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The Effects of Biotelemetry Transmitter Presence and Attachment Procedures on Fish Physiology and Behavior

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ABSTRACT: Biotelemetry — the process of conveying data from a transmitter-attached animal to a data collection site — has received increasing awareness from fisheries researchers. Prior to biotelemetry data collection, it is imperative that researchers are aware of and understand the possible effects that transmitter presence and attachment procedures may have on 'normal' fish behavior and physiology. To allow successful transmitter attachment, numerous methods to anesthetize the study fish, with varied impact and effectiveness, may be employed. Following anesthetization, three standard methods of transmitter attachment have been developed — external attachment, intragastric insertion, and surgical implantation. Although each method has advantages and disadvantages, their success largely depends on factors such as the species, environment, fish and transmitter size, and duration of the telemetry study. Additionally, each method of attachment can affect experimental fish physiology and behavior in varying ways. After describing each of the transmitter attachment procedures, we review the effects of transmitter presence and attachment procedure on fish physiology and behavior, with special focus on implications to aquaculture and fisheries related studies.

KEY WORDS: biotelemetry, transmitter effects, transmitter attachment, surgical procedure, anesthesia, physiology, behavior.

I. INTRODUCTION

Telemetry is the process of conveying information from one location to another in the absence of hard-wired cable links (Geers *et al.*, 1997). Since the first use of telemetry devices to study the biology of fish (Trefethen, 1956; Johnson, 1960), the science of "biotelemetry" has become one of the most widely accepted and evolving disciplines used to study the behavior and physiology of fish in their natural environment. Its continued acceptance has resulted from advances in technology that have allowed transmitters to become smaller and lighter and incorporate GPS and sensor peripherals for measuring numerous environmental parameters (including light, depth and temperature; Voegeli and McKinnon, 1995). New generation telemetry devices will allow for the study of a range of animals from both aquatic and terrestrial environments and over a range of size and age classes.

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Fisheries biologists frequently rely on mark and recapture studies, recreational fishery surveys, and commercial fishery logs to determine fish movement and habitat (Nielsen and Johnson, 1983). Conventional methods, however, only allow discrete fish movement patterns and are limited with respect to the quantity and quality of data collected. Aquaculture researchers may employ direct observation (Sutterlin *et al.*, 1979), underwater camera systems (Furevik *et al.*, 1993; Kadri *et al.*, 1991), or hydroacoustics (Juell *et al.*, 1994) to monitor cultured fish behavior within an aquaculture cage. Although these techniques have merit to provide basic answers regarding behavioral questions, fish are monitored as a unit, frequently in small observation volumes with respect to the total cage volume, and frequently lack 24-h diel data sets. As a potential alternative, biotelemetry technology and methodology may be employed to monitor fish movement and behavior in the natural environment or aquaculture cages and eliminates shortcomings associated with conventional monitoring techniques. Biotelemetry provides numerous advantages over conventional fisheries research methods including continuous physiological and behavioral monitoring, from a passive perspective, following a single intrusive event that may disrupt normal behavior for transmitter attachment. Fisheries telemetry systems may vary immensely with regard to the equipment used and complexity depending on the aquatic environment and scope of the study. Typically, during manual tracking or fixed datalogging, a radio or acoustic transmitter is attached to the study animal and emits the appropriate radio or acoustic signal that is received by either an antenna or hydrophone (Winter, 1996). Transmitters may transmit through a unique frequency for identification or several transmitters programmed to use the same frequency to transmit a unique identification code, which might be accompanied by some other measured parameter of the environment or physiology of the experimental animal.

The key for any successful telemetry study is the ability of the researcher to attach the transmitter to the experimental fish without negatively affecting normal fish physiology or behavior, or causing mortality, post-attachment. This is critical in studies where the fish is immediately released after attachment, without an observation period to ensure proper recovery from both the anesthetic and attachment procedure. In the absence of an observed recovery period, it is also important for the researcher to have insight on the expected duration of recovery to ensure all collected data are indicative of the natural behavior of the fish.

For these reasons, attachment procedures and subsequent rate and degree of physiological/behavioral recovery influence the success of telemetry research. Without acknowledging potential telemetry limitations, researchers may make conclusions based on erroneous data resulting in poor fisheries and aquaculture management plans. The purpose of this article is to review transmitter attachment methodologies that have been used in biotelemetry studies, and provide awareness to telemetry users of the short- and long-term effects those attachment procedures and transmitter presence may have on experimental fish. Although this review focuses on the effects of transmitter attachment and effects, we begin with a brief overview of possible methods to sedate or anesthetize fish to illustrate the importance of this step in transmitter attachment procedures (for a more complete review of anesthetic use see Ross and Ross, 1999).

II. ANESTHESIA

Crowding and excessive handling may result in elevated levels of stress in fish (Wedemeyer, 1976; Strange *et al.*, 1977; Schreck, 1981), particularly in aquaculture settings (Barton and Iwama, 1991), that may become lethal. Prior to attaching a transmitter to the experimental animal, the fish has to be anesthetized, decreasing stress levels and muscle movement, both voluntary and involuntary. Appropriate anesthetics must allow for rapid induction and recovery times (within 3 and 5 min, respectively, for fish anesthetics, Marking and Meyer (1985)) and not cause extreme changes in the physiological state of the fish that would reduce the chance of survival and modify normal behavior. In addition, the anesthetized state must exist for sufficient time to allow transmitter attachment.

A. PHYSICAL SEDATION

Physical sedation of fish includes lowering the water temperature to create a condition of artificial, or seasonal, torpor or through the use of electric shock. Artificial torpor might not produce the desired degree of sedation to allow surgical attachment of a transmitter in all species, but should be sufficient for intragastric placement (Hovda and Linley (2000) used hypothermia to spawn Pacific salmon). However, some species, such as cod, will experience a natural seasonal torpor that may provide sufficient sedation to accomplish either of the transmitter attachment techniques (Arnold *et al.*, 1994 [external attachment]; Paul D. Winger, Memorial University of Newfoundland, unpublished data [intragastric insertion]; Wroblewski *et al.*, 1994 [surgical implantation]). Although electric shock may have the advantages of quick sedation with no chemical requirement (Jennings and Looney, 1998), fish may require a substantial period of time for complete recovery from physiological disturbances associated with this form of anesthesia (Madden and Houston, 1976). Mortality rates and physiological variations associated with electroanesthesia are comparable to those measured from chemical anesthesia (Madden and Houston, 1976; Schreck *et al.*, 1976). However, electrofishing injuries may also include spinal damage (Sharber and Carothers, 1988) and hemorrhaging (Schill and Elle, 2000). Induced injuries may have further consequences on survival, long-term growth (Dalbey *et al.*, 1996), and behavior (Mesa and Schreck, 1989). Caution should be exercised to ensure proper voltage and charges are used to avoid major physiological trauma and mortality (Madden and Houston, 1976). This is particularly important for use in high conductivity water, with little control of the supplied voltage (Gunstrom and Bethers, 1985).

