Anesthesia, Diagnostic Imaging, and Surgery of Fish

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Abstract: *Anesthesia, diagnostic imaging, and surgery of fish have become routine parts of aquatic animal medicine. Anesthesia may be required for simple clinical procedures, diagnostic testing, or more involved surgery. Diagnostic modalities, including radiology, ultrasonography, and endoscopy, can be readily applied to fish and may provide valuable information. Despite some unique challenges, surgery can be performed in fish using basic surgical skills and principles and should be considered as a valid treatment option.*

At a Glance

Although unique and challenging in some respects, anesthesia induction, imaging, and surgery can be performed in fish with standard equipment that is readily available. In addition, the practitioner can take advantage of many of the same diagnostic modalities and drugs that are used in mammals. Therefore, clinicians with the requisite knowledge can offer many of these services with only a modest investment.

Anesthesia

Many substances have been used to anesthetize fish, including ethanol, diethyl ether, halothane, lidocaine, tricaine methanesulfonate (MS-222), eugenol, ketamine, metomidate, propofol, and carbon dioxide.[1–8] MS-222 is the anesthetic agent most used in fish and the only one approved by the US Food and Drug Administration for use in food fish (with a withdrawal time of 21 days).[9] It is a slightly acidic water-soluble powder. When it is used in an anesthetic bath, sodium bicarbonate can be added at a ratio of 1 mg MS-222 to 1 mg sodium bicarbonate by weight to minimize the decrease in pH of the water.[6] However, sodium bicarbonate is commonly used with MS-222 only for freshwater fish, as saltwater fish benefit from the buffering capacity of sea water and saltwater mixes. Higher ratios of sodium bicarbonate are thought to decrease the amount of MS-222 required for anesthesia, but this has not proved

necessary with low-dose MS-222 in our experience.

MS-222 dose requirements vary among fish species, but most teleost fish can be anesthetized at 60 to 150 mg/L, and the dosage does not seem dependent on fish size or age. The dose can be adjusted incrementally as needed to achieve optimal anesthetic depth. Fish such as sharks, skates, rays, and seahorses may be more sensitive to MS-222, and a lower initial dose should be considered (e.g., 50 to 70 mg/L). Starting with lower doses of MS-222 should also be considered for debilitated patients.

Delivery

Depending on the specific drug and circumstances, anesthetic agents may be administered by intramuscular, subcutaneous, or intravenous injection; by mouth; or by immersion (bath). For short procedures, anesthesia is generally achieved by immersing the fish in an MS-222 bath. For longer procedures, continuous delivery can be maintained by pumping anesthetic-containing water over the gills.[1,4] In most cases, this is achieved by manually injecting the water into the mouth using an appropriately sized syringe. MS-222 has been classified as a human retinotoxin, so gloves and goggles should be worn when contact is unavoidable.[10]

Fish surgical tables can be designed with recirculating pump systems to deliver the anesthetic agent and oxygen and

remove carbon dioxide while maintaining surgical access.[7] The system shown in **FIGURES 1 AND 2** was made from a commercial rat cage, with the water and water pump in the bottom and the top serving as the surgery table. The slots in the top allow the water pumped through the fish's mouth to return to the tank after passing through the gill slits. This particular rat cage was chosen because it can be sterilized in an autoclave between patients. Regardless of the system, an aerator stone should be used to oxygenate the water during surgery, maintaining normal arterial blood oxygen levels and exchanging carbon dioxide across the gills. Oxygen levels can be monitored by a meter in the anesthetic bath water. Oxygen can be "bubbled" into the anesthetic bath during long procedures, but a low flow is essential to avoid supersaturation and gas bubble disease.

Stages and Signs

Various indices have been published for the stages of anesthesia in fish.[8] When a fish is placed in an MS-222 bath, it usually exhibits an excitatory phase with increased opercular movements. Within several minutes, however, the fish decreases purposeful movements, becomes ataxic, swims irregularly, and starts turning on its side. Eventually, it turns to dorsal recumbency and sinks to the bottom of the container. The respiratory rate, as identified by opercular movement, slows in a dose-dependent manner. Presumably, ventilation is decreased as the oper-

cular and oral movements decrease. If the fish is deeply anesthetized, opercular movements will cease completely, and assisted ventilation should be instituted. Assisted ventilation may be performed manually by moving the fish forward in the water (never backward) or mechanically with a pump circulating water at a rate sufficient to maintain normal levels of arterial oxygen and carbon dioxide over the gills in a cranial-to-caudal direction. With assisted ventilation, care should be taken to gently keep the mouth open and not hold the animal in a manner that prevents the gill flaps or operculum from opening.

