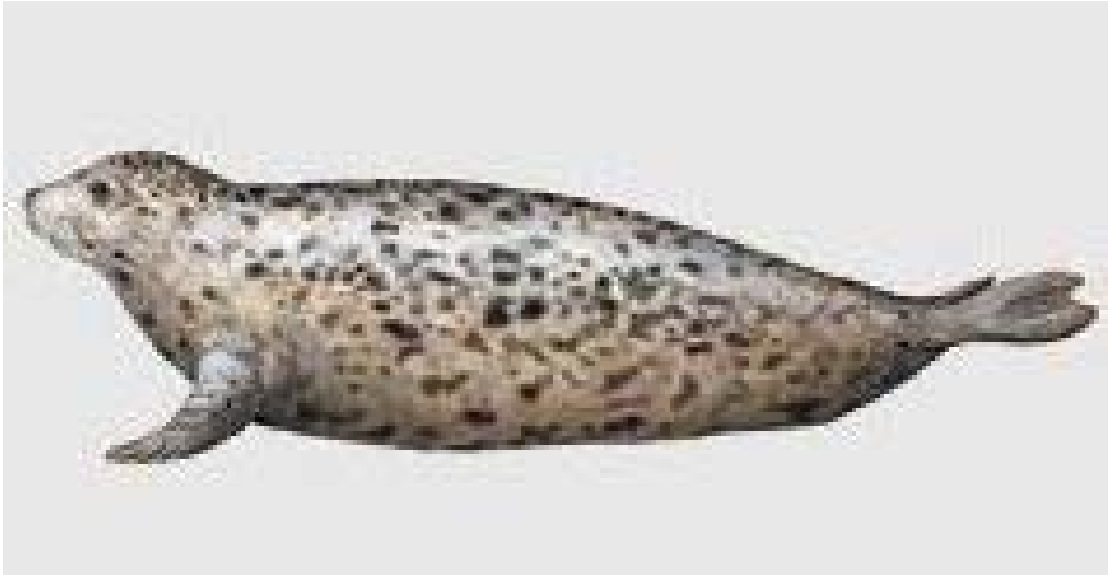
43. *Erignathus barbatus* **bearded seal** skull PHOCIDAE
Average skull length: 21 cm I 2-3/1-2; C 1/1; p 4/4; M 0-2/0-2; X 2 = 26-36

Their diet consists primarily of shrimps, crabs, clams, whelks, and some fish such as sculpin, flatfish, and cod. They feed in water depths of less than 230 m.



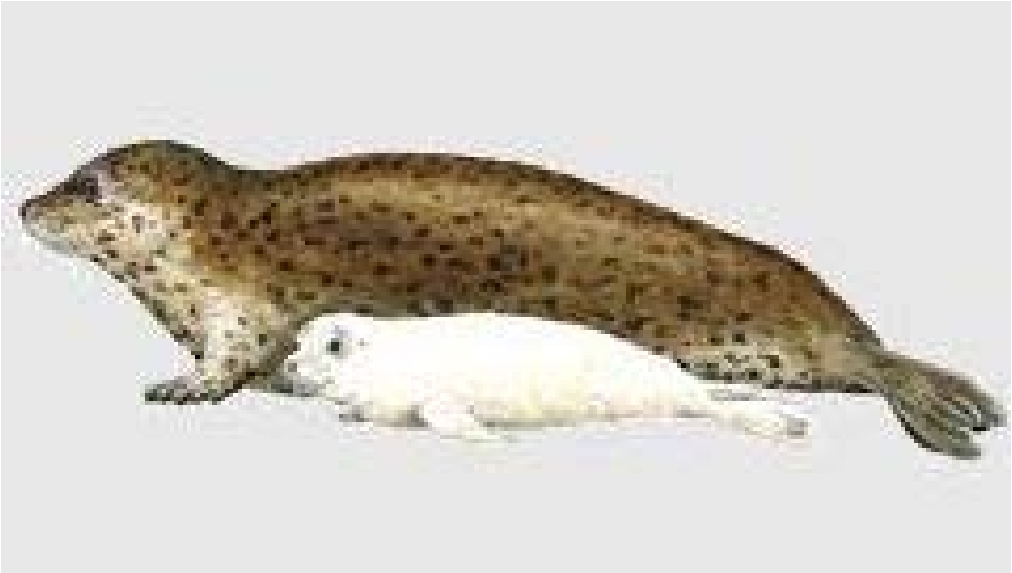
44. *Phoca vitulina* **harbor seal** skull PHOCIDAE
Average skull length: 20 cm I 3/2; C 1/1; pc 5/5; X 2 = 34

They primarily feed on crustaceans, mollusks, squid, and fish. The food is torn into chunks swallowed whole. The molars crush shells and crustaceans for swallowing, but food is generally not chewed.