B. CHEMICAL ANESTHESIA

Chemical anesthesia is administered in the aqueous environment with the fish. The anesthetic is absorbed through the gill epithelia and enters the vascular system at the gills. Factors affecting anesthetic absorption and excretion include gill epithelia surface to body volume ratio, thickness of epithelium, type of anesthetic, dosage, and water temperature (Stoskopf, 1993). Suitable anesthetics result in unconsciousness, pain inhibition, and loss of involuntary muscular reaction. However, care should be taken to avoid consequences of hypoxia and acid-base disturbances associated with the use of chemical anesthesia (Stoskopf, 1993). Chemical anesthetics may also be frequently used in a maintenance dose that is less concentrated while the transmitter attachment procedure is carried out.

The most widely used fish anesthetic is 3-aminobenzoic acid-ethyl-ester-methanesulfate, or MS222 (Marking and Meyer, 1985). The recommended dosage of 50 to 100 mg/l MS222 has been used successfully for anesthetizing numerous fish species in varied environments and temperatures. However, MS222 is acidic and therefore should be buffered prior to use in fresh water. Furthermore, for some species, the effective and toxic doses of MS222 are similar, particularly in warm water. Thus careful monitoring is required throughout the procedure (Gilderhus and Marking, 1987). Ethyl-*p*-aminobenzoate (Benzocaine) is also a commonly used anaesthetic that produces less metabolic changes and agitated swimming often associated with the use of MS222 (Wedemeyer, 1970; Gilderhus, 1990; Gilderhus *et al.*, 1991). 2-methlyquinoline sulphate (Quinaldine sulfate) has been tested for anesthetic use. However, it does not block involuntary muscle reflexes, potentially decreasing its suitability in facilitating transmitter attachment procedures (Gilderhus and Marking, 1987). Schramm and Black (1984) found quinaldine to be a suitable anesthetic for grass carp (*Ctenopharyngodon idella*) in cool water. In warmer water, fish anesthetized with quinaldine recovered normally within two hours post-treatment, but all treated grass carp died 40 hours post-treatment. Possible reasons for such high mortality were increased concentration of the gas phase of quinaldine, diffusion rate, and fish metabolic rate producing more toxic metabolites.

Chemical anesthetics, including those discussed above, are considered potential dangers to both the environment and humans and may require a specific minimum withdrawal period before exposed fish can be released to the wild or processed for human consumption (*e.g.*, MS222 has a U.S. Food and Drug Administration [FDA] minimum 21-day withdrawal period (Marking and Meyer, 1985)). This limits the application of these chemicals within the aquaculture industry and for studies requiring immediate release of experimental fish (*i.e.*, most biotelemetry studies). Therefore, a more appropriate anesthetic that is 'generally regarded as safe' (GRAS) is required. Carbon dioxide (CO_2) can be employed as a GRAS substance to anesthetize fish and allow immediate release or harvest of sampled fish. However, its use is limited by long induction and recovery times (Gilderhus and Marking, 1987) that are not suitable for heavy sedation, and therefore it is limited to light sedation that is appropriate for fish sampling and sizing or intragastric placement of a transmitter (Post, 1979).

Clove oil is a promising candidate as an appropriate fish anesthetic with deep anesthesia, rapid induction, and recovery times, and GRAS classification, allowing aquaculture use or immediate release in both fresh and salt water following invasive field procedures (Soto and Burhanuddin, 1995; Anderson *et al.*, 1997; Keene *et al.,* 1998; Peake, 1998; Prince and Powell, 2000). Clove oil is distilled from the flower, flower stalks, and leaves of clove trees (*Eugenia aromatica*). Eugenol, 4-allyl-2-methoxy-phenol, is the active anesthesizing ingredient, making up 90 to 95% of the oil (Briozzo *et al.*, 1989). It has a germicidal effect on numerous bacterial pathogens (Briozzo *et al.*, 1989) and may prevent the growth of some fungal species (Bullerman *et al.*, 1977). Gaining popularity in its use, clove oil has been reported to have comparable anesthetizing effects to MS222. Anderson *et al.* (1997) compared the efficacy of clove oil with MS222 to anesthetize juvenile and adult rainbow trout (*Oncorhynchus mykiss*). Clove oil was as effective as MS222 at inducing anesthesia in rainbow trout but the recovery times were somewhat longer for clove oil induced anesthesia, although not excessively. In addition, neither clove oil nor MS222 induction caused a change in the swimming performance of the experimental rainbow trout.

III. ATTACHMENT PROCEDURES

Following successful anesthesia, fish may have a transmitter attached either by external mounting, intragastric placement, or surgical implantation in the peritoneal cavity. Although the choice of anesthetic and method and duration of attachment procedure may vary, most procedures follow generic steps for each transmitter attachment method. Regardless of the attachment procedure, it is the desire in biotelemetry studies that neither transmitter presence nor attachment procedure have a negative effect on the survival, growth and behavior of the experimental fish. If such effects occur, the legitimacy of the data collected can be questioned. The researcher must determine which attachment procedure will be most beneficial to the study and subsequent data collection. Attachment considerations should include species being studied, duration of study, size of animal and transmitter, environment, and objectives of the telemetry research. A general "2% rule" has been maintained whereby the transmitter weight should not exceed 2% of the body weight in air of the experimental fish regardless of attachment procedure (Winter, 1996). However, this weight ratio lacked experimental data and has recently been challenged (Brown *et al.*, 1999) using 6 to 12% of the fish body weight and Perry *et al.* (2001) experimented with transmitters representing 3.2 to 9.4% of the fish body weight with no adverse effects. Brown *et al.* (1999) suggests an index should be developed that accounts for the transmitter weight in water and volume to determine the optimal transmitter size for a study; however, they concede that much more research is required to develop a suitable standard index.

