A review of animal and human health concerns during capture-release, handling and tagging of odontocetes

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ABSTRACT

The capture-release of odontocetes allows for tag deployment which provides an opportunity to study behaviour and habitat use by free-ranging animals, as well as clinical assessment of the animal and tissue collection. This review recognises those elements that are common to most capture and tagging projects, identifies collective knowledge of animal and human health concerns during handling of odontocetes and provides guidelines for safer handling techniques. Handling during tagging projects can involve chase, capture, restraint, manipulation, tag application, often removal from the water and release at the capture site. The risk of injury during capture will be reduced by using experienced personnel, adequate technical support and proper equipment. For the duration of the handling process, the animal's stimulus response should be monitored as well as its cardiovascular and respiratory function. Stress response of the odontocete is monitored by behavioural assessments, physiological monitoring and/or blood sampling. Possible complications from tag placement may include infection at the implant site leading to tag failure, behavioural alterations in response to tag placement and tag rejection. During handling of an odontocete, there is the potential for disease transmission between humans and the animal. Exposure to diseases is minimised by wearing protective clothing and gear and exercising caution when working around the animal's blowhole.

KEYWORDS: DISEASE; LIVE-CAPTURE; RADIO-TAGGING; SATELLITE TAGGING; STRESS

INTRODUCTION

Tagging and tracking odontocetes allows biologists and wildlife managers to study behaviour and assess habitat and resource use by free-ranging animals. Current techniques for long-term (e.g. longer than a week) tag attachments to smaller odontocetes require that animals are captured and held for a brief period of time while the tag is attached (Irvine *et al.*, 1982; Würsig, 1982; Tanaka, 1987; Tanaka *et al.*, 1987; Scott *et al.*, 1990; Martin *et al.*, 1993; 2001; Hanson *et al.*, 1998; Hanson and DeLong, 1999; Martin and Smith, 1999; da Silva and Martin, 2000; Ferrero *et al.*, 2000; Richard *et al.*, 2001). Capture of animals for tag deployment also provides an opportunity for clinical health and body condition assessment of the animal (Wells *et al.*, In press), determination of gender, collection of teeth for age determination (Hohn *et al.*, 1989), evaluation of contaminant burdens (Schwacke *et al.*, 2002) and assessment of reproductive condition (Wells, 2003). This review attempts to identify those elements that are common to most capture and radio- and/or satellite-tagging projects, to codify collective knowledge regarding animal and human health concerns and to provide guidelines for safer handling practices. This document is intended as a reference for experienced researchers and a resource for inexperienced researchers contemplating new projects, but is by no means a comprehensive review of specific projects or all odontocete species to be tagged. It should be kept in mind that the capture and handling of odontocetes can vary greatly amongst and within species and locations and that different species may have some different handling and monitoring requirements. Although this review describes procedures requiring varying skill levels and makes recommendations regarding their application, reference to this document should not be considered a substitute for including experienced personnel in the field party. A general rule of thumb is that the degree to which an animal is compromised increases with the amount of handling and the length of time that the animal is handled. An individual animal may not display any obvious outward signs of being compromised beyond a threshold from which it cannot recover, as evidenced by the occasional sudden death of an animal that otherwise outwardly appears to be tolerating handling. Thus, handling should be kept to the minimum necessary to complete the research objectives.

Clearly, capture-release is not the only option and in cases where capture and handling is not feasible or a short-term, less invasive attachment is desired, remote deployment methods such as suction cups and barbed or toggled attachments may suffice especially for larger cetaceans (e.g. blue whales (*Balaenoptera musculus*), northern bottlenose whales (*Hyperoodon ampullatus*), bowhead whales (*Balaena mysticetus*), Dall's porpoise (*Phocoenoides dalli*), fin whales (*Balaenoptera physalus*), humpback whales (*Megaptera novaeangliae*), killer whales (*Orcinus orca*), narwhals (*Monodon monoceros*), northern right whales (*Eubalaena glacialis*) and sperm whales (*Physeter macrocephalus*) – Watkins *et al.*, 1984; 2002; Watkins and Tyack, 1991; Baird, 1994; Baird and Hanson, 1997; Mate *et al.*, 1997; 1998; 1999; 2000; Hooker *et al.*, 2000; Laidre *et al.*, 2002). Although remote attachment methods are not reviewed here, this review will give researchers faced with a choice of methods more information on the tradeoffs involved with a capture and handling technique.

Handling during tagging projects can involve chase, capture, restraint, manipulation, tag application and often removal of the animal from the water, followed by release at the capture site. Several health concerns should be kept in mind during these phases:

(1) physical injury of both humans and animals during the handling process;

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(2) the physical and physiological effects of capture on the animal;
(3) possible complications from tag placement;
(4) the potential for disease transmission between humans and animals.

These concerns are addressed and suggestions are provided to minimise disease transmission and injury to all individuals involved. Methods are outlined for assessing and monitoring the animal during handling and tagging in the field.