45. *Phoca largha* **spotted seal** skull PHOCIDAE
Skull Length: 20.4 cm I 3/2; C 1/1; pc 5/5; X 2 = 34

They feed on a variety of food sources including crustaceans, cephalopods, and fishes.

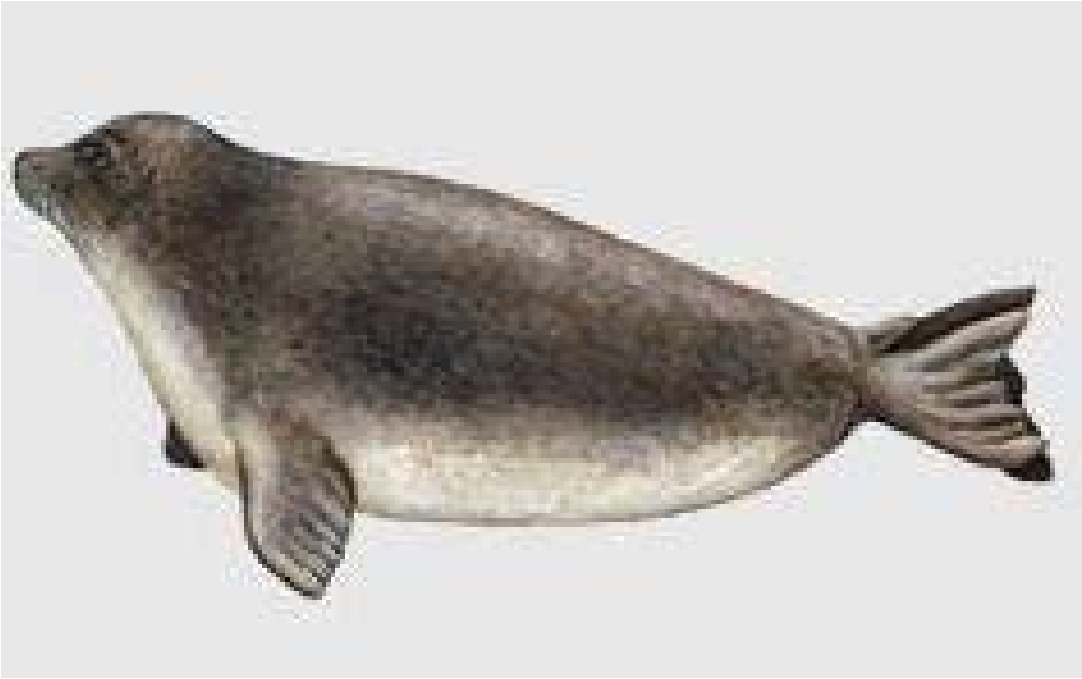


46. *Pusa hispida* **ringed seal** skull PHOCIDAE
Skull Length: 15.3 cm I 3/2; C 1/1; pc 5/5 X 2 = 34

They feed on fish, shrimp, amphipods, and euphausiids.

47. *Pusa sibirica* **Baikal seal** skull PHOCIDAE
Average Skull Length: 16 cm I 3/2; C 1/1; pc 5/5; X 2 = 34

Their main food source is the golomyanka, a type of bullhead fish that lives only in Lake Baikal



48. *Halichoerus grypus* **gray seal** skull PHOCIDAE
Average skull length: 27 cm I 3/2; C 1/1; pc 5/5; X 2 = 34

They feed on a wide variety of fish, crustaceans, and cephalopods. Sand eels or sand lances are the preferred prey in many areas. Gray seals also consume seabirds occasionally.

49. *Histriophoca fasciata* **ribbon seal** skull PHOCIDAE
Average skull length: 19 cm I 2/3; C 1/1; P 4/4; M 0-2/0-2; X 2 = 26-36

They feed on feed on fish, shrimp, squid, and crabs.

50. *Pagophilus groenlandicus* **harp seal** skull PHOCIDAE
Average skull length: 20 cm I 3/2; C 1/1; pc 5/5; X 2 = 34