Monitoring

Heart rate may be monitored by ultrasonography, electrocardiography (ECG), Doppler probe, or pulse oximetry. Ultrasonography can provide a clear image of the heart in most fish to assess rate and contractility. A Doppler flow probe can be placed in one of three positions: on the isthmus (region on the ventral surface of the head between and slightly caudal to the opercula); inside the operculum between the caudal aspect of the gills and the body wall, aiming toward the heart; or on the dorsal surface of the tongue (if the mouth is large enough). Pulse oximetry in fish uses an esophageal, rectal, finger, or tongue probe, but the saturation readings may be inconsistent. The use of peripheral oxygen saturation data in fish is still experimental, but pulse oximetry shows the heart rate, and we have found it useful for anesthetic monitoring compared with ultrasonography in several


FIGURE 1
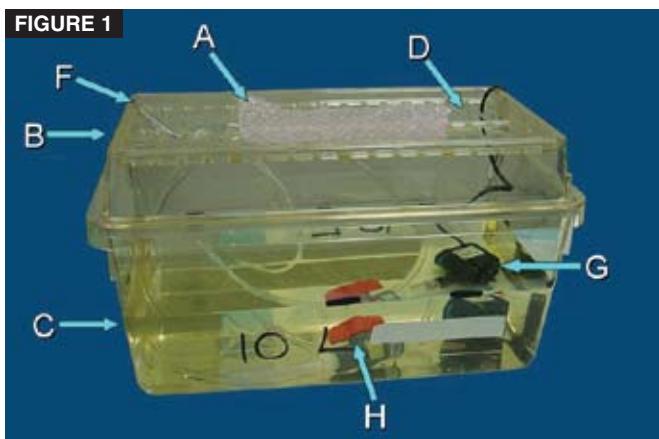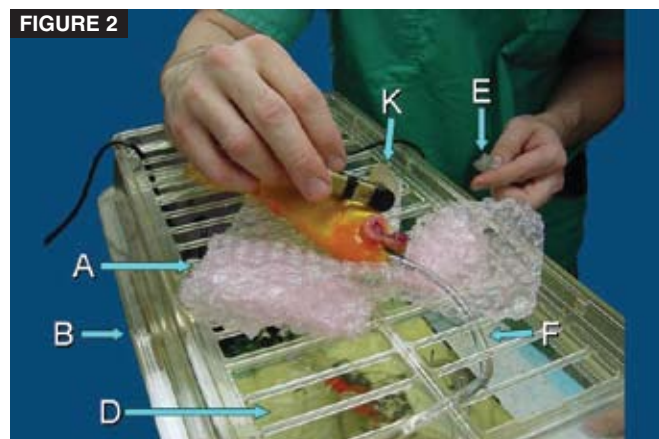
Photograph by Alan M. Klide

A small fish anesthetic unit modified from a rat cage. (A) bubble wrap, **(B)** rat cage cover, **(C)** rat cage, **(D)** slots in cage cover, **(F)** vinyl tubing to fish, **(G)** pump, **(H)** valve in hose.

FIGURE 2

Photograph by Alan M. Klide

Use of a device to anesthetize a common goldfish (*Carassius auratus*) with anesthetic monitoring using a pulse oximeter and Doppler blood flow unit. **(A)** bubble wrap, **(B)** rat cage cover, **(D)** slots in cage cover, **(E)** Doppler probe, **(F)** vinyl tubing to fish, **(K)** pulse oximeter probe on fin.