Evolution in the marine environment, as a herd animal subject to predation, provides marine mammals with unique adaptations to cope with acute, short-term stressors and maintain homeostasis (St Aubin and Dierauf, 2001). However, some aspects of tagging operations, such as restraint in a sling and close proximity to humans, fall outside of the normal range of adaptation. The immediate and long-term effects of handling on the health and behaviour of these animals is unclear, but given the considerable cost for each tag deployment and subsequent data collection and the implicit assumption that the tagged animals are representative of the population of interest, close monitoring of the animal's biological parameters and risk reduction during handling become important.

RISK OF INJURY DURING HANDLING

Capture

The capture itself can be dangerous to both humans and the animals. Several techniques for capturing and handling white whales (*Delphinapterus leucas*) and other small odontocetes have been described. Seine nets have been used to capture species such as bottlenose dolphins (*Tursiops truncatus* —Asper, 1975), killer whales (*Orcinus orca*) and botos, or Amazon River dolphins (*Inia geoffrensis* —da Silva and Martin, 2000). Breakaway hoop nets have been successfully used in capturing bottlenose dolphins and porpoises (Ridgway, 1966; Asper, 1975; Hanson, 1998). In certain areas, pilot whales (*Globicephala melas* —Heide-Jørgensen *et al.*, 2002) and white whales (Sergeant and Brodie, 1969; Martin and Smith, 1992) can be driven to shore at low tide or into shallow water where they can be grounded or captured with a hoop net over the head (Orr *et al.*, 2001). Entanglement in a drift or set-net has been used to capture white whales (Orr *et al.*, 2001) and narwhals (Dietz and Heide-J¢rgensen, 1995; Dietz *et al.*, 2001), and an encirclement technique along with high speed net deployment has also been used to capture white whales in the Cook Inlet, Alaska region (Ferrero *et al.*, 2000) and pelagic dolphin schools in the eastern tropical Pacific Ocean (Perrin *et al.*, 1979; Jennings *et al.*, 1981). A human handling a cetacean may be struck by a thrashing tail or rostrum, or injured by netting or restraining devices. Removing it from a net or other capture device can also be stressful or traumatic to the animal and operators. Handlers can eliminate much of their own risk by minimising carelessness and planning handling practices with forethought. Once restrained, white whales, as well as narwhals, seldom continue to struggle for more than a few minutes (Orr *et al.*, 2001; Heide-Jørgensen, pers. comm.). Smaller species such as harbour (*Phocoena phocoena*) and Dall's porpoise may struggle (Norris and Prescott, 1961) for a longer period of time than larger species such as white whales and killer whales (Walker, pers. comm.). In contrast to most odontocetes, botos are much more comfortable laying on their side than on their bellies and consequently struggle less (da Silva and Martin, 2000). Tucuxis (*Sotalia fluviatilis*), however, are more nervous than botos when being handled (Martin, pers. comm.).

During capture and handling procedures, a cetacean sometimes incurs physical injuries such as abrasions, lacerations, contusions, or injuries resulting from malpositioning or unruly behaviour (Spraker, 1982). The risk of some of these injuries may be reduced by using skilled, experienced personnel and adequate technical support and proper equipment to handle the animal, removing or covering hazardous objects in the work area, and placing padding around its body (sand, foam pads, or other appropriate material) if it is fully or partially removed from the water.

Monitoring of animals caught in a net

In net captures, there are significant risks of death if an animal is trapped below the water surface for too long, or of aspiration of water into the lungs if the animal is trapped near the water's surface and allowed to struggle for a prolonged period (Walker, pers. comm.). If a significant volume of water has been aspirated at the water's surface while struggling in a net, the animal may have difficulty getting sufficient oxygen into its bloodstream and may die during handling or after release. Further, aspiration of seawater into the lungs may introduce infection that can result in pneumonia. On the other hand, cetaceans trapped below the surface may not struggle at all or give little indication of entanglement. A capture team should remain aware of the possibility of multiple entanglements, and that an apparently single animal may have been accompanied by one or more animals unseen below the surface. The following procedures should be undertaken to reduce these risks:

(1) nets in the water should be watched continuously for movement or dips in the floatline and should be patrolled regularly in cases where the full length is not visible from a single vantage point;
(2) in murky or turbid water, where the full depth of the net is not visible from the surface, the net may be raised periodically until the lead line is visible or, alternatively, the cork line may be closely observed for movement or sinking;
(3) nets should not be left in the water unattended and should be removed from the water when not in use;
(4) mesh size should be selected to limit the risk of capturing non-target animals;
(5) net dimensions should be limited to the minimum necessary for the operation to reduce the chance of multiple captures (e.g. during captures of white whales, limited water visibility has prevented detection of non-target animals trapped under the surface with the target animal);
(6) entangled animals should be quickly supported and removed from the net;
(7) team size should be sufficient to provide the appropriate number of people for support of each animal in the water, assuming the maximum number of animals likely to be caught in each set, with at least enough additional people held in reserve on a mobile vessel to get to a place where another animal strikes the net;
(8) the field team should make contingency plans for handling multiple captures, releasing excess animals, captures of mother-calf pairs, and net handling, while the team is occupied with tagging of captured animals;