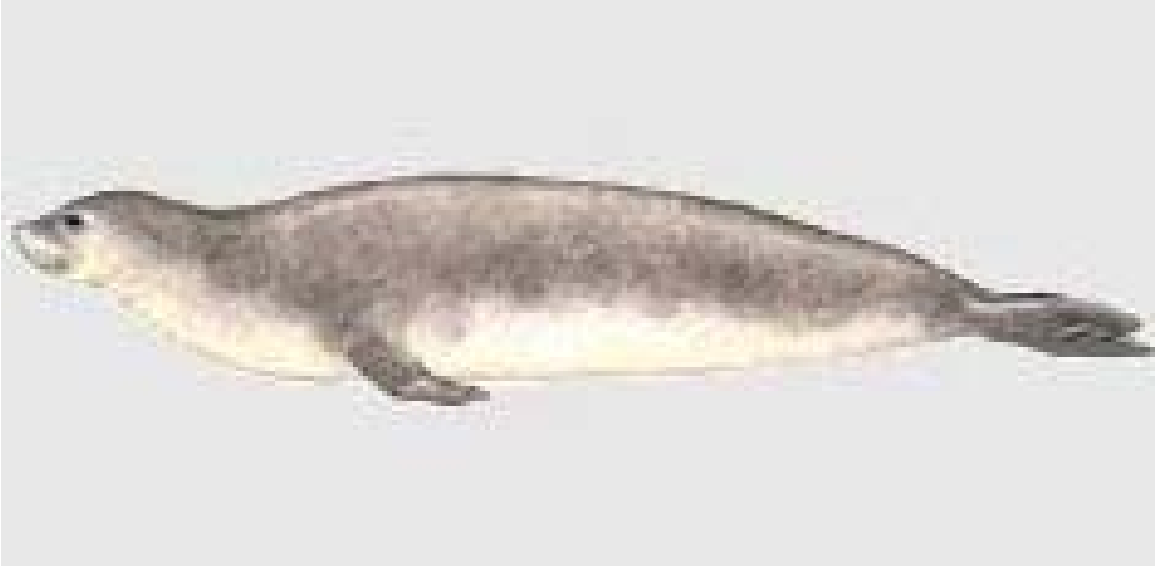
They feed on fish, amphipods, euphausids (krill), shrimps and prawns.

51. *Cystophora cristata* **hooded seal** skull PHOCIDAE
Skull Length: 25.5cm I 3/2; C 1/1; pc 5/5 X 2 = 34

They feed on fish, octopi, squids, shrimps and mussels.

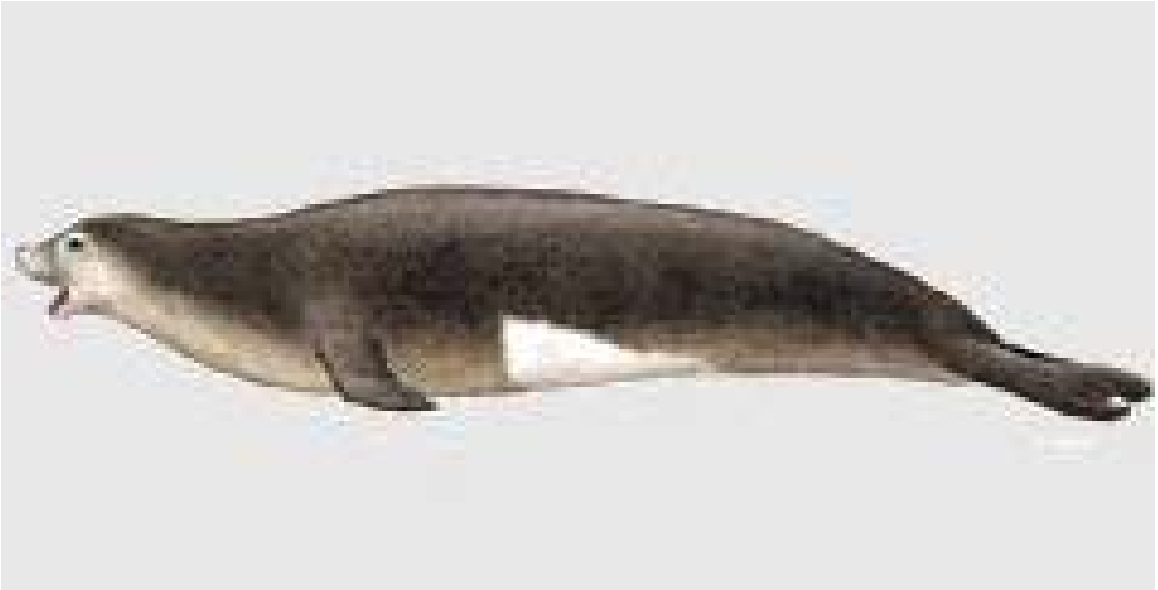
52. *Monachus tropicalis* **Caribbean monk seal** skull PHOCIDAE
Average skull length: 22 cm I 2/2; C 1/1; pc 5/5; X 2 = 32

They probably ate fish, lobsters, and octopi.



53. *Monachus monachus* **Mediterranean monk seal** skull PHOCIDAE
Skull Length: 31.75cm I 2/2; C 1/1; pc 5/5 X 2 = 32

They feed on a large variety of fish as well as lobsters and octopi.

