Fish have been kept as pets for centuries. Concepts of pet fish husbandry originated with carp species in China. During the 20th and 21st centuries, fish have been a staple of the pet industry. Renewed interest in aquatic ponds and gardens has created a demand for more expertise in aquatic animal husbandry, environmental quality, and physiology and medicine. In a survey regarding household pets, pet fish numbered 49.3 million in the United States. A 2005/2006 national pet owners survey conducted by the American Pet Products Manufacturers Association indicated that 14.7 million American households own fish (i.e., 139 million freshwater species; 9.6 million marine animal species). The Ornamental Aquatic Trade Organization recently estimated the pet fish population in the United Kingdom to be more than 144 million (an average of 2.5 pet fish per UK resident).

Only 10 years ago, anesthesia, surgery, and advanced imaging were uncommon in aquatic patients. Today, these procedures are routine. The Animal Health Department at the New England Aquarium (NEAq) manages an average of 20 to 30 clinical piscine cases per month and performs approximately 25 surgeries on fish per year. In addition, as part of its veterinary program, the institution handles several hundred fish annually for routine quarantine procedures. This number of cases is attained because the NEAq is a public aquarium housing 25,000 animals that represent 500 to 600 species from freshwater, brackish water, and marine environments.

Diagnostic evaluations for fish are similar to those for small and other exotic animals. Evaluations of fish can include water and environmental quality testing, skin scrapes, gill biopsies, anal lavage to obtain feces for testing, coelomic centesis, organ or mass biopsies, complete blood counts (CBCs), blood chemistry profiling, bacteriologic culture and antimicrobial sensitivity testing, ultrasonography, and radiography. Less common diagnostic and treatment options include laparoscopy, endoscopy, exploratory surgery, contrast radiography, cryosurgery, and radiation therapy.

With greater husbandry knowledge and baseline data for piscine patients, practitioners can...
begin to encourage clients to consider treatment of their aquatic pets. Through the diagnosis and treatment of piscine diseases and conditions, a greater body of data will become available, providing valuable information for treating aquatic patients more thoroughly and humanely. The medical management of aquatic patients is not only exciting and challenging but also achievable, practical, and necessary.

**BASIC DIAGNOSTICS**

Many standard diagnostic tests can be used for fish. In addition, several simple tests that are more specific to fish are commonly conducted. With fish, as with other species, diagnostic test results must be interpreted with consideration of a thorough patient history, environmental conditions and water quality, physical examination, and clinical appearance. When an ill fish is presented to the NEAq Animal Health Department, the baseline diagnostic routine includes a skin scrape, a gill biopsy, a CBC, a blood chemistry profile, radiography with or without abdominal ultrasonography, a fecal or anal wash, an anal culture (enteric flora and mycobacterial culture), a physical examination, morphometrics, and a water chemistry profile. This clinical information, in combination with the patient’s clinical history, can help create an appropriate differential diagnosis and treatment plan for piscine patients.

**History**

Obtaining a thorough history of a piscine patient, a collection of fish, an aquarium or aquaria, or a pond is of utmost importance. The history should include the origin of the fish (pet store, breeder, wild caught versus captive bred), amount of time the fish has been in its current environment, medical and reproductive histories, feeding behavior, food preferences, and current husbandry. The medical history is vital because many clients receive advice from local pet stores and may have already treated the tank or pond with a number of different chemicals. Important husbandry details include the water-volume, tank or pond location, substrate type, furnishings, lighting type, photoperiod, diet, water temperature, water-quality parameters, frequency and source of water changes, habitat additions and modifications, number and types of plants, and animals sharing the aquatic environment. The owner should also be questioned about social, reproductive, or aggressive behaviors of the fish.

If a clinician cannot visit the site for direct observation, digital pictures and/or video of patients in their aquatic habitats is extremely helpful, especially if the abnormal behavior can be filmed. If a “normal” conspecific can accompany the patient to the hospital, comparisons can be made between the normal and affected fish. The diet, behavior, and anatomy of fish may vary significantly with season, age, and gender. The owner and veterinarian should work cooperatively to obtain thorough natural history information about the species to help understand normal seasonal, sexual, and age-related physical changes.