(9) capture operations should not be conducted in marginal weather conditions;
(10) the person responsible for setting and retrieving the net should have a high level of familiarity with the area, including water depth, current patterns, tide, seafloor type, presence of entangling objects and non target individuals, etc;
(11) every effort should be made not to set a net around very young calves and their mothers, or other potentially compromised individuals, unless they are the targets of the study as nursing calves are especially sensitive to capture and handling;
(12) a cetacean found entangled should be brought quickly to the surface and supported with its blowhole well exposed and protected from waves washing over − if the animal does not clear its blowhole and begin breathing normally, or it begins breathing in a weak, laboured or unusual manner, respiration can sometimes be stimulated by light splashing of the forehead region with water.

The design of the net (e.g. length, depth, mesh size and twine, type and size of lead and float lines, single panel or multipanel, how it hangs) is dependent upon the species to be captured and the field situation. There is no substitute for experience with nets and capture techniques. Rather than incurring unnecessary risk, prolonged trial and error should be avoided by consulting knowledgeable researchers involved with a similar species and field setting. Even with the best advice, however, it can take several field seasons to develop a reliably efficient and safe technique for a novel situation (Ferrero *et al.*, 2000; Orr *et al.*, 2001). A simulated capture, using the boats and equipment, may help avoid errors during the actual event.

Moving and restraining a captured odontocete

An animal should be lifted using an appropriately designed sling. A compartment syndrome can develop if an animal is restrained without appropriate support. This syndrome is a condition in which increased pressure within a limited space compromises the circulation and function of the tissues within that space (e.g. muscle compressed by an ill-fitting sling), as a result from trauma, prolonged recumbency or physical activity (Matsen, 1975). In order of preference:

(1) the animal should be restrained and moved while fully supported in water;
(2) the animal should be restrained by a sling, stretcher or straps, or intentionally grounded and held or moved while partially supported in water;
(3) in water that is over the handlers' heads, the animal may be placed on floating mats for disentanglement and monitoring;
(4) once on a mat, the animal may be towed to the processing vessel or shoreline;
(5) depending on its weight, the animal should be either carried in a sling or stretcher or restrained and moved while properly supported on a sponge rubber mattress (e.g. placed on the deck of a ship, a pallet, gurney, or sledge − a hoist can lift the animal onto the processing vessel);
(6) if on a beach, the animal can be carefully dragged on a tarp or blanket if the substrate is reasonably smooth.

The animal's comfort can be maintained during tagging and blood drawing by: protecting the eyes and blowhole from direct sunlight, dirt and debris; allowing the flippers to lie in a natural position either tucked or extended; keeping the skin moist; and distributing and minimising pressure to the abdominal and thoracic cavities. Care should be taken to ensure that water does not enter the blowhole, as cetaceans do not have a mechanism to expel water from the lungs. Further, water may carry infectious microorganisms into the lungs. Pressure can be minimised by padding as discussed above, or in the case of intentional grounding, by grounding the animal at the deepest depth that still allows necessary control of the animal. If sand or debris adheres to the corneal surface, the eyes should be rinsed with salt water. If the substrate allows, holes can be dug under the flippers, or if the animal is to be held in a sling or stretcher, holes are cut to allow the flippers to protrude. A pressure sprayer provides an excellent way to keep a cetacean's skin moist throughout all procedures, but buckets and sponges work as well. Moistened towels may also be used. When the air temperature and humidity are high, and if the water temperature is significantly higher than typical for the species, the water being used for moistening should be cooled if possible.

Physical and chemical restraint or sedation

Physical restraints should be made of pliable material and be broad enough to avoid pressure points. Cargo lifting straps (5-10cm width), broad nylon straps (5-10cm width), canvas or nylon slings that are fleece or foam covered, head nets with foam covered rims and tail ropes with garden hose sleeves are some examples of restraints. Tail ropes may also be made of heavy cotton or 3.5cm soft braided nylon. However, if the animal is struggling, nylon rope might create burns unless covered by a protective material such as a hose. Restraints should be simple and convenient to use, and, most importantly, easy and quick to remove when the procedure is completed, or if the animal should be immediately released. Restraints should be checked regularly to ensure they are not too tight. If the restraint squeezes, lifts, bears the weight of the animal or puts even moderate pressure along the length of the animal, then the animal should be checked at regular intervals (e.g. every 5mins) and shifted if possible to redistribute its weight avoiding subsequent pressure sores. Finally, some thought should be given as to how the restraint will fall off the animal if it should swim away with a restraint device still in place.