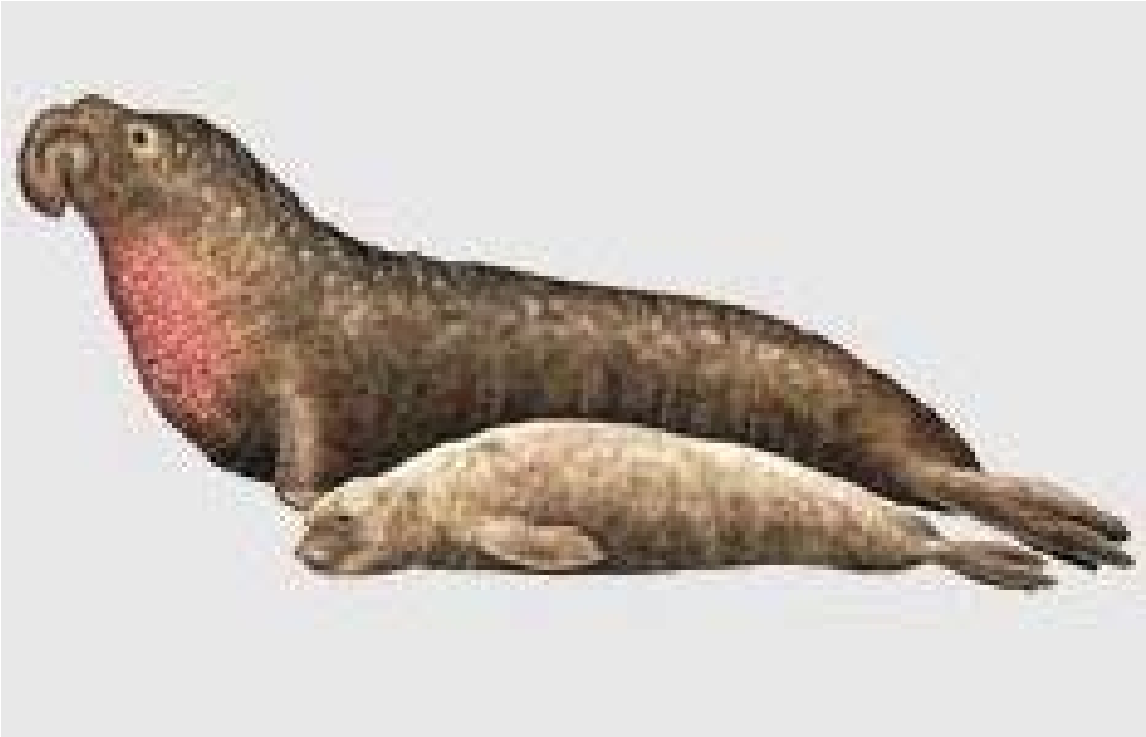


54. *Monachus schauinslandi* **Hawaiian monk seal** skull PHOCIDAE
Skull Length: 24.2cm I 2/2; C 1/1; pc 5/5 X 2 = 32

They feed on fish, octopi, and lobsters.

55. *Mirounga angustirostris* **northern elephant seal** skull, baculum PHOCIDAE
Average skull length: 61 cm (♂); 33 cm (♀) I 2/2; C 2/2; pc 5/5; X 2 = 36

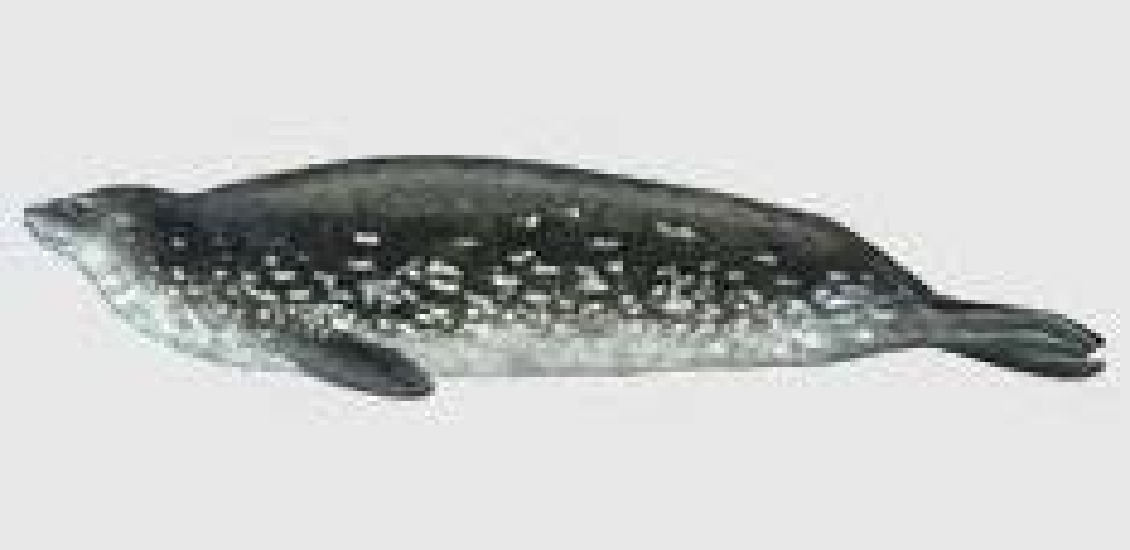
They feed on cephalopods, bony fishes, skates, rays, sharks, and pelagic red crabs.