The owner should transport the patient in a couple liters of water from the patient’s tank or pond. This water can be used during the examination as long as the water quality is normal when tested. A separate sample of the patient’s tank or pond water can be tested for chemical- and physical-quality parameters. Additional water can also be used to make an anesthetic bath for the fish. The temperature and aeration of the transport water must be maintained during the trip to the hospital, throughout the examination, and during the trip home. It may be necessary to heat or chill the water while the fish is away from its home environment and to provide aeration using a battery-operated air pump, airline tubing, and an airstone, which can be purchased at pet stores.

**Water Quality**

Quality profiles for the water’s chemical and physical parameters should be assessed to determine environmental quality. It is critical to have clients bring a sample of water from their tank or pond. The sample should be kept cool and separate from the fish during transport to help ensure that it represents current water conditions in the aquatic system. Parameters that should be analyzed include temperature; dissolved oxygen; pH; ammonia,
nitrite, and nitrate content; alkalinity; and salinity. Certain chemical additions (intentional or unintentional; e.g., copper sulfate, chlorine) may also be measured or tested. Many clinical problems in aquatic animal medicine result from cumulative stress, and poor water quality is often one of the precipitating factors. Common problems in the aquatic environment include sudden changes in pH, temperature, alkalinity, or salinity; low dissolved oxygen; waste accumulation resulting in high ammonia, nitrite, or nitrate levels; the presence of chlorine or chloramines; the presence of heavy metals; and/or low levels of essential elements and ions, such as iodine. In some instances, intensive monitoring of environmental quality may be required to detect pollutants and/or identify microbiologic agents, such as protozoans, bacteria, fungi, and algae.

Anesthesia

Most species at the NEAq are restrained using tricaine methanesulfonate (MS-222) in a preparation called Finquel (Argent Chemical, Redmond, WA). Although Finquel is FDA approved for use in many species of food fish, many animals in the ornamental trade and in public aquaria require off-label use. In most species at the NEAq, anesthesia is induced and maintained at 50 to 90 mg/L, and maintenance adjustments are determined during the procedure while the patient is being monitored for opercular movements, gill perfusion, and heart rate. Anesthetic is added directly to the water and mixed to distribute it uniformly. The use of an air pump and airstone in the water helps maintain the oxygen level and, therefore, adequate respiration. A separate container of appropriate recovery water should also be prepared in case the anesthetic depth becomes too great or the patient exhibits a poor response to chemical sedation. Because MS-222 is highly acidic, when using it in poorly buffered freshwater, an equal amount (50 to 90 mg/L) of sodium bicarbonate should be added to the anesthetic concentration. Although gill pathology has been documented at the NEAq when using high concentrations (>300 mg/L) of MS-222 for euthanasia in elasmobranchs (i.e., sharks, rays, skates), no histopathologic changes have been noted in hundreds of marine species when using 50 to 90 mg/L of unbuffered solution in seawater.

If opercular movements cease when a patient is sedated, the patient should be moved immediately to the recovery water. Respiration can be augmented by gently injecting recovery water through the mouth and over the gills using a 60-ml syringe. It is important to have the water flow cranially to caudally (through the mouth and over the gills) because fish have a counter-current exchange of blood flow through the gill filaments to help optimize waste excretion and increase oxygen uptake (Figure 1).

To allow normal oxygen transfer across the gills, the clinician should try to perform most of the examination, including palpation and the physical examination, while the fish is completely or partially in the water. When necessary, the fish can be removed from the water for a short time, but respiration reduces greatly as the gills start to dry. Out-of-water examinations and procedures should be limited to 30- to 90-second periods, unless the fish’s body and gills are moistened periodically with the anesthetic bath water. Returning the patient to the anesthesia water for 30 to 60 seconds allows respiration, after which the examination can be continued. With experience, examinations of fish can be efficient and safe. Longer examinations can be performed using a piscine anesthesia machine.

Physical Examination

To begin the physical examination, the piscine patient should be observed while it swims and the following noted:

- Abnormal behavior or swimming patterns
- Body condition
- Anatomic conformation and abnormalities of the eyes, skin, or fins
- Buoyancy problems
- Abnormal fin movement

Fish with dermal irritation often twist rapidly, appearing to scrape their body on hard surfaces and briefly
exposing their ventrum. This behavior is often called flashing and may be a sign of parasitic or bacterial skin disease. The frequency and intensity of gill movements should be noted because respiration can be compromised by a number of water-quality problems or infectious agents. Gilling rates (i.e., the number of times per minute that the operculum opens and closes) greater than 60/min or less than 10/min may be seen with respiratory compromise. Opercular movements should appear symmetric. Respirations can increase normally throughout the day, particularly around feeding time. Increased respiratory effort can be marked by exaggerated flaring of the operculum. Fish that have died from hypoxia often have flared opercula and exposed gills. Respirations vary greatly among species (e.g., catfish versus trout) and seasons (e.g., winter versus summer for fish in ponds). Piping at the surface is a sign of respiratory difficulty in many species.