FIGURE 3
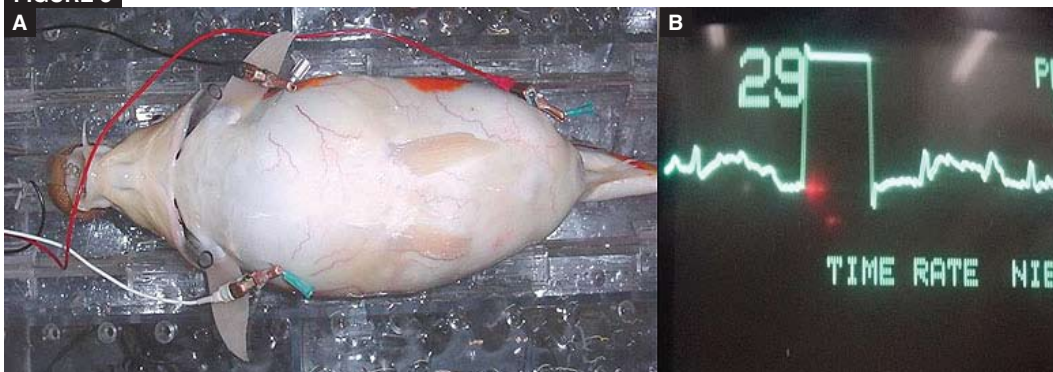
Electrocardiography (ECG) probe placement using the pectoral fins of a koi (*Cyprinus carpio*) (**A**) and electrocardiographic tracing (**B**) from a koi under anesthesia. The ECG tracing is displayed using a 1-mV standard.

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species. The rectal pulse oximetry probe can be held on the dorsal surface of the tongue of many fish species. The tongue probe works on the fins in some fish, but this varies among fins and fish (**FIGURE 2**). An ECG monitor can be connected to hypodermic needles placed subcutaneously, with one needle on each side between the branchiostegal membrane and the pectoral fins, and one needle approximately 25 mm caudal to the anal fin (depending on fish size; **FIGURE 3**). ECG may be inconsistent in some fish, and in some cases, the electrical activity of the heart may not be reflected accurately in the ECG trace.

Recovery

Fish should be allowed to revive in anesthetic-free water until strong opercular movements are observed, then put into a recovery tank when they can swim and maintain proper buoyancy. If the fish does not maintain strong opercular movements and forward swimming motion, water should be pumped into the mouth or the fish manually moved forward until it can swim and ventilate normally. The water should have an aerator stone in it for aeration using a standard aquarium air pump that uses room air. Adding oxygen directly to the tank can cause sedation of fish or result in the pathologic formation of gas bubbles in the branchial or ocular capillaries.

Analgesia

Several reviews of nociception in fish have been recently published,[11,12] and there are some research data about the use of opioids in fish.[13–15] Dosing is complicated by the fact that the temperature and pH of fish are different from those of warm-blooded animals (i.e., the

temperature is lower, and the pH is higher). This may influence the chemical configuration of some analgesics and, hence, their availability and dose.[16] While there are few clinical data on the use of analgesics in fish, most fish practitioners assume that fish sense pain and use analgesics accordingly. Clinically, we have used both opiates and NSAIDs in fish, but no definitive trends have been noted. Drugs such as carprofen, meloxicam, ketoprofen, flunixin meglumine, butorphanol, morphine, and buprenorphine may be used, but the dose and dosing interval are at the discretion of the clinician. At New England Aquarium, flunixin meglumine is routinely used in teleost fish at a dose of 0.25 to 0.5 mg/kg IM every 3 to 5 days. Butorphanol at 0.1 to 0.4 mg/kg IM as a single dose just prior to recovery from anesthesia has produced no obvious adverse effects, but antinociception has not been demonstrated.[17]

*Quick**Notes***

Safe and effective anesthesia of fish is easily performed to permit both brief diagnostic procedures and long, complex surgical procedures.
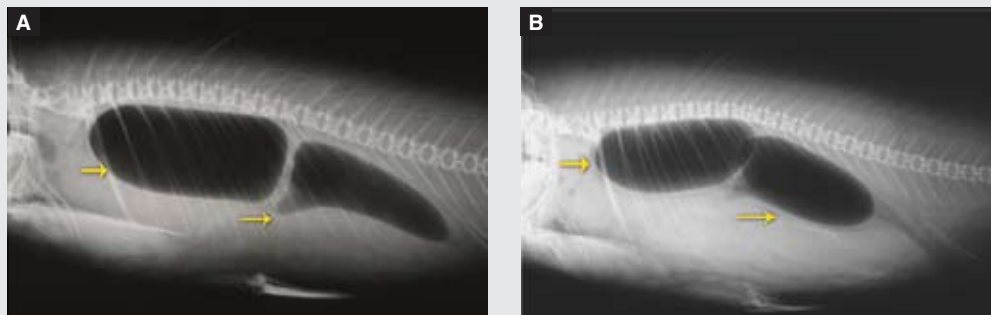
FIGURE 4

Courtesy of New England Aquarium

Oranda goldfish (*C. auratus*) being positioned in left lateral recumbency on a radiographic film cassette.