56. *Mirounga angustirostris* **northern elephant seal** skeleton PHOCIDAE

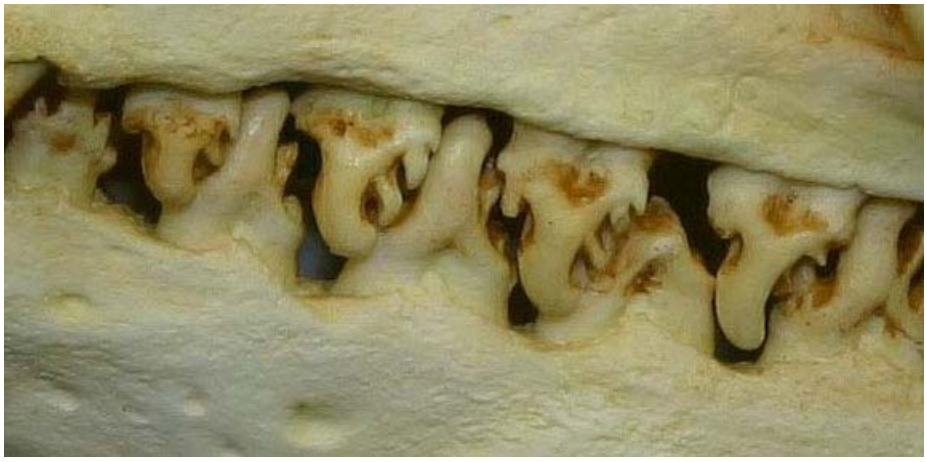
57. *Leptonychotes weddellii* **Weddell seal** skull PHOCIDAE
Skull Length: 28cm I 2/2; C 1/1; pc 5/5 X 2 = 32

They feed on fish, cephalopods, and crustaceans.



58. *Lobodon carcinophaga* **crabeater seal** skull PHOCIDAE
Average skull length: 28.5 cm I 2-3/2-3; C 1/1; P 4/4; M 0-2/0-2; X 2 = 26-36

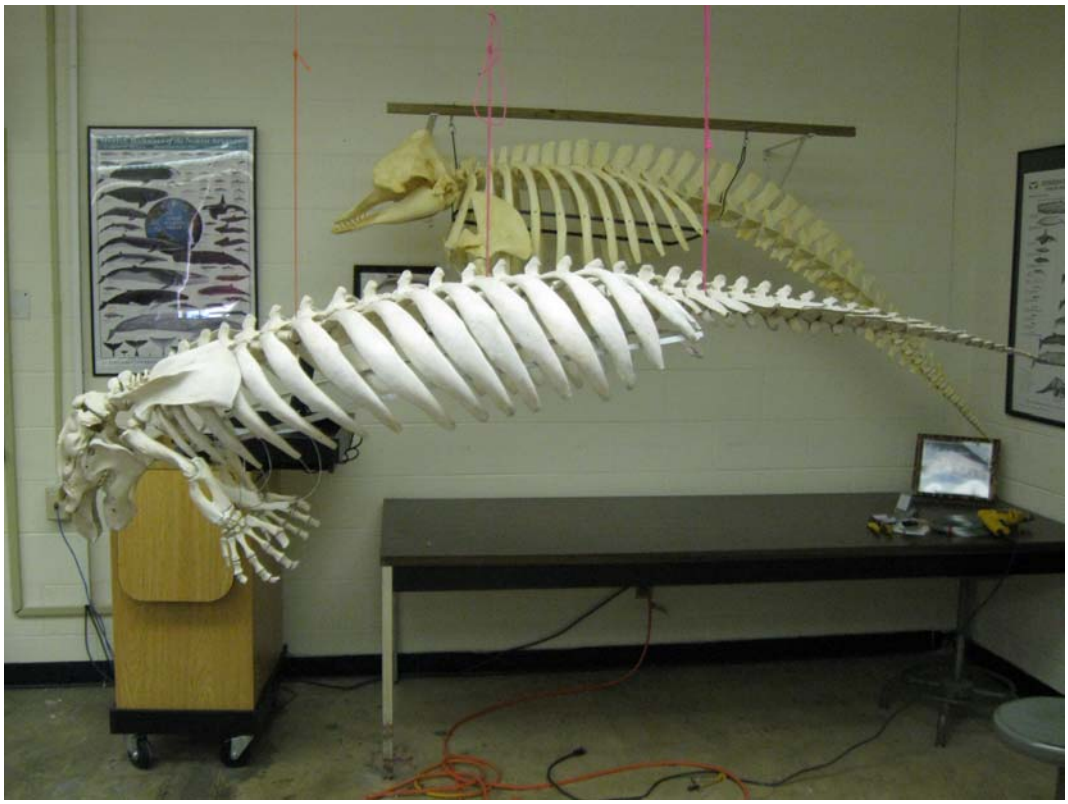
They have distinctive and complex teeth. Each tooth has tubercles, or bony protuberances, with spaces between them. The upper and lower jaws fit together so that when mouth is closed, the teeth and tubercles can strain krill. Thus, krill, such as *Euphausia superba*, not crab, are their primary food. They feed by swimming through schools of krill with their mouth open, sieving the water out using their sophisticated teeth. When they migrate outside the Antarctic, crabeater seals are thought to feed on other invertebrates as well as small fishes.