In general, once a cetacean is properly restrained, it tends to calm down (time frame is variable by species). In some instances sedation may be used to assist in restraint. In very rare instances, chemical immobilisation may be needed, which has been accomplished by careful use of various chemical agents (Joseph and Cornell, 1988; Reidarson *et al.*, 1998). While chemical immobilisation should not be administered to animals that are to be immediately released, they may be useful in cases where the animal is fractious or longer duration procedures are anticipated. Sedatives and other chemical agents are ideally only administered by veterinarians, as several of these agents are controlled substances and response of an animal to them may, at times, require resuscitation or other medical intervention such as administration of reversal agents. The risk of using a sedative, tranquiliser or other chemicals on a potentially stressed odontocete must be weighed against the benefits gained from using such agents to achieve a research objective. Temperature and respiration should be monitored at regular intervals. Adequate positioning of the animal is needed to prevent ischemia or compartment syndrome due to inappropriate weight bearing. Where it is not feasible to have a veterinarian in the field, at least one member of the tagging team should be trained by a veterinarian to estimate proper doses, identify symptoms of overdose, and be prepared to abort tagging efforts if the animal is in unacceptable or life-threatening distress. Precapture training and preparation can reduce the risk of loss of a valuable research animal, decrease the time and effort involved in capture, ensure the release of a healthy animal and improve the quality of the resulting data.

PHYSICAL AND PHYSIOLOGICAL EFFECTS ON THE ODONTOCETE

One of several methods may be used to monitor a cetacean's condition throughout the handling process. A collective group of signs must be monitored since no one sign will necessarily indicate trouble. Once caught, the animal should be observed for a period of time (5-10mins), during which time minimal data gathering may begin, to see if it becomes immediately distressed. When a period of observation is not feasible or significantly prolongs the holding time, one member of the team should be given the responsibility to monitor the animal closely. The individual should record the specific times of respirations and note the strength of breaths and body posture, particularly as the animal is being brought aboard the boat or beached and preliminary measurements are being taken. Depending on the primary objectives of the project, the most critical procedures should be done first (e.g. length measurements, photographs, blood drawing) in the event the animal has to be released back to the water prematurely. An animal in distress may exhibit an arching of the body (e.g. flukes and head bent upward while breath holding) often followed by thrashing, or may have very shallow, erratic respirations (Walker, pers. comm.). If so, it should be splashed with a bucketful of seawater over the melon to stimulate breathing. If there is no response, it should be returned to the water (with the potential for drug administration) and monitored until it returns to normal. It can then be released or evaluated for tagging. Behavioural criteria such as response to stimuli can be assessed as follows.

Assessing stimulus response

An animal should appear to be aware of its surroundings and respond fairly readily to stimuli. Gentle tapping near the eye should elicit a blink (Geraci and Lounsbury, 1993). A blank, unresponsive 'stare' warrants real concern and should prompt immediate action to be taken by the individual monitoring the animal.

Monitoring cardiovascular function

Cardiovascular and respiratory function can be roughly evaluated in the field by monitoring heart and respiratory rates, respectively (Table 1). A stethoscope (for smaller species e.g. harbour or Dall's porpoise) or hand (larger species e.g. bottlenose dolphin and larger) may be placed firmly in the axillary region (where the pectoral flipper joins the body wall; Fig. 1) to detect a heartbeat and determine heart rate (Geraci and Lounsbury, 1993). In small species, one can sometimes see a heart rate 'flutter' externally in the axillary region just behind the pectoral flipper. Heart rate may also be monitored with a heart rate sensor that allows data to be recorded and stored continuously during the handling process. Heart rate can and should vary considerably, even under normal conditions. For example, the heart rate of a bottlenose dolphin increases to a rate of 70-100 beats per minute (bpm) just after inspiration. As the animal continues to hold its breath, heart rate falls to between 30-40 bpm until the next breath (Ridgway, 1972). The rate will remain low regardless of the length of the apneustic plateau. Thus a normal respiratory rate of 2-3 times a minute will be accompanied by an increase, then decrease in heart rate as just described (Ridgway, 1972). If this normal sinus arrhythmia is absent, a pulse that is rapid or weak signals the onset of cardiovascular deterioration (i.e. shock, hyperthermia).

Monitoring of respiratory function

Monitoring of respiratory function should begin as soon as the animal is captured in the net. Respiratory rate for smaller species is usually 2-3 respirations per minute (rpm), but may increase to 6-8 rpm in excited individuals, and 1-2 rpm for larger species (Geraci and Lounsbury, 1993). The researcher should come into the field with the best available knowledge of average and maximum breath intervals for the species. Irregular or increased respiratory rate (i.e. >10 and 6, respectively) can signal respiratory fatigue and distress. In some species, however, respiratory intervals may become prolonged. In bottlenose dolphins, for example, an interval between respirations that extends to >1-1.5min with little respiratory chest movement occurring, is cause for concern. These 'ineffective respirations' may require immediate action on the part of the individual monitoring the animal. Changing the animal's position (e.g. from lateral to sternal recumbency) and splashing water on the melon can improve the respiratory rate and quality. One should also watch for a cetacean keeping its blowhole open and breathing in a rapid, shallow manner that often indicates stress. If this behaviour occurs or the animal's respiration 'shuts down', sometimes a

Table 1

Physical parameters for monitoring condition of cetacean during handling process (adapted from Ridgway, 1972).