A. EXTERNAL ATTACHMENT AND EFFECTS

External attachment procedures are suitable for short-term research in environments that lack high velocities or physical obstructions such as vegetation that can elevate the risk of entanglement. External attachment has the benefit of being quick and easy, requiring minimal handling of the fish, but has greater risk of abrasion and transmitter loss due to snagging. Additionally, transmitters may not be internally attached to some fish species owing to the biological limitations associated with stomach and peritoneal cavity size (*e.g.*, flatfish species; and, in fact, external transmitters have also been used to study squid [Yano *et al.*, 2000] and lobsters [O'Dor *et al.*, 2000]). External transmitter placement has also proven useful in studying gravid females, with surgical implantation after spawning often resulting in higher mortality rate (Winter, 1996). Transmitters are externally attached directly to the body surface either dorsally, laterally above the lateral line, or ventrally, and most often positioned anterior to the dorsal fin. Transmitters may be sutured directly to the body, although it is more common to pass attachment wires through the dorsal musculature of the fish, similar to the method used for attaching carlin tags and illustrated in Wydoski and Emery (1983). Once attachment location and material are determined, individual fish are lightly anesthetized and placed upright on foam support. Two hypodermic needles, large enough to pass restraining wire, are pushed through the dorsal musculature at an appropriate distance to secure the transmitter. The wire is passed through the needles, the needles are removed, and the wire is looped and tied around the front and back of the transmitter, where it is tied together and tightened (Figure 1). An external attachment procedure should require a maximum of 10 min to complete (Thorstad *et al.* (2000) completing the procedure in 3.3 to 5.5 min). A plate may be fixed to the opposing side (Mellas and Haynes, 1985; McKinley *et al.*, 1994) or a pannier arrangement used with an equal load attached to the body surface on the opposite side of the transmitter (Thorpe *et al.*, 1981; Greenstreet and Morgan, 1989). Herke and Moring (1999) described a "soft" harness to externally attach transmitters and protect the dorsal musculature of the experimental fish.

Very few studies have employed external transmitter attachment (e.g., Ross and McCormick, 1981 [yellow perch, *Perca flavescens*]; Greenstreet and Morgan, 1989 [Atlantic salmon, *Salmo salar*]). Consequently, there has been relatively little information collected on this attachment procedure compared to other techniques. Assuming tagged fish survive the procedure, it is critical that they display normal behavior to allow indicative data collection and extrapolation to the whole population. Significantly lower swimming speed has been noted for externally tagged Atlantic salmon smolts (McCleave and Stred, 1975) and juvenile white sturgeon, *Acipenser transmontanus* (Counihan and Frost, 1999). In contrast, no difference in swimming endurance was observed for adult Atlantic salmon having small or large external dummy transmitters attached compared with control fish (and surgically implanted individuals; Thorstad *et al.*, 2000). Arnold and Holford (1978) measured the physical characteristics of external transmitters to develop theoretical calculations of external transmitter effects, followed by some experimental validation. They calculate that a 40-cm fish gliding in the water column will have 2 to 4% speed reduction attributed to the transmitter, and to maintain the swimming speed of an untagged individual of comparable size will require a 1 to 4% increase in power output. Both tail beat frequency (TBF) and opercular beat rate were affected by externally attached transmitters (Lewis and Muntz, 1984). In addition, pannier arranged transmitters had higher drag effects with a greater transmitter cross-sectional area. However, negative effects were dependent on water velocity with pannier tagged fish requiring greater TBF in high water velocity, while single-cell tagged fish required greater TBF in slower moving water. Higher TBFs are likely associated with the effects of the asymmetrical load of the transmitter on body mechanics (*i.e.,* degree of bending; Lewis and Muntz, 1984). To compensate, fish have greater fin movements at low speeds. Interestingly, shorter fatigue times are also noted for externally tagged rainbow trout compared to other tagging methods (Mellas and Haynes, 1985).

Additional negative effects of external tagging include irregular swimming, occasional substrate scraping (Collins *et al.* (2000) observed shortnose sturgeon (*Acipenser brevirostrum*) rubbing external transmitters along tank sides and bottom