59. *Hydrurga leptonyx* **leopard seal** skull PHOCIDAE
Skull Length 35cm I 2/2; C 1/1; pc 5/5 X 2 = 32

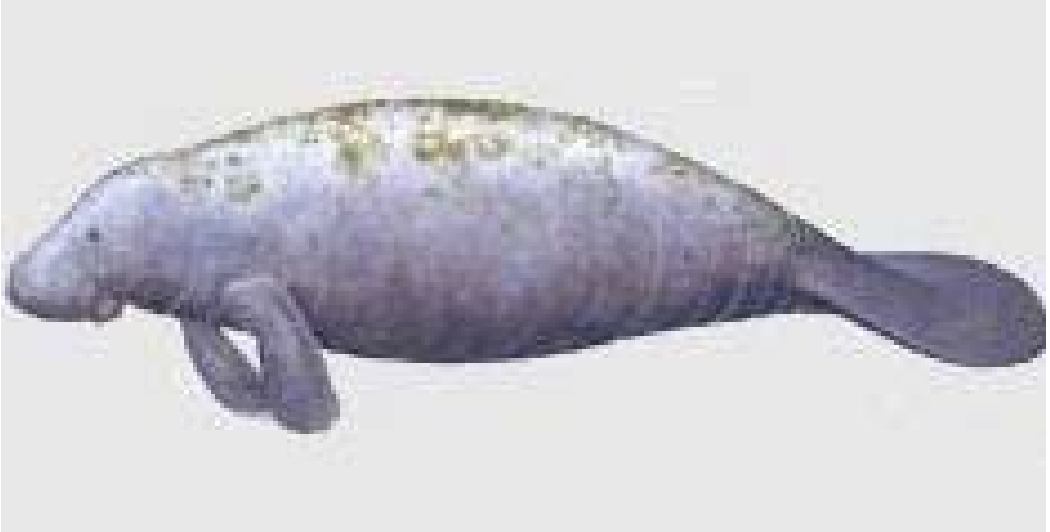
They feed on krill, other seals, penguins, fish and cephalopods.

60. *Trichechus manatus* **Caribbean** (**West Indian) manatee** skeleton
Length: 336 cm ♀ TRICHECHIDAE



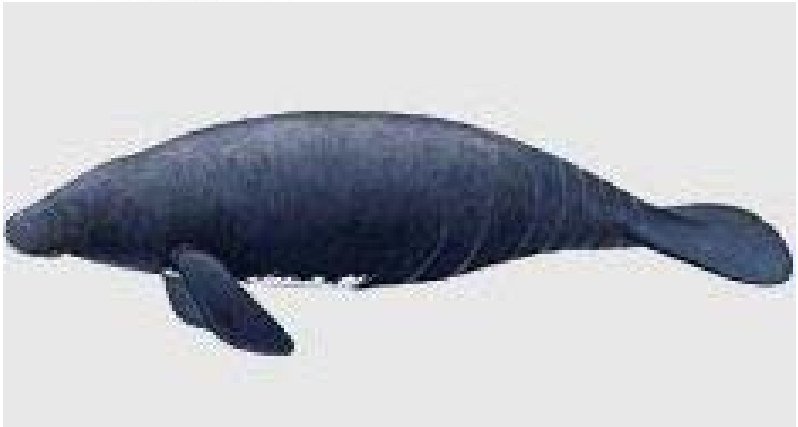
61. *Trichechus manatus* **Caribbean (West Indian) manatee** skull, scapula, humerus, hand TRICHECHIDAE
Average skull length: 35 cm I 0/0; C 0/0/; P 0/0; M 6/6; X 2 = 24

Manatees are completely herbivorous. They eat aquatic plants which they graze along water bottoms and on the surface. Because of the low nutritional value of the plants consumed, manatees must graze for 6-8 hours a day. Each day they consume 5-10% of their body weight, which can be over 100 kg in a large individual. Manatees feed on abrasive plants and, as a result, their molars are continually replaced throughout life as they wear down. Hind-gut fermentation is another adaptation to the herbivorous diet of the manatee, aiding in breaking down the cellulose of the plants eaten.