FIGURE 5

Radiographic Projections

A

B

Courtesy of New England Aquarium

Dorsoventral whole body radiographic projections of koi (*C. carpio*) with a normal swimbladder (A) and a deviated swimbladder caused by an old spinal fracture (B). The swimbladders have two compartments (*arrows*).

Diagnostic Imaging and Endoscopy

Fish were among the first species to be evaluated by radiography.[18] However, the use of radiography and computed tomography (CT) for diagnosis in fish is relatively recent.[7,17,19–32] Radiography is easily performed while the fish is sedated in lateral recumbency (**FIGURE 4**). The ability to obtain a dorsoventral view depends on morphology. Suspected skeletal and coelomic abnormalities are the most common indications for radiography. Most fish lack sufficient intracoelomic fat to provide radiographic detail of the internal soft tissue organs. However, the swimbladder is generally obvious and can be evaluated for abnormal shape, compression, or deviation[7,19–21] (**FIGURE 5**). Vertebral deviations and malformations of the spinal cord are common in fish[22,28,29] (**FIGURE 6**). Iodinated or barium sulfate contrast media can be used for gastrointestinal (GI) contrast studies and to evaluate the origin of coelomic masses[24,30] (**FIGURE 7**). Contrast medium is generally administered orally to the sedated fish via gavage tube. To avoid repeated sedation, the contrast agent can be infused into the stomach and the colon simultaneously; this produces a nearly complete GI contrast study within several minutes in most carnivorous and omnivorous species with short GI tracts.

Helical CT is an excellent way to investigate skeletal and soft tissue abnormalities in fish and has the advantage of rapid acquisition of full-body images. Compared with conventional radiography, CT can provide more detail for assessing skeletal malformations and soft tissue masses in fish (**FIGURE 8**).

Diagnostic ultrasonography has been used in larger pet fish and in aquaculture.[33–41] Clinically, ultrasonography is most useful for evaluation of ascites, soft tissue masses, reproductive organs, and liver size and echogenicity (**FIGURE 9**). Other imaging modalities such as fluoroscopy and nuclear scintigraphy have also been used in fish, with some success.[23]

Endoscopy may be a useful diagnostic technique for evaluating fish.[42] Rigid and flexible endoscopes from 1.5 to 3 mm in optical diameter can be used for GI endoscopy, examina-

*Quick**Notes***

Radiography, ultrasonography, and endoscopy are useful for diagnosing medical disorders in fish.

FIGURE 6

Courtesy of New England Aquarium

Left lateral whole body radiographic projection of a juvenile sand tiger shark (*Carcharias taurus*) with scoliosis and kyphosis.

FIGURE 7

Courtesy of New England Aquarium

A left lateral radiographic projection of a largemouth bass (*Micropterus salmoides***) given 10 mL of barium sulfate suspension 60% for a gastrointestinal positive-contrast study.**
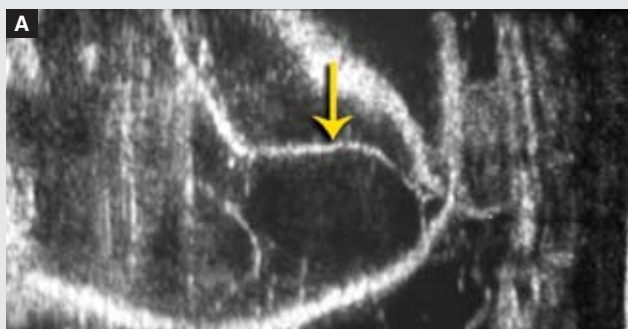

FIGURE 8

Courtesy of New England Aquarium

Computed tomography image of a largemouth bass (*M. salmoides***) in lateral recumbency after oral administration of 2 mL of water-soluble organic iodine iohexol (300 mg/mL).**

tion of the gills, and laparoscopy (**FIGURE 10**). Biopsy channels and surgical sheaths in these endoscopes allow for the introduction of specialized biopsy and surgical instruments. For gastroscopy and colonoscopy, visualization may be improved by infusing sterile 0.9% saline solution through the endoscope sheath or biopsy channel, similar to the technique used for cystoscopy in mammals. The saline helps dilate the lumen of the GI tract and clear away debris. For colonoscopy, the endoscope is introduced into the cloaca with the aid of continuous saline infusion. The endoscopist must differentiate the rectal orifice from the urethral and oviduct orifices. The amount of saline infused into the cloaca should be monitored carefully to avoid excessive pressure. Although GI mucosal lesions are relatively rare in fish, endoscopic examination and biopsy provide a simple method of excluding them.

