COURSE CODE: **VCM 602**

COURSE TITLE: **Wildlife and Fisheries Management and Medicine**

NUMBER OF UNITS:

COURSE DURATION:

COURSE DETAILS:

COURSE DETAILS:

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COURSE CONTENT:

Dynamics and characteristics of wildlife populations. Feeding of wild animals in captivity. Capturing and sampling methods. Diagnosis and treatment o major diseases of wild primates carnivores, ungulates, reptiles, birds and amphibians. Aquatic ecology, fisheries management and fish cultures. Feeds and feeding of fish. Fish equipment and methods. Diagnosis, treatment and control of diseases of fish.

COURSE REQUIREMENTS:

READING LIST:

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LECTURE NOTES

**DYNAMICS AND CHARACTERISTICS OF WILDLIFE POPULATION**

Wild animals are animals running unrestrained in a natural environment. They include (wild mammals, birds, reptiles and amphibians). Zoo animals are wild animals kept in close or open confinement, usually for public viewing.

“Wildlife” often includes only vertebrates, particularly popular species such as those that are hunted, trapped, cause problems, or are endangered.  Vertebrates are animals with backbones, including birds, mammals, fishes, amphibians, and reptiles.  A more expansive definition of wildlife would include all animal life-forms in an ecosystem.

The term *"game species"* refers to an animal that is either hunted or trapped

*Non-game species"* are all the other animals.

*"Endangered and threatened species"* are a special group of non-game species whose populations are low.

An *"extinct"* species is one that can no longer be found *anywhere* in the world.  An *"extirpated"* species is one that no longer occurs in a place (such as a state or region) where it once used to. In developed countries, populations of many game species are directly managed through hunting and trapping regulations.

Habitat for some endangered and threatened species is sometimes managed exclusively for that species.  In other cases, restrictions on activities are prescribed by law, such as the prohibition of certain practices during a certain time period (usually breeding season), within a stated distance or area.  Rather than direct management of a species population, *habitat* is managed for as much diversity as possible, with the explicit assumption that by providing as many alternatives as possible, each species of wildlife will find what it needs to maintain a viable population.

Habitat features can be expressed in five categories:

* Site Quality
* Space and Home Range
* Food & Water
* Shelter
* Variability

*Site quality"* incorporates factors such as soil, topography, climate extremes, precipitation, and drought frequency.

Every species has a minimum *"space"* requirement.  Space is needed to obtain life's necessities.

*Home range* is the area within which an animal will feel comfortable, and is some cases, actively defend

*Food and water* are obvious needs of every living thing.  During the course of a year, availability and quality of food and water can change dramatically.  Wildlife will often migrate to avoid lean times.  Others might hibernate or undergo other metabolic changes.

*Shelter"* is needed for a variety of purposes.  The first to come to mind is protection against adverse weather.  Shelter is also needed to escape predators. Shelter or specific habitat conditions are also needed for courtship displays, nesting, rearing young etc

*Variability* of habitat quality and habitat needs makes management difficult, as discussed above.

Wildlife needs vary with the season and life stage of a species.  Additionally, all species have preferred habitat and minimum habitat conditions. A species that is flexible in its habitat adaptability is sometimes called a *"generalist"*.   A species with a rather narrow and specific range of requirements may experience severe population fluctuations with changes in the environment.  These species are referred to as *"specialists".*  Species that are very sensitive to certain environmental changes are sometimes used as *"indicator species*

**Wildlife Population**

A wildlife population is a group of individuals of the same species that have some basis of commonality.  Populations can be linked to a feature in the landscape, to other populations, a time period, or other criteria.

Wildlife populations have inherent characteristics that help in defining the welfare of various species. Including:

* Age Structure
* Lifespan
* Sex Ratio
* Natality & Mortality
* Interspecific Dynamics
* Intraspecific Dynamics
* Territoriality & Home Range
* Migrations
* Carrying Capacity

**Age Structure**:  There should be some kind of balance among the classes and the "proper" balance will vary by species and season.   Generally, the age structure can be depicted by a triangle, with the numerous young on the bottom and the very few oldsters at the top. Balance is usually affected by hunting/fishing pressure.

**Lifespan**:  Obviously, different species have different life spans. Species toward the end of food chains are usually much longer-lived that those in the beginning however, Long-lived species have strategies that favor the survival of fewer individuals.

**Sex Ratio**:  Each species has an "ideal" sex ratio.   Usually this is somewhere around 50:50. A particular sex ratio will help maximize *"fecundity"*, or the ability of a species to produce new individuals.

**Natality and Mortality**:  Natality is the inherent ability of a population to increase in numbers.  Mortality deals with the level of death within a population.   These terms are usually expressed as *rates* that reflect pressures to increase and decrease population size. Affected by litter size, disease conditions.

**Inter-specific Dynamics**:  These are relationships *among* or *between* species e.g., *predator-prey*. Interspecific dynamics can be antagonistic or beneficial.  *"Mutualistic"* relationship is one working in concert to the benefit of both.  A *"commensal"* relationship is where one species requires another, but the host is relatively unaffected.    Another kind of interspecific relationship would be *parasitic*.