The body-scoring system in Table 1 may be used for fish. Body condition can help distinguish chronic wasting from acute anorexia. Failure to provide the proper diet can lead to poor nutrition and cause several disease-related syndromes in fish. When assessing abdominal distention, the clinician must differentiate obesity from ascites, neoplasia, air or gas, a foreign body, generalized edema, or gonadal development. Severe edema in fish can cause the external, unattached edges of scales to rise off the body wall, resulting in a “pinecone” appearance. This nonspecific clinical sign of edema is also called dropsy, which is commonly used in the lay literature.

Fish can be manually restrained for many basic procedures; however, a more thorough physical examination requires anesthesia. Chemical restraint can decrease the risk for skin abrasion, damage to the epidermal mucous layer, eye injury, or other physical trauma.

Latex gloves should be worn during the examination to minimize damage to the fish’s integument and mucous layers as well as to protect the handler from zoonotic disease, such as mycobacteriosis. The spines in the dorsal and ventral fins, operculum, and caudal peduncle (base of tail) can be a hazard for the handler. Chamois cloths can effectively protect the animal’s integument during handling for digital imaging or transport from the anesthetic bath to the examination table. For most fish species, chamois often does not greatly abrade the integument. Synthetic chamois cloths tend to be used more often than natural ones because synthetic ones last longer and may have holes that allow water to drain through, preventing them from getting as heavy as natural chamois. Low-friction nylon stretchers and plastic or rubber totes are available for moving large patients from tanks, exhibits, or ponds to the examination area. Only trained personnel should handle venomous or toxic species, and an emergency plan should be available for these cases. Many public aquaria, zoos, and high-end aquarium fish retailers are excellent sources of information regarding which species are toxic and/or venomous and how they should be handled.

A systematic approach to the piscine physical examination follows:

Observe the integument for color or texture changes; abrasions and lacerations; ulcerations; excessive mucus; areas of missing scales; white, black, or red spots; and external parasites. The integument is best viewed while the patient is in water. External parasites may not be as visible when the patient is on an examination table. The use of a magnifying glass may help identify some of the larger metazoan parasites. The use of fluorescein ophthalmic stain to highlight breaches in fish integument has been described.

Observe the gills by elevating the operculum with sterile swabs. Gills should be deep red, uniformly shaped, and even along the outer margins. Note ragged edges, blunting, mottling, or pallor. Pallor generally indicates poor perfusion or anemia, whereas brown discoloration may be seen with nitrite toxicosis.

The heart rate can be determined using a handheld Doppler ultrasound device or standard ultrasonography.

Noga E: American College of Veterinary Pathologist Fish Pathology Workshop, 2005.

### Table 1. Body-Scoring System for Fish

<table>
<thead>
<tr>
<th>Score</th>
<th>Description</th>
</tr>
</thead>
<tbody>
<tr>
<td>6</td>
<td>Obese</td>
</tr>
<tr>
<td>5</td>
<td>Moderately overweight</td>
</tr>
<tr>
<td>4</td>
<td>Normal</td>
</tr>
<tr>
<td>3</td>
<td>Mildly underweight (e.g., a thin cranial ventral abdomen; intact musculature along the spinal column)</td>
</tr>
<tr>
<td>2</td>
<td>Moderately underweight (e.g., the vertebral column is visible)</td>
</tr>
<tr>
<td>1</td>
<td>Severely underweight (e.g., a “lollipop” appearance: a large head and severe muscle atrophy directly behind the head along the spinal column)</td>
</tr>
</tbody>
</table>
Place the ultrasound probe on the ventral abdomen just anterior to the pectoral fins and caudal to the operculum. Heart rates vary in different species of fish and are primarily monitored during anesthesia. The piscine heart has four components in series: a sinus venosus, atrium, ventricle, and bulbus or conus arteriosus. In general, blood flowing through the heart is deoxygenated and pumped to the gills for oxygenation. From the gills, oxygenated blood travels to the rest of the body before returning to the heart. Blood pressure and pulse oximetry are being investigated for more regular clinical applications in fish.