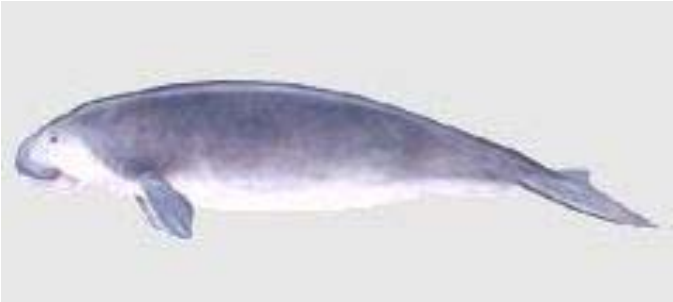
62. *Trichechus inunguis* **Amazon manatee** skull TRICHECHIDAE
Average skull length: 33 cm I 0/0; C 0/0/; P 0/0; M 6/6; X 2 = 24

The Amazonian manatee is a herbivore that feeds on aquatic vegetation near lake edges, such as aquatic grasses, and floating vegetation such as water lilies. Captive adults daily consume from 9 - 15 kg (20 - 33 lb) of leafy vegetables.



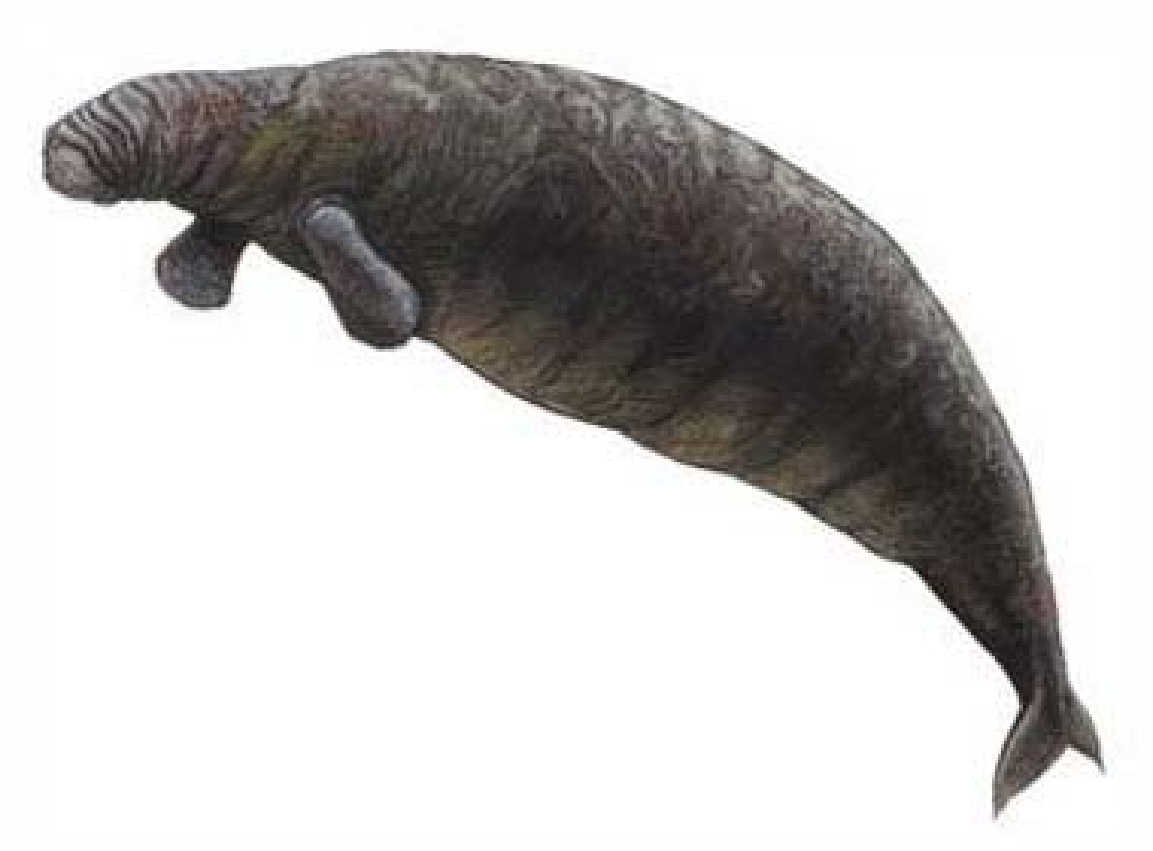
63. *Dugong dugon* **dugong** skull DUGONGIDAE
Average skull length: 29 cm I 1/0; C 0/0; P 0/0; M 3/3; X 2 = 14

Dugongs feed on flowering sea grasses of the families Potamogetonaceae and Hydrocharitaceae. Also reported to occasionally eat algae, and crabs have also been found in the stomachs of dugongs. The lower lip and distal parts of the palate have horny pads used to grasp vegetation, which is then uprooted with the strong upper lip. Dugongs have 10-14 teeth in adults. The molars are rootless, circular in cross-section and lack enamel, males have long, tusklike incisor teeth.