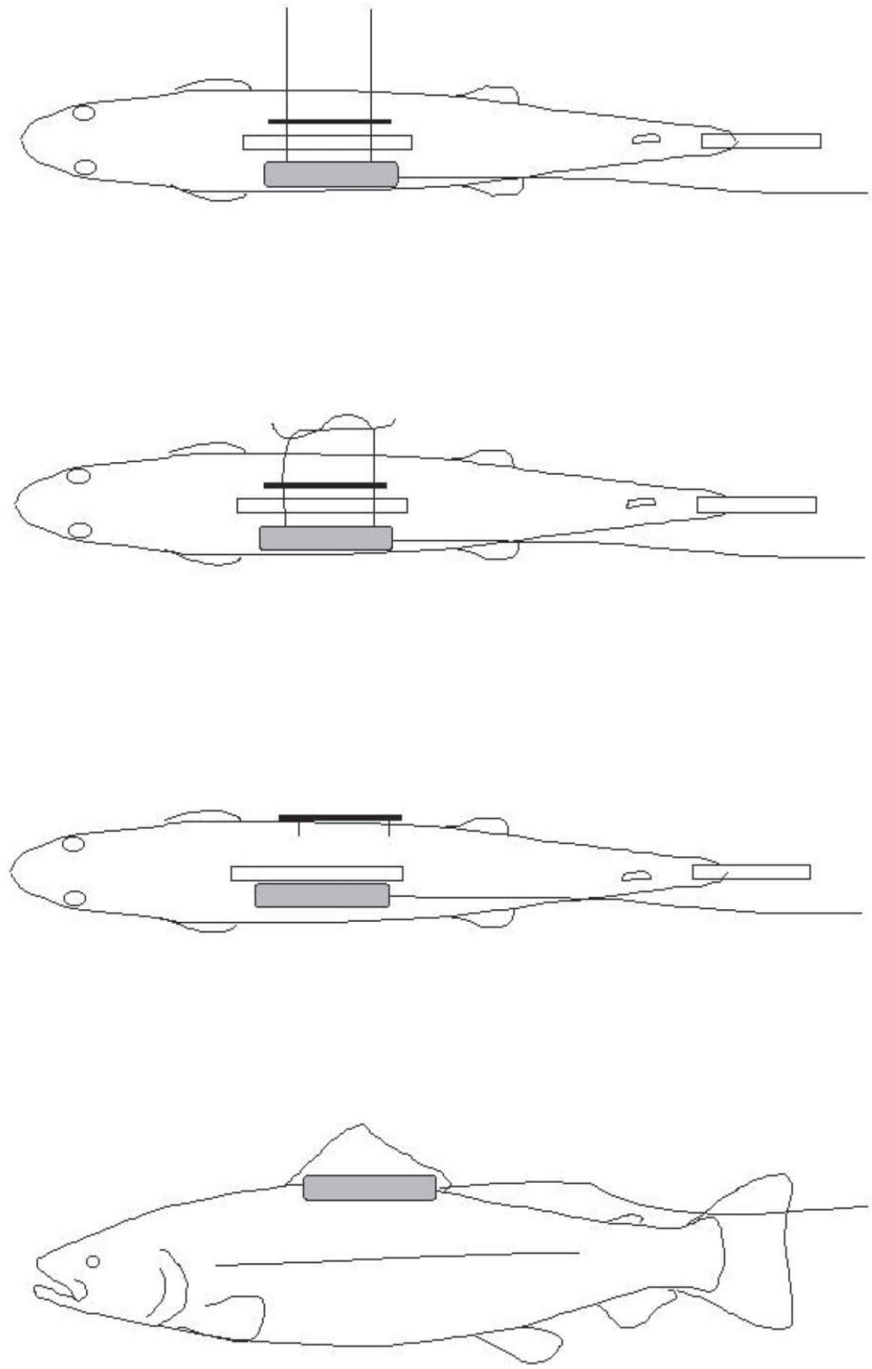

FIGURE 1. External transmitter attachment procedure positioned laterally above the lateral line. Attachment wires are passed through the dorsal musculature of the fish, looped, and tied together and tightened at the opposite side of the transmitter. The figure also illustrates use of an attachment plate on the opposite side of the transmitter to ease attachment.

causing eventual transmitter loss) and entanglement of the externally attached transmitter in the environment. External transmitter size may also affect the number of wounds and degree of inflammation. Thorstad *et al.* (2000) noted adult Atlantic salmon having large transmitters (1% of the fish body weight in water) externally attached displayed wounds compared with those having small transmitters (0.7% of the fish body weight in water). Further, external transmitters have caused severe muscle damage and dorsal scale loss in the transmitter and attachment wire vicinity (Mellas and Haynes, 1985). Growth rates may also be affected by external tagging. Greenstreet and Morgan (1989) reported attachment procedure had no significant effect on growth rates, but the presence of an externally mounted transmitter reduced the growth rate of all Atlantic salmon size classes tested. In addition, reduction in growth rate increased with time indicating potential problems with interpretation of data from long-term studies. This could have been the result of a loss of appetite by tagged individuals as demonstrated by decreased minnow consumption by dummy radio tagged bass, *Micropterus salmoides* (Ross and McCormick, 1981). Contrary to those results, Begout Anras *et al.* (1998) reported no effect of attachment procedure or transmitter presence on lake whitefish (*Coregonus clupeaformis*) mortality, growth rate, or swimming activity level.

At high aquaculture stocking densities, external transmitters may act as a lure, attracting the attention of other farmed fish. Although not an aquaculture related study, Ross and McCormick (1981) reported evidence of predation on tagged yellow perch by northern pike (*Esox lucius*). Within an aquaculture setting, this, in effect, may result in tagged fish becoming subordinate within the school and result in inaccurate data collection in high stocking densities. In addition, changes in both appetite and growth rate may affect the hierarchy of tagged individuals, potentially resulting in previously dominant fish becoming subordinate post-tagging.

B. INTRAGASTRIC INSERTION AND EFFECTS

Transmitters can be implanted intragastrically by pushing the transmitter down the pharynx, past the cardiac sphincter and into the fish's stomach. Intragastric insertion is generally considered a gentle, quick, and easy procedure that can be accomplished using a low level of anesthesia. Intragastric insertion has the advantage of eliminating the risk of snagging on surrounding vegetation or structure experienced by external attachment and avoiding additional drag forces by placing the transmitter internally. Fish are lightly anesthetized and placed upright on foam support. A plexiglass tube is used to push the transmitter into the abdominal cavity. Prior to insertion, both the tube and transmitter may be dipped in a lubricant, such as glycerin, to ease insertion. The transmitter is inserted into one end of the tube, which in turn is inserted into the mouth, and gradually, but firmly, pushed into the abdominal cavity. Once the transmitter is midway down the abdominal cavity, a rod is used to expel the transmitter from the plexi-glass tube, followed by rod/tube withdrawal (Figure 2). Radio transmitters require the antenna to be trailed out of and sutured to the roof of the mouth or passed through a gill arch opening, sutured and trailed externally along the body. The antenna should be bent prior to implantation so that the portion protruding from the fish points posteriorly. Antennas exiting the mouth may result in abrasions and subsequent infection to the corner of the mouth