Most of the neurologic and buoyancy examination should be performed while observing the fish in the transport container when it first arrives. Can the fish maintain a normal body position in the water column? Can it move its fins and operculum? Is the fish using the appropriate fins for locomotion and stability (e.g., a mako shark uses its caudal fin for movement and pectoral fins for stability, whereas most seahorse species use the pectoral fins for locomotion and caudal fin for grasping to substrate)?

Examine the eyes with a penlight and ophthalmoscope. Note that fish pupils are generally fixed and non-responsive to light and that the lens normally protrudes slightly through the pupil. Common eye lesions, such as corneal edema, cataracts, lens luxation, and hypopyon, can easily be detected. Mechanical injury to the eyes is common in large-eyed species. Examine the cornea with fluorescein ophthalmic stain and an ultraviolet light to detect corneal abrasions and ulcers. The presence of gas bubbles within the eye may occur with gas supersaturation of the water, intraocular disorders, or systemic illness. Moderate to severe exophthalmos or buphthalmos may occur secondary to any ocular or systemic problem in fish. The trematode *Neobenedinia melleni* has caused corneal edema and permanent ocular damage in several species of Atlantic reef fish at the NEAq.

With the use of a penlight, examine the oral cavity for asymmetry, broken teeth or bones, masses, foreign material, bleeding, or other abnormalities. To examine animals with large teeth and strong jaws, use bite blocks with appropriately sized segments of polyvinyl chloride piping. The pipe can be padded with clean vet wrap to protect the teeth. In large sharks, the use of polyvinyl chloride has allowed manual removal of ingested foreign bodies in the back of the oral cavity. Palpate the coelom extending from the ventral base of the pectoral fins to the vent and laterally to the vertebral column. Palpate and visually examine the musculoskeletal system, noting asymmetry, atrophy, deformities, and other abnormalities. Weigh the patient, and measure its total length (i.e., tip of the rostrum to the tip of the tail; using a straight rather than curved measure) and girth (i.e., width immediately posterior to the dorsal fin). Many fish biologists also use a quantitative measure called condition factor to determine the robustness of individual fish in a population. Condition factor is calculated using the following formula:

$$\text{Condition factor} (K) = 10^n \times \frac{\text{Weight (g)}}{\text{Length}^3 (\text{mm})}$$

(This is an example for salmonid fish; $n=5$.) This calculation is not commonly used in clinical practice but may be valuable when monitoring population trends in aquaria or ponds for similarly aged members of the same species.

**Skin Scrape**

A skin scrape should be performed on every sick fish and during routine quarantine examinations. This procedure can be performed without sedation in most fish and often detects bacteria, fungi, algae, and protozoan and metazoan parasites. Some parasites are more likely to be found at the base of fins, where they are less likely to be dislodged by hydrodynamic forces. Sample the epidermis by holding a coverslip at a 45° angle to the integument and, with mild pressure, scraping skin cells and mucus from the lateral flank from the base of the pectoral fins or dorsal fin, if possible. Scrape only a few centimeters of the fish in the direction of head to tail. If this is performed correctly, there should be a layer of mucus on the
edge of the coverslip (Figure 2). To prevent drying of the specimen, examine the sample as soon as possible using a light microscope. A drop of freshwater or saline that corresponds to the patient's typical environment can be added to the slide. Slides are best viewed by starting at 100× normal magnification and increasing to 400×. While the sample is still moist, the slide should be examined using both light- and dark-field condensers to detect slight movement of certain parasites. Fluorescein ophthalmic stain can be used to detect breaches in the skin integument, as described by Dr. Edward Noga of North Carolina State University.

Gill, Fin, and Skin Biopsies

A gill biopsy should be performed on all sick fish and during routine quarantine examinations. This procedure may be performed using manual restraint for some fish, but strong, active fish may require sedation. Lift the operculum gently as during the gill examination. Obtain a biopsy sample of the tips of a few primary lamellae using sharp, clean suture scissors perpendicular to the primary lamellae (Figure 3). Make a clean cut and avoid pulling or tearing the lamellae. The sample will cling to the scissors. Bleeding may occur but usually stops quickly; mild pressure or cautery using silver nitrite can be used on the cut to stop excessive or prolonged bleeding. Carefully transfer the gill tips to a microscope slide with a drop of water and place a coverslip on the sample. Examine the gill sample under a light microscope as soon as possible. Use both light and dark fields, if available, focusing up and down on the tissue structure. Gills should have a consistent structure and pattern of primary and secondary lamellae. Abnormalities may include cartilage damage; the presence of metazoan or protozoan parasites, bacteria, or fungi; hyperplasia of gill tissue; stunted or collapsed secondary lamellae; clubbing of the gill lamellae; and excess mucus.