Laparoscopy may be used in fish to evaluate reproductive activity and perform organ biopsies and endosurgical procedures.[42,43] At the New England Aquarium, laparoscopy has been used to obtain liver biopsy samples and examine the reproductive tract. Typically, a ventral paramedian incision is made 1 to 3 cm anterior to the anus, depending on the species. The incision need only be large enough to allow introduction of the endoscope (generally about 1 cm). Insufflation of the coelom can be achieved by infusing a 0.9% saline solution or carbon dioxide through the endoscope sheath. A precision digital insufflator must be used with carbon dioxide to ensure that intracoelomic pressure does not become excessive. In most cases, a pressure of 3 to 4 mm Hg suffices. Minimally invasive endosurgical techniques (e.g., sterilization) are being developed for fish and will undoubtedly help to reduce

FIGURE 9

Ultrasonographic Images

A

B

Courtesy of New England Aquarium

Ultrasonographic images of a cystic ovary (*arrows***) in a koi (***C. carpio***) in the caudal coelom 2 cm cranial to the anus (A) and in the cranial coelom 2 cm distal to the pectoral fins (B).**
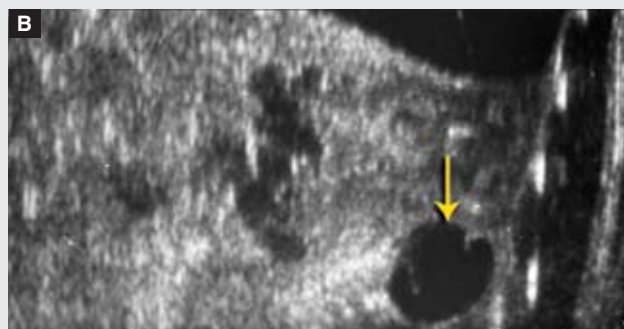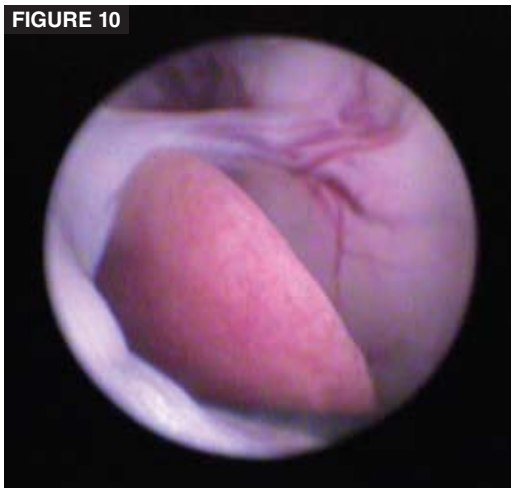
FIGURE 10

Endoscopic view of normal hepatic parenchyma in a brook trout (*Salvelinus fontinalis*).

QuickNotes

Many surgical procedures can be performed in fish using standard surgical skills and instruments.

patient stress, pain, and healing time compared with traditional open laparotomy.[43] Laparoscopy incisions can usually be closed with one or two simple interrupted nylon skin sutures.
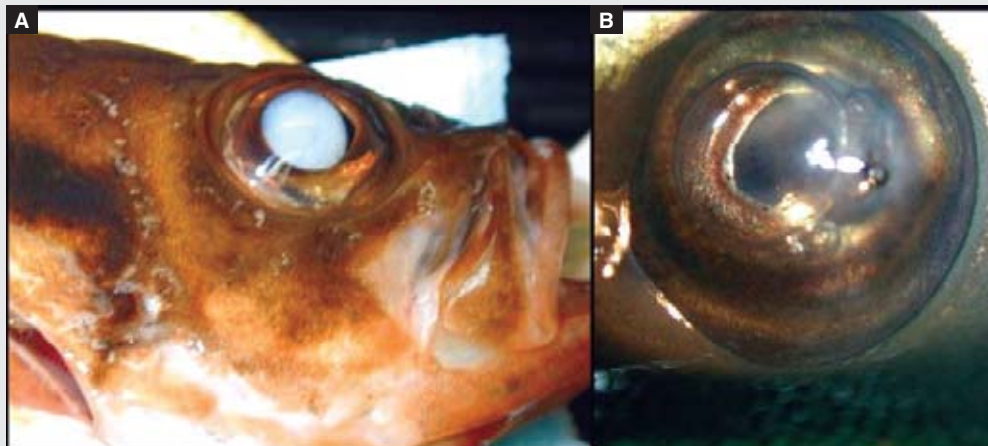
Surgery