**Intra-specific Dynamics**:  There are relationships among individuals of a population.   Competition for food, shelter, and other requirements are common examples.   Mating and establishing territories are other examples.  A species might be colonial in nature or live primarily as individuals.

**Territoriality and Home Range**:  An individual or population of a species may actively mark and/or defend a particular area.

**Migration**:  Winters and dry seasons result in less available food and water.  Animals have a wide range of strategies to accommodate these seasonal fluctuations.  Migration is one such strategy.

**Carrying Capacity**:  The physical and biological resources of an area, varying with the season, will support only so many individuals.  This maximum amount called the *"carrying capacity"*.  When most species approach their particular carrying capacity, mortality factors overtake natality factors and the population growth declines.

**TRANSPORTATION, HANDLING, RESTRAINT, CARE** **AND FEEDING OF WILD ANIMALS IN CAPTIVITY**

**1. INTRODUCTION**

The management of these wild animals may require their capture and handling for the application of marking devices, the collection of genetic material, the identification of specific characteristics and collection into zoological gardens.

When handling wild animals, you should:

1. Be familiar with the terrain, correctly identify the species to be captured and seek the advice of experienced colleagues especially on its response to disturbance, its sensitivity to capture and restraint, and its requirements for captive maintenance, if it is to be held for any length of time. Do not allow unsupervised, inexperienced persons to handle any animal species until adequately trained to restrain, manipulate and release the animals properly. You should consult the current literature and experienced peers before handling an unfamiliar species.
2. Uphold all regulations pertaining to the species with which you are working.
3. Ensure the safety and welfare of the animals you study and handle, and treat all study animals with care and respect. They should be as healthy and free of trauma as possible.
4. Avoid or minimize distress and pain and reduce risks of injury or death to the study animal(s).
5. If they occur, promptly treat all injuries to wild animals in the most appropriate and humane manner.
6. Train and supervise all assistants to follow the same ethics and standards.
7. Special consideration should be given when study animals have dependent young. As a general principle, the removal or disturbance of animals from the wild during sensitive periods, such as breeding and egg-laying periods, should be avoided, unless justified for scientific reasons.
8. When wild animals are being trapped or collected, they should not be exposed to excessive or extended handling, aggression, predation, adverse weather, or temperature extremes or undue suffering. Trapping and handling equipment should be routinely inspected, maintained and repaired as necessary. Traps should be monitored as often as is considered appropriate for each trap type and species involved. At the end of each collecting period traps should be properly closed or removed.
9. The living conditions of captive animals should maintain them in an adequate state of health and well-being. Captive conditions should satisfy the standards of hygiene, nutrition, group composition and numbers, refuge-provision, and protection from environmental stress.
10. Be aware of, and prepare to avoid, the potential risks of a variety of transmissible diseases and parasites to other animals and humans, as well as other hazards associated with the handling of wild vertebrates.

**2. COMPLIANCE WITH LAWS AND REGULATIONS**

Regardless of nationality or location, you should be aware that there are local, state/provincial, federal/national, or international laws or regulations that pertain to activities involving wild animals. Ignorance of the law or even inadvertent violation of regulations may result in prosecution.

Have full knowledge of all local, provincial and federal regulations pertaining to the animals under study, and must obtain all necessary permits that are required. Be aware that regulations may vary with each country and must ensure that you comply with all wildlife regulations of the country in which you are working. You must obtain and comply with all permits required for the capture, handling and collection of the correct species and in the appropriate jurisdiction. In addition, be familiar with the current list of threatened and endangered species and must comply with all rules and regulations pertaining to these and all other categories of wild animals.

For example, researchers based in or conducting research in the United States must obtain permits issued by various federal agencies for the following purposes:

1) to import or export specimens of non-endangered species through a non-designated port of entry;

2) to import or export endangered wildlife through any port;

3) to import injurious wildlife;

4) to import, export, ship interstate, take, or possess endangered species or parts thereof for research or propagation;

5) to take, harass, possess, or transport marine mammals;

6) to import or transfer etiological agents or vectors of human disease and living non-human primates;

7) to collect scientific specimens on national wildlife refuges;

8) to import ruminants and swine, including parts, products, and by-products; and

9) to import organisms or vectors, tissue cultures, cell lines, blood, and serum.

You must be familiar with the current list of mammalian species deemed threatened or endangered and must comply with all rules and regulations pertaining to capture of these and all other categories of mammals. A list of threatened or endangered species/subspecies under the United States Endangered Species Act is available from the Office of Endangered Species, United States Department of the Interior, Fish and Wildlife Service, Washington, DC 20240.

Many institutions, as well as state, provincial and federal governments, have regulations or recommendations concerning the handling and sampling of rodents that may be carriers of serious human diseases. You must ensure your safety and that of your employees or students by understanding the disease carrying potential of wild animals, by taking appropriate safety precautions, and by complying with appropriate regulations.