Fin and skin biopsies can be valuable if skin scraping results have not been informative. A small wedge of tissue can be excised from between the rays of the fin using sharp scissors. Full-thickness skin biopsies can be performed with a scalpel, scissors, or a dental punch biopsy instrument. The skin and protective mucous layer can be easily disrupted and damaged in fish. Many undiluted surgical scrubs and surgical preparation techniques can damage the fish integument, making patients more susceptible to secondary bacterial, fungal, and protozoan infections. The biopsy site should be rinsed with copious amounts of sterile saline or freshwater, depending on the species. The site can be cleaned using a dilute povidone–iodine solution (1:20) or Virkon (Dupont Animal Health; 0.2%). The resulting wedge of tissue can be used for cytology, histopathology, culture, and/or molecular diagnostics at the discretion of the clinician. Depending on the size of the biopsy site, it can be left open or closed using synthetic suture. Because suture material seldom dissolves in fish, it should be rechecked and removed in 2 to 3 weeks.

Hematology

Blood sampling is an excellent diagnostic tool for assessing piscine health. In general, a maximum blood volume of 0.5% to 1% of body weight may be safely collected. With the reduced sample volume requirements of some modern blood chemistry analyzers, a full chemistry panel can be obtained with as little as 0.1 ml of whole blood. Small volumes can also provide a hematocrit and blood smear, from which an estimated total leukocyte count and cell differential can be obtained. With larger sample volumes, more precise automated CBCs can be obtained using laser flow cytometry.

Blood is generally collected with the patient under sedation, unless it is severely compromised and may not survive anesthesia. Venipuncture is commonly performed using the caudal vein, which is ventral to the vertebral column in the area of the tail. This site can be approached either laterally or ventrally (Figure 4). A 25- to 22-gauge needle is appropriate for most fish, with the length of the needle varying according to the species.
Blood is generally transferred to lithium heparin tubes (i.e., green-top blood tubes) after collection. Blood smears should be made immediately to prevent cell degradation. Additional whole blood can be submitted for a CBC and centrifuged to separate the plasma for chemistry analysis.

Because of wide variation in cell morphology among fish species, a laboratory with experience in fish hematology should be used. These laboratories are accustomed to small sample volumes from patients that may weigh as little as 50 to 100 g.

Hematology and plasma chemistry abnormalities are common in ill fish. Changes in the CBC can indicate underlying infection, erythrocyte abnormalities, and intracellular leukocytic or erythrocytic pathogens. Leukocytosis, anemia, hypoproteinemia, hypoglycemia, and elevated levels of aspartate and alanine transaminase are common in ill fish. However, many ill fish can have normal CBCs and serum or plasma chemistry results.

Microbiology and Cytology
External lesions can be swabbed for microbiologic evaluation. Before sampling, contaminants can be rinsed from affected areas using sterile saline or water. Cytologic preparations of lesions should be evaluated with Gram’s stain, acid-fast stain, and a standard in-clinic cytology stain, such as eosin and hematoxylin. Gram-negative bacteria, fungi, and acid-fast organisms are common pathogens in fish. Standard culture swabs can be submitted for aerobic and fungal culture, whereas specialized sample media may be needed to grow mycobacteria. As with hematology, a laboratory with aquatic organism expertise should be used for piscine microbiology. Growth of microorganisms from fish may be enhanced using temperatures and salinity levels that are markedly different than those used for domestic mammal samples. Culture results must be interpreted cautiously with the knowledge that contamination of skin by environmental organisms is common, suggesting that a positive culture result and an isolated organism may not always be the primary cause of illness.