Common indications for surgery include neoplasia; ocular conditions; swimbladder disorders; biopsy of the liver, kidneys, or spleen; reproductive problems; and foreign body ingestion. In public aquaria, surgery may be required for both cosmetic and medical conditions. For example, fish can become unexhibitable due to dermal or ocular lesions that can be corrected surgically[7,17,19–21,25,42–44] (**FIGURE 11**).

Fish surgery requires an intimate knowledge of comparative anatomy and presents some unique challenges. The presence of running water makes it difficult to maintain a clean, dry, sterile surgical field and raises safety concerns in the presence of electrical equipment. Fish depend on mucus to protect their epidermal and dermal skin layers; therefore, the skin must be kept moist during long procedures. The presence of large scales and a typically immobile skin surface can further complicate certain procedures. Some species possess venom glands located near fin rays or barbs. Also, many species of fish have no eyelids, so the eyes can be easily damaged during surgery. The eyes can be protected using a sterile eye lubricant, which may have to be applied frequently during the procedure. On some occasions, sterile gauze can be loosely placed over a 1-mm layer of eye lubricant to afford greater protection if the procedure involves manipulation or will last longer than 45 minutes.

Perioperative antibiotics have historically been recommended for clean-contaminated (or "dirtier") procedures in mammals or when infection would be catastrophic. In our practices, all fish undergoing intracoelomic procedures receive perioperative antibiotics, as well as a postoperative course lasting 1 to 4 weeks (e.g., ceftazidime 30 mg/kg IM q72h). Fish undergoing uncomplicated external surgery usually receive only perioperative antibiotics or none at all.

FIGURE 11

Severe Cataract

A severe cataract in a red fish (*Sebastes marinus*) **(A)** and image of the same right eye 6 weeks postsurgery after manual cataract removal with fine forceps **(B)**.
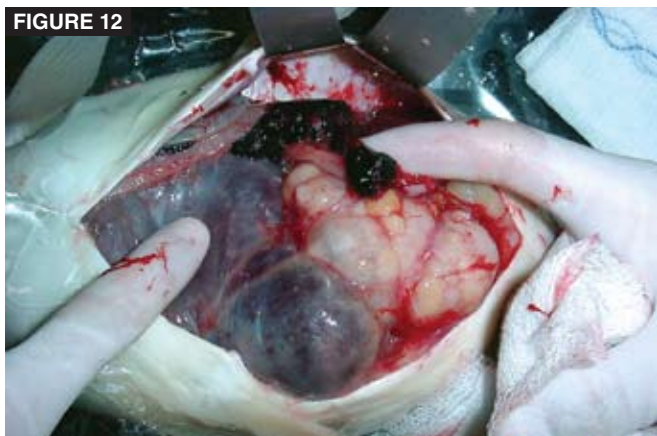
FIGURE 12

Photograph by Chick Weisse

Gross view of a koi (*C. carpio*) with a right finger pointing to an interstitial cell ovarian tumor and demonstrating an extremely thin body wall.

FIGURE 13

Photograph by Chick Weisse

Soft tissue reaction and expulsion of suture material (monofilament nylon) 14 days postsurgery in a black drum (*Pogonias cromis*).

Sterile preparation of the surgical field is typically omitted in fish to preserve the skin and its protective mucus. Wiping the area with a sterile saline-soaked cotton swab or dilute povidone–iodine or chlorhexidine can remove gross contamination, although at least one study found no difference in wound healing among trout given presurgical and postsurgical topical povidone–iodine antiseptic preparation versus those that received none.[45] For marine species, rinsing the surgical site with sterile fresh water may help to reduce surface bacterial numbers. If very large scales are present, removal of a row of scales will facilitate both incision and closure. This is accomplished by applying gentle caudal tension on each scale with a hemostat. If performed with care, the scales will regrow quickly.