Parameter	Normal range	Interpretation of abnormal values
Heart rate (bpm[1])	70-100 (after inspiration) 30-40 (during breath hold)	<30 = hypothermia >100 = hyperthermia or shock
Respiratory rate (rpm[2])	2-3 (resting smaller species)	>8 = distress (respiratory fatigue or hyperthermia)
	6-8 (excited smaller species)	
	1-2 (resting larger species) 3-6 (excited larger species)	>6 = distress (respiratory fatigue or hyperthermia)
Body temperature (°C)	36.5-37 (small to medium species)	<35.6 = cardiovascular collapse[3] >40 = impending hyperthermia

[1]bpm = beats per minute (depends on size of species, as smaller animals may have faster rates);
[2]rpm = respirations per minute (depends on size of species, as smaller animals may have faster rates);
[3]white whales and some larger whales may have lower core body temperatures.

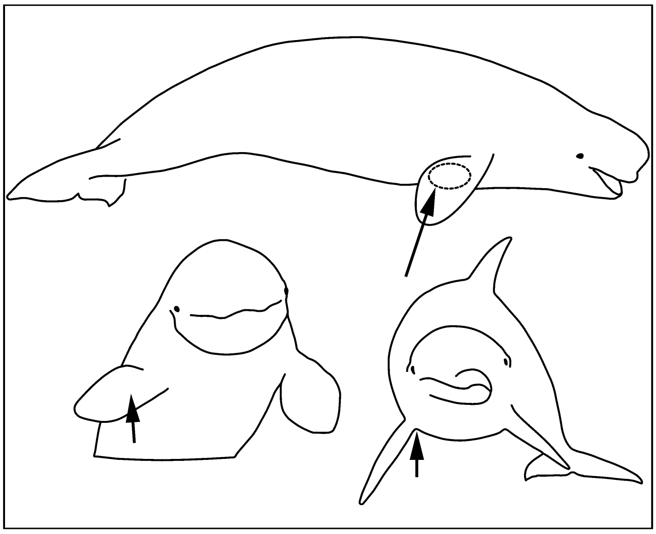

Fig. 1. Head-on and lateral views of odontocetes demonstrating placement of stethoscope or hand on the thoracic wall in the axillary region for heart rate determination (arrows).

light spraying of the head region with water will stimulate resumption of a normal respiratory pattern. Splashing a sponge full of water directly over the blowhole area will often stimulate a respiration when the dolphin is 'breath holding' (i.e. holding its breath longer than a minute). If these methods are not successful, the animal can be placed back in the water, and if it settles down, an attempt to finish the needed procedures can be made in the water. Alternatively, it may be prudent to release the animal without further procedures once the animal resumes normal respiratory patterns.

Body temperature monitoring

Body temperature can be monitored when practical to do so by insertion of a flexible temperature probe into the anal opening (at least 15-25cm or more depending on the animal's size). With the animal properly restrained, another person can reach under the animal and insert the probe. In males, one must avoid placing the probe near the gonads to prevent a false reading of hypothermia due to the presence of a vascular plexus that cools the testes (Rommel *et al.*, 1994). If insertion of a probe is not practical, the pectoral flippers, flukes and dorsal fin may be felt frequently to help assess changes in body temperature. This is the most commonly used method to monitor body temperature in a field setting. In small to medium-sized species, the normal body temperature range is 36.5°-37°C. Temperatures below 35.6°C signal the onset of hypothermia or cardiovascular shock in some species (Geraci and Lounsbury, 1993).

Hyperthermia is less likely to occur during capture and handling of cetaceans if proper procedures are followed and the team is vigilant in monitoring the status of the animal. Although hyperthermia may damage all organ systems, the nervous and reproductive systems are the most sensitive (Fowler, 1996). This is less likely to happen in the polar latitudes. Clinical signs of hyperthermia observable in the field include a shallow, rapid respiratory rate, increased heart rate, increased body temperature (>40°C) and decreased blood pressure. The core body temperature is one of the most sensitive and accurate measures of an animal's change above its thermal neutral-zone into hyperthermia. If the animal's temperature rises more than 1.5-2°C, cold water or crushed ice should be applied to the flukes, flippers and dorsal fin. The ability to monitor blood pressure may be impractical or limited in a field setting. Preventative measures are usually the best approach, such as providing shade and keeping the animal cool, particularly if longer procedures are anticipated. It is also advisable to keep handling procedures as brief as possible and, if practical, try to schedule captures in the early morning when the ambient temperature is cooler.