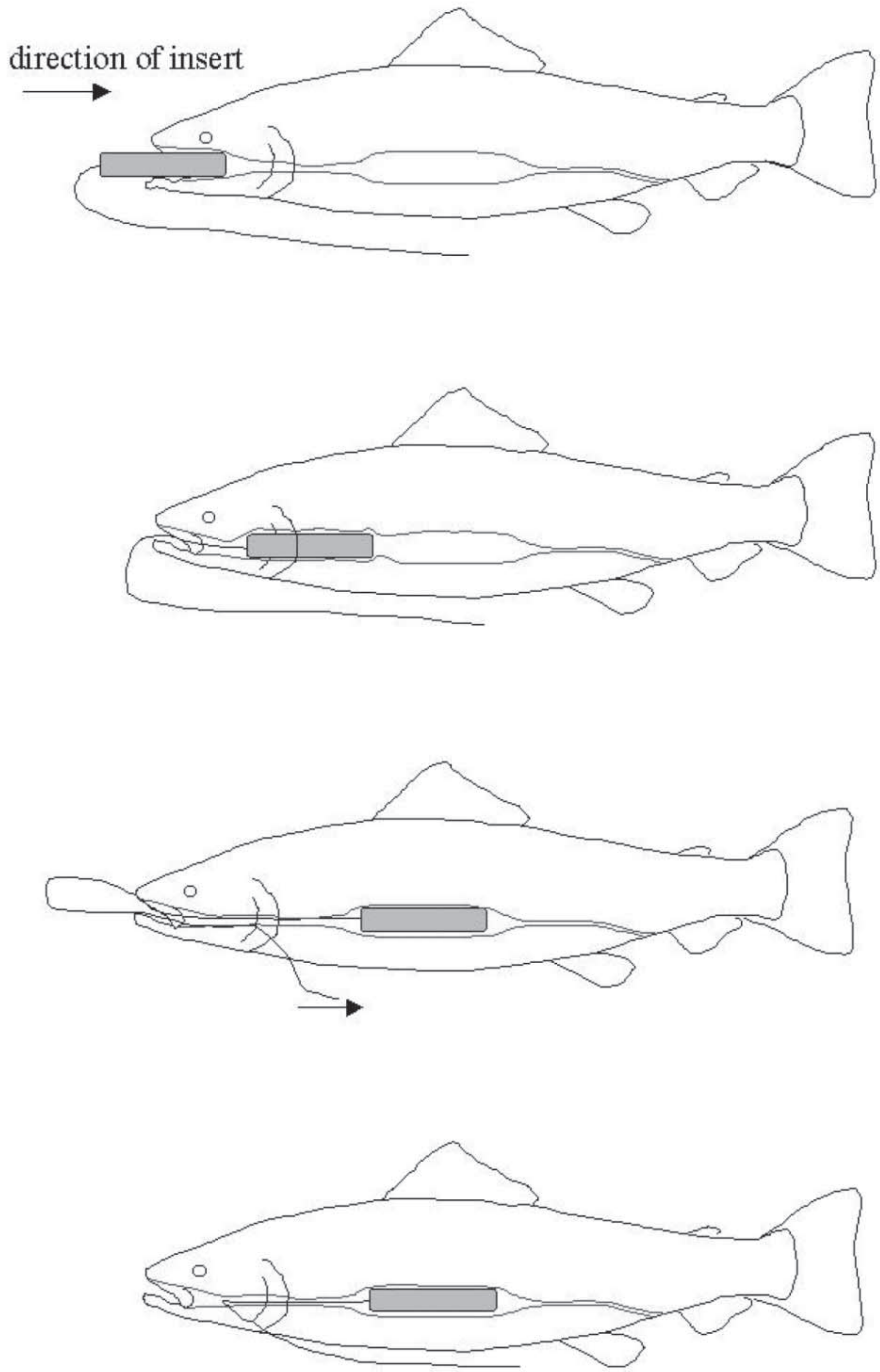

FIGURE 2. Intragastric implantation of a transmitter by pushing the transmitter down the pharynx, past the cardiac sphincter and into the fish stomach. Radio antennas are either trailed out of and sutured to the roof of the mouth or passed through a gill arch opening, sutured, and trailed externally along the body.

(Adams *et al.*, 1998a). With appropriate practice, this procedure can be accomplished within 30 sec.

Transmitter regurgitation is a concern associated with intragastric insertion. Mellas and Haynes (1985) reported 80% of experimental rainbow trout regurgitated the transmitter at least once in a 2-week period. Similar retention time for feeding cod (*Gadus morhua*) has been reported with 50% of tagged individuals regurgitating the transmitter within 5 days (Lucas and Johnstone, 1990), but more recently 32 days (Winger and Walsh, 2001). To decrease the stress and effects of fish decompression associated with bringing fish to the surface for transmitter attachment, experimental fish may voluntarily ingest transmitters placed within baitfish while still in the water column (Priede and Smith, 1986). Voluntary ingestion may extend retention time of intragastric implants in cod to eight days before 50% regurgitation (Armstrong *et al.*, 1992). The longest retention recorded to date under field conditions for a voluntary ingested transmitter is 68 days (Engås *et al.*, 1996).

Feeding can decline following intragastric insertion possibly from blocking feed intake by the transmitter or decreasing the stomach volume available for ingested feed. Armstrong and Rawlings (1993) documented reduced feeding by Atlantic salmon parr following transmitter insertion in autumn. Likewise, Adams *et al.* (1998a) and Jepsen *et al.* (2001) reported decreased feeding and growth of gastrically implanted chinook salmon (*Oncorhynchus tshawytscha*). Salmonid study results are in contrast to Atlantic cod, displaying no significant difference in average food consumption per fish (Winger and Walsh, 2001). Decreased feeding potential may limit the effective use of transmitter stomach implants to times of reduced feeding to extend the retention time of the transmitter and therefore extend the telemetry study. Observing comparable decreased growth, Martinelli *et al.* (1998) suggested using gastric implants only for short-term studies, and surgical implantation (discussed later) for longer-term studies.

Swimming performance of Atlantic salmon with stomach implants is similar to control salmon (McCleave and Stred, 1975). Additionally, following intragastric insertion, a larger proportion of inconnu (*Stenodus leucichthys*) resumed upstream migration and migrated a greater distance than externally tagged inconnu (Brown and Eiler, 2000). Atlantic salmon smolts also begin buoyancy compensation to counteract the additional transmitter weight within 1 h post-implantation, and many tagged fish regain initial buoyancy (Fried *et al.*, 1976). A buoyancy recovery period illustrates the need to recover tagged fish that gulp air in shallow water with access to the surface for several hours prior to data collection. Additionally, demersal fish may be held in cages at depth for a recovery period to ensure appropriate buoyancy is attained in their natural environment prior to data collection. Data collected immediately following release may be questionable owing to anticipated behavioral changes. For aquaculture-related studies, intragastric insertion is limited owing to the highly competitive aquaculture environment with limited food supply. Transmitters would impede the quantity of feed ingested, increasing the likelihood for transmitter regurgitation, and limiting telemetry data collection. Blood chemistry changes may also be expected following gastric insertion of transmitters (Jepsen *et al.*, 2001). Plasma cortisol levels of gastrically implanted fish significantly increased compared to unhandled fish for 24 to 48 h post-attachment, in contrast to handled and anesthetized fish increasing plasma cortisol levels but returned to unhandled fish levels just four hours post-handling. Plasma lactate and glucose levels were also significantly higher in gastrically implanted fish than controls 24 h post-insertion, but comparable to unhandled fish seven days post-insertion.