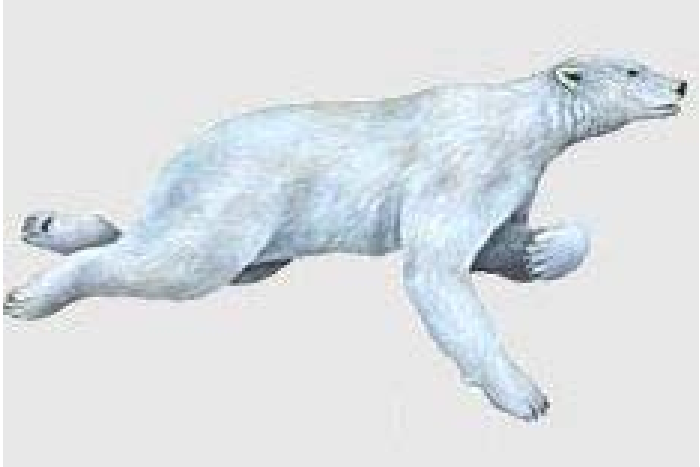
64. *Hydrodamalis gigas* **Steller's sea-cow** skull DUGONGIDAE
Average skull length: 62.2 cm I 0/0; C 0/0; P 0/0; M 0/0; X 2 = 0

They ate sea algae lying near the surface, sea grasses, but primarily soft kelp. Since lacking teeth, it ground its food by its deeply grooved keratinous plates in the mandibles. Seasonal food availability may have been a problem for the Bering Sea population, as Steller described individuals losing enough weight during the winter months to cause their ribs and vertebrae to be visible under the skin.



65. *Ursus maritimus* **polar bear** skull, baculum and claw URSIDAE
Average Skull Length: 46 cm I 3/3; C 1/1; p 4/4; M 2/3; X 2 = 42

They feed on seals and walruses.



66. *Ursus maritimus* **polar bear** hide URSIDAE

67. *Enhydra lutris kenyoni* **sea otter** skeleton MUSTELIDAE
Length: 1.5 m ♂

68. *Enhydra lutris* **sea otter** skull, baculum MUSTELIDAE
Average skull length: 14.5 cm I 3/2; C 1/1; p 3/3; M 1/2 X 2 = 32

Sea otters eat clams, crabs, snails, starfish, abalone, and 40 other marine animal species.

69. *Enhydra lutris kenyoni* **sea otter** pelt MUSTELIDAE

Length: 1.5 m ♂

Sources

The artwork of *Pakicetus*: http://critters.pixel-shack.com/WebImages/crittersgallery/Pakicetus.jpg

The artwork of *Ambulocetus*: http://cgi.ncrv.nl/ncrv/walkingwithbeasts/evidence/prog1/images/evi_amulocetus_large.jpg

The artwork of *Remingtonocetus*: http://kawa3104.hp.infoseek.co.jp/remingutoketasu.JPG

The artwork of *Protocetus*: http://www.museumkennis.nl/sites/nnm.dossiers/contents/i003354/wal.2003%20protocetus.jpg

The artwork of *Basilosaurus*: http://www.museumkennis.nl/sites/nnm.dossiers/contents/i003354/wal.2005%20basilosaurus.jpg

The artwork of *Dorudon*: http://www.museumkennis.nl/sites/nnm.dossiers/contents/i003354/wal.2004%20dorudon.jpg

The picture of *Ambulocetus natans* is from the website of Research Casting International (http://www.researchcasting.ca/overview.htm) who are the suppliers of the skull cast at the Arkansas State University teaching collection.

Pictures of skulls, bacula, sperm whale tooth, pygmy sperm whale skeleton, goosebeak (Cuvier's beaked) whale teeth, Hubbs' beaked whale teeth, narwhal tusk, and polar bear's claw are from Skulls Unlimited (http://www.skullsunlimited.com/) who is the supplier of those casts to the teaching collection at Arkansas State University. For the fossil sperm whale tooth the picture was obtained from BoneClones (http://www.boneclones.com/catalog_marine_mammals.htm) who are the suppliers of that cast.

The artwork representing the different species is from: T.A. Jefferson, S. Leatherwood and M.A. Webber. Marine Mammals of the World http://ip30.eti.uva.nl/BIS/marine_mammals.php?menuentry=zoeken&id=&selected=wetenschap