A member of the technical support team should be trained to record respiration times – the time will allow the recorder to inform appropriate staff when an inordinately long time has passed since the last breath – and heart rate, although the latter may be difficult to monitor depending on the species and blubber thickness. Ideally, vital signs for an animal should be recorded continuously over the entire span of the event (e.g. every 5-10mins during each hour, or preferably, continuously) with directions to notify the veterinarian of any changes in that animal's baseline rates. Subtle changes may be difficult to recognise, so it may be necessary to rely more on trends over time or other indicators (e.g. behaviour, posture, alertness, reflexes, analysis of blood sample in the field if practical) to ensure the animal's status is not deteriorating.

STRESS RESPONSE MONITORING

A stress response brought about by chase and capture has been shown to trigger changes in the hematological and plasma chemical constituents of some cetaceans (St. Aubin and Geraci, 1988; 1989). These physiological imbalances may impair an animal's immune system, rendering them more susceptible to pathogenic organisms that otherwise might not pose a threat. Changes in blood constituents were noted in two white whales caught for tagging and then recaptured several days (19 and 24) later for removal of data loggers (St Aubin *et al.*, 2001). Both animals showed evidence of white blood cell responses consistent with inflammation and stress. Since the interval between chase and collection was the same for the two whales, the observed hematological changes were assumed to be due to tissue damage and repair from satellite tag application. The response of blood constituents to handling and tagging operations suggests that these procedures represented an immune system challenge under these conditions.

Behavioural assessments may be used in the field to recognise acute stress. Anxiety is one of the most common manifestations presented by animals under stress, although passivity may also be a sign as well as increased respirations (Hanson *et al.*, 1998). It is prudent to try to minimise the chase duration and expedite handling procedures to prevent acute intense or prolonged stress. Although a suspected case of capture myopathy was reported by Colgrove (1978), it is rarely encountered in cetaceans (Schroeder *et al.*, 1985a).

Individuals resighted or recaptured after several months or years appear to be completely healed (Orr *et al.*, 1998) suggesting that for at least some individuals, handling activities do not severely compromise survivability. However, no direct comprehensive, controlled studies on cetaceans have been conducted to determine if survivability has been compromised to some extent. Long-term survivability and reproduction of bottlenose dolphins studied in Sarasota Bay, Florida, for more than 30 years does not appear to have been compromised due to capture-release techniques utilised in that project, given that more than 40% of the dolphins first tagged in 1970-1971 were still observed more than 30 years later, and the population size of the resident dolphin community has increased significantly during this period (Wells, 1991; 2003). Furthermore, health assessment and monitoring of four generations of resident bottlenose dolphins have not found any complications from capture-release. Blood sampling of individuals before (e.g. as soon after capture as practical) and several days after tagging would allow for evaluation of the physiological impact of these activities on stress indicators in the blood. This would require more sample handling but is of great research value. Some other cetacean species, however, may not be as resilient or as easily handled as bottlenose dolphins.

Treating shock and allergic reactions

The use of medications to treat shock, potential infection or allergic reactions is rarely needed. They may be used under conditions where quick release is not possible and there will be an extended period of time before the animal can be returned to the environment. Corticosteroids may be used to treat shock during which time an animal's condition may be declining rapidly (i.e. body temperature < 35°C, respiratory rate > 8bpm). Epinephrine can be used for adverse or allergic reactions to other medications administered such as antibiotics. Signs of an allergic reaction may include: agitation, increased heart rate, difficulty in obtaining blood samples due to circulatory collapse, possible swelling at an injection site or of soft tissues of the head. Once treated, the tagging team should support the animal until it is stable (e.g. respiratory rate and body temperature have normalised), after which tagging procedures should be aborted and the animal released. If it is not feasible to have a veterinarian available during tagging procedures, it may be logistically difficult or impossible to obtain timely veterinary support in an emergency situation at a remote tagging site.

POSSIBLE COMPLICATIONS FROM TAG PLACEMENT

Tag attachment systems range in impact from suction cups, to surgical implantation of attachment pins or the entire tag, to attachment of the tag by barbed spear. Except for the suction cup, each of these involves piercing of the skin and blubber and possibly other structures as well. Health considerations include maintenance of aseptic conditions at the tag placement site during deployment, preventing introduction of microorganisms into the pin tract, preventing pressure necrosis by the tag and its pins, minimising tissue reaction to the tag materials with subsequent tag rejection and promoting wound healing (see Irvine *et al.*, 1982; Scott *et al.*, 1990; Wells, 2002).