We have found large, clear, transparent plastic drapes with no adhesive or precut apertures to be the most useful for isolating the surgical field. These drapes adhere well to the wet skin of the fish and help prevent desiccation. A small bead of lubricating jelly can be applied to the surface of the fish to aid in securing the drape. The small size and typically scaly skin surface of patients preclude the routine use of towel clamps, although they may help in certain cases.

Different surgical procedures require different surgical instruments, but we regularly use surgical loupes, vascular clips, and microsurgical, bipolar electrocautery, and suction devices. A number of surgical procedures have been described.[7,17,19–21,25,43,44] Coelomic incisions are preferably closed in two layers when possible. However, large abdominal tumors leading to massive coelomic swelling, diminished appetite, and muscle loss can result in thinning of the body wall, and only a single-layer closure may be possible (**FIGURE 12**). Evacuation of excessive coelomic air is recommended following tumor removal to aid in recovering buoyancy.

The choice of suture material depends on the type of procedure, anatomic location, patient size and type, tension, healing processes, surgical performance and expectations, and surgeon preference. Most surgeons avoid multifilament sutures because of their wicking potential when submerged in water containing bacteria. It is unclear whether absorbable sutures are degraded and phagocytized in fish or simply expelled as foreign bodies, but some (e.g., polyglyconate) have been confirmed to undergo degradation by hydrolysis.[46] One study showed that synthetic suture materials are generally better tolerated than organic sutures, concluding that monofilament polyglyconate produced the mildest inflammatory reactions at 7 and 14 days.[46] This study also reflected our experience that monofilament nylon (typically a nonreactive suture material in mammals) causes a substantial inflammatory reaction[46] (**FIGURE 13**). Therefore, synthetic, absorbable, monofilament suture material is recommended. Cyanoacrylate has been shown to delay healing and result in surgical complications, although one study reported complete, uncomplicated wound healing within 14 days when it was

used in channel catfish.[47] We have observed tissue reactions to cyanoacrylate in some species of marine fish. The choice of suture pattern has not been demonstrated to alter surgical outcome, but continuous patterns are often preferred to facilitate rapid closure and improve resistance to fluid leakage into the coelomic cavity. We prefer an appositional pattern to an inverting or everting closure (**FIGURE 14**).

Suture removal is performed without anesthesia if possible, or under light sedation for intractable fish. Skin healing has been reported to occur in various species in 8 to 26 days.[45,47,48] We remove sutures 2 to 3 weeks postoperatively in temperate and tropical species. For species with very low metabolic rates (i.e., those from cold marine environments), suture removal is often delayed for 6 to 10 weeks. **C**

FIGURE 14

Ford interlocking suture pattern used for appositional skin closure in a goldfish (*C. auratus*) following coelomic surgery.

Photograph by Chick Weisse

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3 CE CREDITS

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1. The most commonly used FDA-approved anesthetic agent for fish is
   a. eugenol.
   b. clove oil.
   c. lidocaine.
   d. tricaine methanesulfonate (MS-222).

2. Most anesthetics are administered to fish
   a. via the mouth.
   b. via injection.
   c. via immersion (bath treatment).
   d. via gas inhalation.

3. The respiratory rate in fish is determined by counting _____ movements.
   a. thoracic
   b. opercular
   c. lip
   d. fin

4. To decrease nociception in fish, the surgeon can use
   a. butorphanol.
   b. flunixin meglumine.
   c. morphine.
   d. all of the above

5. One of the most common indications for radiography in fish is
   a. visual impairment.
   b. neurologic signs.
   c. coelomic abnormalities.
   d. respiratory deficiency.

6. Which imaging technique has been used to evaluate abnormalities in fish?
   a. endoscopy
   b. contrast radiography
   c. helical computed tomography
   d. all of the above

7. _____ is/are not a common indication for surgery in fish.
   a. Vestibular disease
   b. Reproductive issues
   c. Neoplasia
   d. Swimbladder problems

8. _____ presents a challenge when performing surgical procedures in fish.
   a. Protecting the eyes
   b. Oral damage
   c. Loss of scales
   d. Compromise of the dorsal fin

9. Which solution is recommended for surgical preparation in fish?
   a. 4% chlorhexidine gluconate surgical scrub
   b. 7.0% isopropyl alcohol scrub
   c. sterile water or 0.9% saline solution
   d. 7.5% povidone–iodine solution

10. The best suture material in fish, as reported from research studies, is
   a. monofilament gut.
   b. monofilament nylon.
   c. monofilament polyglyconate.
   d. stainless steel (staples).