C. SURGICAL IMPLANTATION AND EFFECTS

Similar to intragastric placement, surgical implantation in the peritoneal cavity has the advantages of placing the transmitter near the center of gravity of the fish, protection from environmental entanglement, and eliminating drag forces against swimming velocities. However, peritoneal implants require a longer time, deeper anesthesia, and may result in the greatest amount of attachment-associated infection. Surgical implants typically require the fish to be placed dorsal side down in a V-shaped surgical trough (for an example of a surgical table see Courtois, 1981) following anesthesia, and constant gill irrigation with a dilute anesthetic solution throughout the procedure. It is essential to keep the skin of the fish moist/wet at all times; therefore, the foam should be wet. Oxygenated water, pumped from a small submersible pump, is directed across the head and gills for sufficient oxygen uptake. An incision approximately 2 to 5 cm long, transmitter size dependent, is made on the ventral side to allow insertion of the transmitter in the peritoneal cavity. Following successful insertion, the incision is typically closed with 3 to 5 independent sutures (Figure 3). Wagner *et al.* (2000) recommended use of a simple interrupted suture pattern to decrease suturing time and associated inflammation. An antibiotic may be injected intraperitoneally prior to the placement of the final suture to aid healing and prevent undesirable infection. However, Wagner *et al.* (1999) reported comparable wound healing and healing rate in rainbow trout treated with a topical antiseptic and those not treated. Numerous methods have been cited to close surgical incisions in fish (Table 1). However, most studies have not commented on the performance and suitability of the chosen suture material. Gilliland (1994) compared the performance and effects of four suturing materials. Natural gut sutures were the fastest to dissolve with no differences in healing response and infection between either of the suture materials. Transmitters may be lost from the body cavity if absorbable sutures dissolve prior to incision healing (Bunnell *et al.*, 1998). Studies involving implantation of radio or combined acoustic and radio transmitters (CART) require the radio antenna to exit the body cavity prior to suturing (Figure 3). The antenna may be trailed out of the posterior end of the incision, or a needle may be pushed through the fish body wall away from the incision, with the use of a scalpel guide, creating a channel for the antenna to pass through (Ross and Kleiner, 1982; Bridger *et al.*, 2001). The transmitter antenna is then threaded through the needle, at which point the needle is pulled completely out through the side of the fish leaving the antenna in place. Walsh *et al.* (2000) reported no differences in transmitter loss, mortality, or hybrid striped bass growth (*Morone saxitilis* × *Morone chrysops*) regardless of antenna placement. Alternatively, radio antennas may remain coiled within the body cavity of the fish with the transmitter. Cooke and Bunt (2001) determined no significant differences exist between the antenna configurations with respect to swimming performance or fish movement. It should be noted, however, that coiling antennae within the body may result in signal attenuation leading to decreased transmitter performance such as a reduction in range (Collins *et al.*, 2000). Finally, some transmitters may have additional appendages that must exit the body

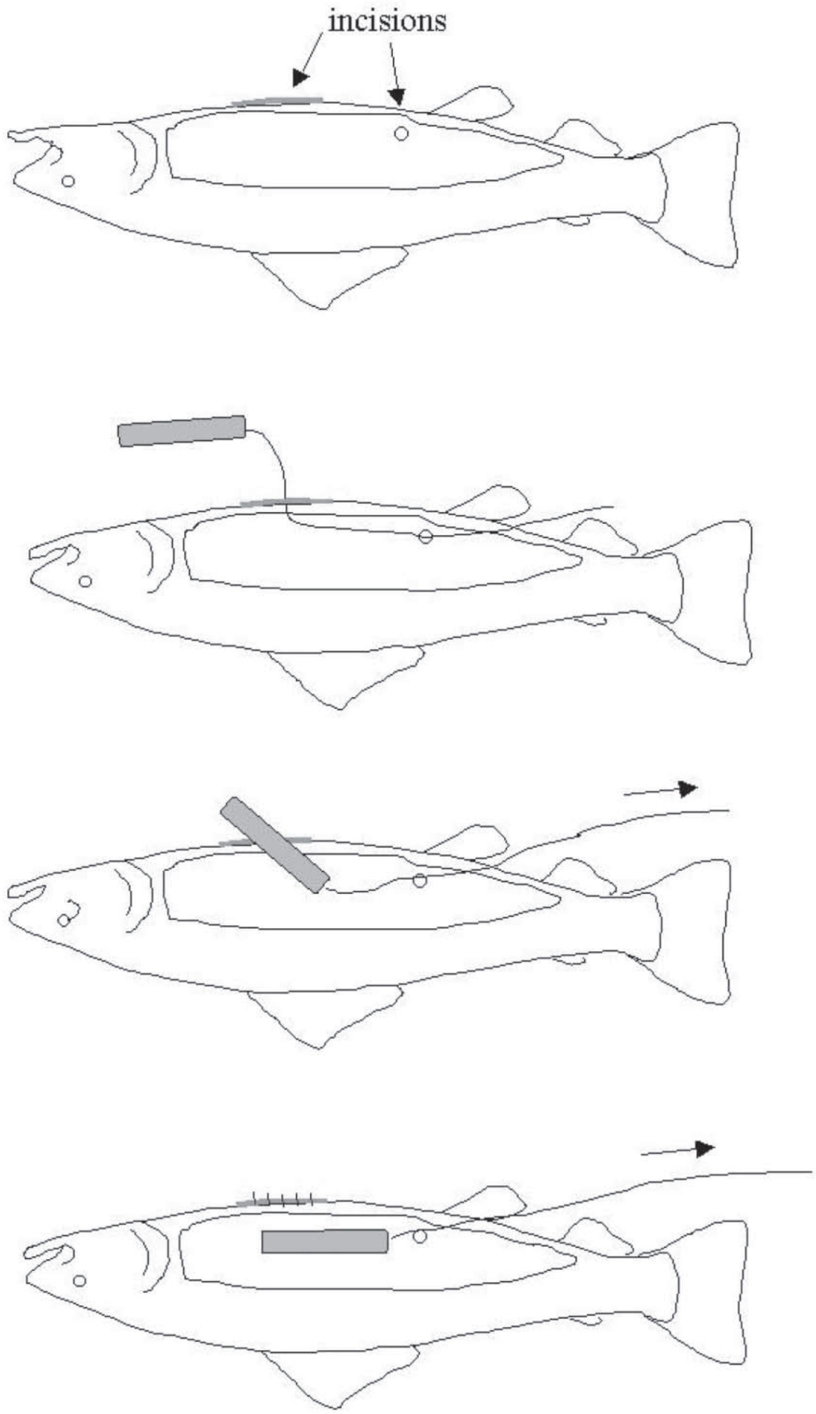

FIGURE 3. Surgical implantation of a radio transmitter into the peritoneal cavity. An incision approximately 2 to 5 cm long is made on the ventral side to allow insertion of the transmitter. Following successful insertion, the incision is closed with 3 to 5 independent sutures. Radio antennas exit the body through a needle channel and using a scalpel guide.