Dermatological effects of tag attachment

Several studies have characterised the rate of cutaneous wound healing in bottlenose dolphins (Brown *et al.*, 1983; Bruce-Allen and Geraci, 1985; Geraci and Smith, 1990) and white whales (Geraci and Bruce-Allen, 1987; Geraci and Smith, 1990). Cutaneous wound healing was found to take place at a quicker rate in bottlenose dolphins than in white whales. Geraci and Smith (1990) stated that two principal factors decrease the life of an implant: infection and movement in the tissue. Infection may be minimised by including a slow-release, broad-spectrum antibiotic in the implant and preventing contamination while embedding the implant. Movement of a tag or attachment pin or spear within a tissue can be decreased by careful engineering of the attachment to distribute pressure and minimise jerking due to variation in hydrodynamic drag such as during exit and re-entry of the tag during a breathing cycle. With an implanted tag, this can be accomplished by embedding the head of the tag deeply and with a sufficient number of anchors to stabilise the implant within the tissue. Much is still unknown about the effects of epidermal thickness, ambient temperature, salinity and stress on cutaneous wound healing. These factors could have implications with regard to healing of wounds caused by placement of transmitter devices.

Steps to help minimise implant rejection during attachments using surgically implanted pins

A suggested procedure for skin preparation, bolt placement and tissue boring is described below. This may not be feasible in all cases, but before a researcher chooses to simplify the suggested procedures, careful thought should be given to weighing the importance of speed and convenience of the tagging process against the increased risk to the animal. As noted earlier, an animal compromised during handling may not behave in a manner representative of the population, thus providing data that are possibly misleading and wasting a research opportunity.

Before placing a tag, a site must be chosen that is reasonably devoid of blood vessels so there is minimal impact to heat-exchanger vessels and the general blood supply of the area (Lander *et al.*, 2001). To accomplish this, the dorsal fin (or ridge, if working with a finless species such as white whales) of a dead specimen should be examined. It is recommended to use a sterile hypodermic needle to probe anticipated pin sites to determine if major blood vessels, particularly arteries, are present before actual pin placement on a live animal (Chilvers *et al.*, 2001; Lander *et al.*, 2001).

Prior to any puncture or pin placement, the skin surface should be cleaned properly with an antiseptic such as isopropyl alcohol or 10% Povidone-Iodine solution (Poviderm Solution, Vetus c/o Burns Veterinary Supply, Inc., Dallas, Texas)[1] (Westgate *et al.*, 1998), then the area should be surgically scrubbed twice. This technique involves starting at the point of pin placement and, using small circular scrubbing movements, working circumferentially to the periphery, being careful not to touch the centre of the scrubbed area with the same gauze sponge once it has reached the outer limits of the sterile field. This will prevent contamination of the sterile field's centre with organisms from the periphery. Upon completion of the first scrub, a second should be completed in the same manner. In a field situation, a researcher may find it challenging to maintain sterility of the pin placement site while working with a live animal in a less than ideal environmental setting (e.g. seawater washing over the sides of the boat or deck).

The area of pin placement is then locally anesthetised with lidocaine HCl 2% with epinephrine 1:100,000 using a 21 or 22-gauge needle. Some individuals have found this to be more effectively accomplished using an injector needle gun (Miltex Inc., Bethpage, New York[1]). In larger species such as the killer whale, difficulty may be encountered trying to inject directly into the tough dermis. Alternatively, the anesthetic agent may be injected into the base of the dorsal fin using a 2-inch 19-gauge needle so that the anesthetic may be drawn up the fin from the injection site. Following placement of the local anesthetic, another surgical scrub of the area is performed following the same technique as for the first two. All equipment (tags, pins, bolts) is cleaned with

[1] Use of trade names does not imply endorsement by the National Marine Fisheries Service.

isopropyl alcohol prior to utilisation. Holes for pins are typically established using specialised hole cutters, similar to a laboratory cork borer, that have been cleaned and disinfected prior to each use. The borehole should be made slightly smaller in diameter than the pin to be used to reduce bleeding and loosening at the tag site and to prevent disruption of the healing process (Hanson, 2001). If nuts are used on the ends of the pins to secure the transmitter package, one must be cautious not to overtighten the nuts as compression and ischemic necrosis may occur at the attachment site. A small amount of triple antibiotic ointment (i.e. polymixin B-bacitracin-neomycin) may be applied to the tag entry point. Injections of antibiotics are not routinely given after tag placement unless the animal has sustained numerous abrasions/lacerations during capture.

Behavioural effects of tag placement

There are reports of telemetry devices leading to changes in behaviour of an animal subject through increased drag or discomfort of the attached instrument package (Irvine *et al.*, 1982; Tanaka, 1987; Tanaka *et al.*, 1987; Scott *et al.*, 1990). Increases in drag from 12-15% to as much as 27% are reported for a dorsal fin mount tag on a harbour porpoise model (Hanson, 1998). Würsig (1982) found that radio-tagged dusky dolphins (*Lagenorhynchus obscurus*) were not bothered by the transmitters, but seemed to swim slower than normal after two or more days post-tagging. Foraging and attendance behaviour of female Antarctic fur seals (*Arctocephalus gazella*) was altered with instrumented animals, but the biological significance and long-term effects of telemetry devices is unclear (Walker and Boveng, 1995; Boyd *et al.*, 1997). Dorsally attached transmitters did not seem to affect behaviour in white whales (Richard *et al.*, 1997).