Fecal Analysis and Culture
Fecal samples can be collected from the bottom of the transport container using a syringe or can sometimes be expressed with gentle abdominal palpation. Direct and indirect (flotation) fecal analysis can be conducted on these specimens. Fish can be infected with numerous metazoan and protozoan parasites, including trematodes, nematodes, cestodes, acanthocephalans, *Spironucleus*, *Hexamita*, and *Coccidia* spp. An eosin and hematoxylin stain, a Gram’s stain, and/or an acid-fast stain can be used to evaluate gastrointestinal flora and determine whether a patient is shedding acid-fast bacteria. With the use of this technique, several coccidian species and flagellated protozoans have also been identified in teleost fish and elasmobranchs.

Samples may also be obtained from anal swabbing or lavage. Lavage samples should be collected by rinsing the vent area with sterile saline or water, inserting a sterile French catheter into the anal opening, and injecting and aspirating sterile saline or water. These samples can be prepared for cytology, direct smears, special staining, and microbiology. In certain species, such as sharks, skates, and rays, coelomic lavage can help determine the...
presence of coelomic coccidia. Coelomic lavage requires a sterile needle, a syringe, and saline. Ultrasound guidance should be used to avoid accidentally injecting the liver or perforating the alimentary tract. Some elasmobranch species have anal pores ventral to the cloaca that may allow passage of coccidia directly into the coelom, resulting in a coelomic coccidian infection, but more research is needed to confirm this.\(^b\)

**DIAGNOSTIC IMAGING**

Diagnostic imaging (i.e., radiography, contrast radiography, ultrasonography, magnetic resonance imaging, nuclear scintigraphy, computed tomography) is readily available and can be conducted in piscine patients. With the advent of spiral computed tomography, fish can be anesthetized and scanned out of water within minutes. At the NEAQ, ultrasonography is frequently used in fish to monitor heart rate, guide needle aspiration, evaluate abdominal distention, and determine egg maturation in the ovaries. Ultrasonography is commonly conducted with the patient in water and lightly sedated or manually restrained. The probe is protected from water using a plastic rectal sleeve. The water interface mimics ultrasonic gel and minimizes artifacts created by the uneven surface on the scaly exterior. Radiography is commonly used for most animals during routine physical examination. In general, dorsoventral and lateral views are obtained for each patient. Radiograph cassettes can be placed in plastic bags for protection in water. To obtain the best images, clinicians should anesthetize patients and remove them from the water; however, in rare cases, patients can be anesthetized in plastic bins or bags, and radiography can be performed through water. Contrast radiography has proven to be a valuable tool in diagnosing some gastrointestinal neoplasms.

**ENDOSCOPY AND EXPLORATORY SURGERY**

When the diagnostic modalities that have already been discussed do not provide a clear diagnosis, endoscopy and exploratory surgery can be used. Endoscopy, colonoscopy, and gastroscopy can easily be performed in anesthetized patients and can provide valuable information and diagnostic samples with minimal invasiveness. General surgery may be required as a final exploratory\(^b\) option and can be readily performed when piscine anatomy and physiology are understood and basic surgical techniques are applied.

**REFERENCES**


---

\(^b\)Whitaker B: Personal communication, National Aquarium, Baltimore, MD, 2001.
3. Water-quality assessment for fish includes
   a. pH.
   b. temperature.
   c. heavy metal content.
   d. alkalinity.
   e. all of the above

4. ________ is common in fish infected with an external parasite.
   a. Splashing
   b. Scratching
   c. Itching

5. Fish should be removed from water for examination for no more than _______ seconds.
   a. 10 to 20
   b. 30 to 60
   c. 30 to 90
   d. 60 to 90
   e. 90 to 120

6. ________ is not a sign of respiratory difficulty in fish.
   a. A gilling rate greater than 60/min
   b. Opercular flaring
   c. Piping at the surface
   d. Symmetric opercular movement
   e. A gilling rate less than 10/min

7. ________ is/are generally normal in fish.
   a. Gas bubbles within the eye
   b. Moderate exophthalmia
   c. Fixed pupils
   d. Cataracts
   e. none of the above

8. The most common method of obtaining a blood sample from a fish involves
   a. the caudal vein.
   b. cardiac puncture.
   c. the segmental fin arteries.
   d. the branchial vein.
   e. none of the above

9. __________ in fish is equivalent to a lung biopsy in mammals.
   a. A skin scrape
   b. A fin clip
   c. A scale sample
   d. An otolith freeze-fracture
   e. A gill clip

10. Ultrasonography is commonly used to detect ________ in fish.
    a. metabolic disease
    b. reproductive abnormalities
    c. skeletal abnormalities
    d. vascular anomalies
    e. all of the above