TABLE 1
Methods to Close Surgical Incisions in Fish Following Transmitter Implantation with Selected Species Examples

Incision Closure Method	Species	Reference
Not closed	Yellow Eels	Baras and Jeandrain, 1998
Non-absorbable sutures	Catfish	Hart and Summerfelt, 1975; Marty and Summerfelt, 1986
	Rainbow Trout	Chisholm and Hubert, 1985; Lucas, 1989
Absorbable sutures	Grass Carp	Schramm and Black, 1984
	Atlantic Salmon	Moore *et al.*, 1990
	Rainbow Trout	Martin *et al.*, 1995
Surgical skin staples	Striped Bass	Mulford, 1984
	Rainbow Trout	Swanberg *et al.*, 1999
Cyanoacrylate adhesive (superglue)	Channel Catfish	Nemetz and MacMillan, 1988
	Black Crappies	Petering and Johnson, 1991

to measure environmental parameters. An example would be the dynamic CART described by Niezgoda *et al.* (1998) having an appropriate sensor to measure the water electrical conductivity to choose the optimal mode of transmission, thereby maximizing the detection range and increasing transmitter longevity (compared with a static CART that alternates its transmission mode between radio and acoustic irrespective of water conductivity). Although dynamic CARTs required more surgical time, having an antenna and conductivity sensor that must exit the fish body, Bridger *et al.* (2000) observed no significant difference in survival between fish implanted with static (90.7%) and dynamic (88.0%) CARTs.

The location of the surgical incision is species and age dependent, with maturing and smaller fish requiring implantation anterior to the pelvic girdle. Schramm and Black (1984) tested a dorso-ventral incision, anterior and dorsal to the pelvic girdle, for transmitter implantation in grass carp. The technique reduced suture failure owing to less gravity-associated strain on the incision from the transmitter and viscera. However, increased suturing difficulty through the thicker lateral body wall, presence of developing ovaries in female fish, and involuntary muscle contractions from touching a rib while making the incision all limit lateral incisions for transmitter implantation.

Surgical implantation appears to have a minimal effect on survival, growth, and behavior of salmonids (Adams *et al.*, 1998a) and Atlantic cod (Wroblewski *et al.*, 1994; Cote *et al.*, 1999). However, Morris *et al.* (2000) noted that 50% mortality was observed in Arctic broad whitefish (*Coregonus nasus*) surgically implanted with radio transmitters in water temperature above 16°C. There is also no evidence to support changes in buoyancy, orientation, swimming, feeding, hematological response, predation, or gonad development and gamete production following transmitter implantation in rainbow trout, Atlantic salmon, Atlantic cod, or largemouth bass (Crumpton, 1982; Lucas, 1989; Moore *et al.*, 1990; Martin *et al.*, 1995; Cote *et al.*, 1999; Thorstad *et al.*, 2000; Perry *et al.*, 2001). Koed and Thorstad (2001) also demonstrated that adult pikeperch (*Stizostedion lucioperca*) retaining surgically implanted radio transmitters for 1 year displayed no significant difference in swimming performance compared to control pikeperch. In contrast, Adams *et al.* (1998b) observed significantly lower critical swimming speeds by juvenile chinook salmon and significantly higher predation on gastrically and surgically implanted fish. Additionally, tilapia (*Oreochromis aureus*) required up to three days for buoyancy compensation post-implantation (Thoreau and Baras, 1997). Perry *et al.* (2001) conclude that complete buoyancy compensation of juvenile salmonids using the swim bladder may not occur and fish will require additional energy to be expended in swimming to maintain a depth in the water column. These authors further speculate that transmitter implanted juvenile fish might remain in shallow water to reduce additional energy expenditure required for buoyancy compensation, potentially affecting data collection from tagged fish that is not necessarily indicative of natural fish behavior. To eliminate swimbladder complications resulting from bringing deep fish to the surface, Starr *et al.* (2000) developed a surgical procedure performed by scuba divers at depth. Additionally, Star-Oddi has recently developed a commercial deep-sea robot tagger (www.star-oddi.com).

Although somewhat debatable, results from recent studies have failed to show an effect of transmitter attachment on the growth rate of fish. For example, Martin *et al.* (1995) noted that control fish gained weight, while implanted fish lost weight during their trial, and Jepsen and Aarestrup (1999) indicated lower growth rates of tagged pike possibly from a short-term negative effect that was later compensated for. Paukert *et al.* (2001) also determined transmitter size affected growth and consequently fish condition in bluegill (*Lepomis macrochirus*). No initial differences have been noted with regard to social interactions of implanted rainbow trout with control rainbow trout (Swanberg and Geist, 1997). Dominant trout prior to implantation remained dominant following surgical implantation in equal-size pair-wise challenges with no significant differences in the number of nips, chases, or interaction time pre- and post-implantation. Additionally, Wroblewski *et al.* (1994) noted a surgically implanted Atlantic cod was later captured in a cod trap with several thousand other cod illustrating implanted cod remain healthy and display natural behavior post-surgery. This is extremely important for aquaculture research where the behavior of cultured fish, in an aquaculture setting, is to be determined, and where growth rate effects as mentioned above may result in long-term size effects on implanted fish, possibly affecting the social behavior within an aquaculture cage.

Aside from effects on growth rate, there may be some short term physiological responses to transmitter attachment. Moore *et al.* (1990) found that opercula rate increased for approximately 1 h post-implantation, possibly associated with surgical stress. Jepsen *et al.* (2001) observed an almost three times increase in plasma cortisol levels and significant increases in plasma lactate and glucose in surgically implanted juvenile chinook salmon 3 h after surgery, compared with unhandled control fish. In addition, surgically implanted chinook displayed significantly less plasma lactate levels than gastrically inserted chinook, and glucose levels were between gastrically inserted and control chinook, 24 h after the procedures. Likewise, transmitter implanted bluegill had significantly lower hepatosomatic (HSI) and viscerosomatic (VSI) indices than control bluegill (Paukert *et al.*, 2001) that was attributed to greater stress in transmitter-implanted fish.