POTENTIAL FOR DISEASE TRANSMISSION

Although there are few reports of disease transmission between marine mammals and humans (Geraci and Ridgway, 1991), one must always be aware of the possibility of being exposed to new diseases (e.g. Buck and Schroeder, 1990; Tryland, 2000). Many bacteria are shared by humans and cetaceans, and some cause disease in both, however, there is not an excessive health risk to humans from association with cetaceans. Early studies (Johnston and Fung, 1969) suggested humans could be a source of infection to cetaceans in a captive environment, but it is not certain if the same risk exists during handling of wild animals. Several species of bacteria have been recovered from stranded cetaceans that have been associated with a variety of infections in humans (Buck, 1984). Similarly, marine mammals harbour microflora that are commensal and usually pose no health threat under normal circumstances. Under conditions of stress the animal may be debilitated or immunosuppressed and be predisposed to infection by these organisms or others encountered in the environment. Exposed mucous membranes and cut skin surfaces are especially prone to potential pathogens. The same may apply to humans handling the animals.

Most bacteria associated with marine mammals are not a public health concern. More thorough discussions of potentially zoonotic diseases between humans and marine mammals are available in Geraci and Ridgway (1991), Higgins (2000) and Cowan *et al.* (2001), however, a few warrant special mention here since they are more commonly recognised pathogens of humans (Schroeder *et al.*, 1985b; Suer *et al.*, 1988; Palmer *et al.*, 1991). These organisms could be infectious for persons with compromised immune function or could be inoculated into cuts, bites or abrasions. Infections with *Brucella* sp. have been reported in cetaceans (Ross *et al.*, 1996; Miller *et al.*, 1999). Although there have only been two published reports of a marine *Brucella* isolate infecting a human (Brew *et al.*, 1999; Sohn *et al.*, 2003), one should recognise its zoonotic potential. *Mycobacterium marinum* has been transmitted to a human via a dolphin bite on a finger and thus should be considered zoonotic (Flowers, 1970). Blastomycosis infection of a veterinarian's hand followed examination of an infected bottlenose dolphin (Cates *et al.*, 1986). *Erysipelothrix rhusiopathiae* is a pathogen that causes cutaneous infarcts or generalised septicemia in many species of cetaceans and contributes to pain, swelling and/or more generalised illness in humans (Medway, 1980). This pathogen was once thought to be the cause of seal finger, but subsequent studies have implicated a mycoplasma (Stadtlander and Madoff, 1994). Marine morbilliviruses have not shown to be infectious to humans (Cowan *et al.*, 2001).

Steps to minimise exposure

Personnel handling cetaceans must exercise caution to diminish exposure to potentially hazardous microorganisms. Care should be exercised when working around the head region of cetaceans. One should avoid being in the path of the blowhole exhalation due to the risk of exposure to microorganisms, particularly those that are known to be human pathogens. Cases of such transmission have not been demonstrated, nevertheless, the potential still warrants mention. Surgical masks may be worn if handlers will potentially be exposed directly in the face to a cetacean's breath or bodily fluids. However, this is often not a realistic option in the field setting especially during activities such as capture and release. Exposed mucous membranes and cut or abraded skin surfaces are especially prone to potential pathogens. Gloves and other gear such as strong, heavy footwear should be worn to protect against abrasions, cuts and bites. Personnel should avoid touching their eyes and face during handling to minimise transmission of an organism to mucous membranes. Handlers should thoroughly wash their hands in disinfectant between and after handling animals. Immunocompromised or pregnant field staff should avoid direct exposure to cetaceans.

In the very rare instances of infection acquired from a cetacean, the infection may begin subtly following an encounter with the animal. If an infectious condition is suspected, the handler should present the history of contact with a marine mammal to the physician, as the clinician may not think to ask about such an exposure.

CONCLUSION

Anecdotal evidence suggests that most capturing and tagging of odontocetes occurs without incident to either the animal or the handler. Handlers must be aware, however, of the potential health risks to both cetaceans and humans. These risks will be reduced by including knowledgeable, experienced personnel in all aspects of the project, keeping handling times to the minimum necessary to safely complete the objective, careful planning which includes contingencies for potential problems, having adequate personnel and equipment on hand to meet the contingencies, as well as maintaining vigilance when monitoring the animal's condition throughout the handling process. It is important to use caution when working around the animal in order to avoid cuts, abrasions and other wounds that might facilitate the transfer of a potential pathogen between cetaceans and humans. Care in handling procedures will result in fewer lost animals and more reliable data.

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