Some movement of intraperitoneally implanted transmitters may be possible (Mellas and Haynes, 1985). To prevent this, internal suturing of the tag has been suggested (Schramm and Black, 1984). However, natural encapsulation of transmitters in a fibrous capsule is evident for several species, preventing internal movement of the transmitter and decreasing internal organ damage (Summerfelt and Mosier, 1984 [catfish]; Marty and Summerfelt, 1986 [catfish]; Lucas, 1989 [rainbow trout]; Moore *et al*., 1990 [Atlantic salmon]; Thoreau and Baras, 1997 [tilapia]; Cote *et al*., 1999 [Atlantic cod]; Koed and Thorstad, 2001 [pikeperch]).

A disadvantage to surgical implantation is the potential loss or expulsion of the transmitter from the body cavity. Schramm and Black (1984) reported 20% of grass carp lost their surgically implanted transmitters through broken sutures. Biological expulsion of a transmitter may occur either through the anus or body wall and the mode is not species dependent (Moore *et al*., 1990; Summerfelt and Mosier, 1984; Marty and Summerfelt, 1986; Chisholm and Hubert, 1985; Lucas, 1989; Baras and Westerloppe, 1999). Tilapia, however, retained transmitters for up to 30 months (Thoreau and Baras, 1997). In contrast to those studies, Martin *et al*. (1995) reported no observed evidence of transmitter encapsulation and no loss of surgically implanted transmitters from rainbow trout. Baras and Westerloppe (1999) suggests decreasing the degree of contact between the transmitter and intestine may reduce the expulsion likelihood (*e.g*., positioning the tag toward the tail, larger volumes of fat or gonads). Additionally, Helm and Tyus (1992) reported the transmitter coating type influenced the retention of surgically implanted dummy transmitters. Bees wax coated dummy transmitters were more likely to be retained compared to paraffin and silicone coated transmitters. Finally, water temperature differences in all these experiences may affect transmitter expulsion (Knights and Lasee, 1996; Bunnell and Isely, 1999).

D. ADDITIONAL ATTACHMENT METHODS

A transmitter may be inserted in the female oviduct of salmonids. Peake *et al*. (1997a) described a method of gently pushing a transmitter into the oviduct of Atlantic salmon and rainbow trout through the genital papilla and allowing the antenna to freely exit the oviduct. Following insertion, no behavioral or survival differences were observed; however, 33% of the tagged rainbow trout died during the observation period. Similar to surgical implantation, transmitters were encapsulated by a thin sheath of tissue and 31% of oviduct tagged Atlantic salmon expelled the transmitter 7 to 13 days post-insertion. Obviously, oviduct insertion would be limited to female fish but might offer a suitable alternative to transmitter attachment in some species.

CONCLUSIONS

Biotelemetry success requires secure attachment of the transmitter without unduly affecting the natural behavior or physiological fitness of that animal. Prior to attachment, individual fish must be sedated or anesthetized, with the level and method dependent on the species, attachment procedure to be employed, environmental conditions, and local regulations prohibiting some chemical use. The three major transmitter attachment procedures discussed — external attachment, intragastric insertion, and surgical implantation — have advantages and disadvantages that must be considered prior to use. The researcher must carefully consider the study with regards to the study species and characteristics including morphology and individual size; study objectives and duration; and, environmental conditions, including habitat for possible tangling, water temperature, and potential fouling organisms to determine the appropriate attachment method.

Arguably, intragastric insertion has the least detrimental effect on fish. However, a high percentage of regurgitation is evident for some species and often dependent on season and fish behavior perhaps limiting intragastric attachment to short-term studies less than 20 days duration. Surgical implantation involves considerable handling, deeper anesthesia, and longer recovery time. However, these disadvantages are far outweighed by benefits to long-term studies (> 20 days duration) of most fish species. External transmitters have been demonstrated to influence swimming performance and often problematic in some environments. For these reasons, external attachment procedures should be avoided in all cases unless it is deemed, with a doubt, to be the only effective method of attachment (*e.g.,* fish morphology). Care should be exercised in direct comparisons between cited experiments and researcher's ongoing studies owing to different species, transmitter types and shapes, tag-to-fish weight ratios, and researchers performing the attachment procedures. It is therefore highly recommended that whenever possible each researcher develop the most suitable anesthetizing and attachment procedure for their studies and test each candidate procedure for adverse effects on fish behavior prior to data collection. Additionally, experimental fish should be taken from the study population. Hatchery fish – aquaculture or stock enhancement purposes – of most species are readily available. However, owing to morphological and behavioral differences between hatchery and wild individuals (reviewed in Bridger and Garber, 2002), caution should be exercised in interpreting results attained from hatchery individuals and extrapolating to wild conspecifics (Peake *et al.*, 1997b).

Working closely with telemetry transmitter manufacturers, telemetry researchers may develop transmitters that have less potential impact on study animals. For instance, transmitters may be developed that are neutrally buoyant. Such transmitters would not affect the quantity of air required in the swim bladder to compensate for the transmitter weight, and therefore transmitter-attached fish would be expected to behave in a comparable fashion to untagged individuals. Additionally, to alleviate attachment-associated stress in the data, transmitters may be programmed to delay signal transmission to after a predetermined period following attachment. Delayed transmission would ensure that data are collected following a sufficient recovery period and therefore indicative of natural behavior. Alternatively, from the studies reviewed, researchers would be well advised to exercise caution when analyzing data collected within 24 h of transmitter attachment to ensure fish have properly recovered from the anesthetic, attachment procedure, and transmitter presence.

Telemetry is becoming a powerful tool for fisheries and aquaculture research. When used properly, the quantity and quality of data collected from telemetry studies cannot be equaled employing conventional research methods alone. However, it is critical that all researchers become aware of possible effects that transmitter presence and attachment procedure may have on fish behavior and physiology.

Telemetry systems may vary in total system costs, dependent on the equipment used and complexity required in the aquatic environment and scope of the research study. For most research projects, it is highly recommended that a combination of telemetry and conventional research methods will provide the most complete data set. A hybrid methodology design will ensure the desired level of population sample size and study power is achieved to acquire the most complete picture of fish behavior and physiology in relation to the environment. Proper study design will ensure that appropriate conclusions and management plans are created from collected telemetry data, augmented by other data collection methods.

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