**3. METHODS OF EUTHANASIA**

When wild animals are injured or distressed and cannot be used after capture, they must be euthanized humanely. Field methods used to euthanize mammals should be quick, as painless as possible, and compatible with the behavior of the species.

Acceptable methods of euthanasia vary among species but typically are related to size of the animal. Use of inhalants such as carbon dioxide, halothane, methoxyflurane, ether (carcinogen, flammable and potentially explosive), or other gases (except chloroform, which is not recommended) for euthanasia is acceptable, but sometimes is impractical under field conditions. Under open-air field conditions, chloroform may be appropriate due to the fact that it also kills ectoparasites, that may pose greater risks to the researcher through transmission of diseases such as plague and typhus. If chloroform is used, it always should be outside in well-ventilated areas and by experienced personnel. For euthanizing small mammals, cervical dislocation and thoracic compression are commonly used methods because they are quick and impart little pain, thus meeting the criteria for euthanasia methods.

Euthanasia by shooting or other traumatic means also is humane and effective if the result is instantaneous death, but should not be employed except by experienced investigators. Regardless of method used, death of the animal should be confirmed.

**4. RESTRAINT AND HANDLING**

Most of the live capture techniques utilize bait, decoys, recorded calls or lures to attract animals to trapping sites. The health and well-being of animals should be the primary concern. Wild animal capture is an activity that is strictly controlled in most countries; permits are usually required in developed countries.

The nature of restraint will depend upon the animal species involved. Restraint techniques can range from confinement in an enclosure through various types of physical restriction, to chemical immobilization. Any decision to use physical or chemical restraint should be based upon an understanding of the behavioural and physical characteristics of the species to be restrained, the field conditions under which the procedure will occur, the knowledge and skill of those persons handling the animals, and the availability of appropriate equipment and facilities. You must use the least restraint that is necessary to do the job in a humane and effective manner, with least stress to the animal.

**4.1. GENERAL PRINCIPLES OF PHYSICAL RESTRAINT**

Because many species of wild animals are capable of inflicting serious injury to themselves or those handling them, some form of restraint is usually necessary. The well-being of the animal under study is of paramount importance and it must be emphasized that improper restraint, especially of frightened or stressed animals, can lead to major physiological disturbances, including hypothermia, hyperthermia, stress, shock and capture myopathy. Many species of wild animals do not tolerate physical restraint and in some cases there is a great potential for animal or handler injury. The following are general guidelines that must be considered when you want to physically restrain a wild species:

1. Wild animals should be handled quickly and without sudden movements, utilizing the minimum number of personnel that are required to safely and efficiently perform the task.
2. Darkened chambers and/or blindfolds alleviate stress and subdue animals. They should be used whenever possible. Excessive noise from loud equipment, vehicles, or talking should be minimized. In addition, the handlers should be aware of the negative responses wild animals may have to touching of any kind.
3. Excessive struggling or stress in the restrained animal can lead to hyperthermia and muscle damage (capture myopathy), especially during warm or hot conditions. In some cases the time of day will also be an important consideration with handling efforts focused during cooler periods (dawn/dusk). When birds are restrained by hand, the hold must include the wings and legs in order to prevent damage to these appendages. Certain species may have specific requirements for physical restraint, including those with long legs and necks. Birds breathe by a bellows-like action of the ribs and sternum. Therefore, care should be taken so that the method of restraint does not interfere with the ventilatory movements of the sternum or impede the respiratory air flow. Birds that are allowed to struggle excessively can potentially injure handlers, injure themselves and/or become hyperthermic.
4. The time of year can be an important consideration when handling and restraining wild animals. For example, bison and elk tend to be less aggressive and more easily handled in the wintertime. Many animals may be more readily baited into traps and holding areas when the natural conditions are at their poorest.
5. If possible, avoid capturing and restraining animals which are pregnant, tending young or breeding.
6. When restraining an animal by hand, the force applied and technique should be appropriate for the species in question.
7. If muzzles, hoods or holding bags are being used as part of the restraint, you must ensure that the animal's breathing or thermoregulatory ability are not compromised.
8. The mesh size and construction of nets must ensure that the animal cannot force its head through the mesh or easily chew through the net material.
9. Most amphibians and reptiles are relatively small and slow moving, and can be restrained by hand or in a net. However, many small species are easily injured if the handler uses excessive force. Tail autotomy (tail shedding) can occur in most lizards if they are restrained by the tail. Although this is not a serious injury, it will influence future growth and reproduction by depriving the animal of fat stores as well as the integrity of the specimen. Tail loss may also affect the behaviour of the animal. Because some reptiles may struggle excessively when manually restrained, the use of nets, hooks, tongs or handling bags may be required to reduce injury.

For venomous snakes and some large turtles which are potentially dangerous and require special methods of restraint, adherence to the following general guidelines is recommended when working with hazardous reptiles:

1. Procedures chosen should minimize the amount of handling time required, and reduce or eliminate the contact between handler and animal.
2. Those handling dangerous species should not work alone. A second person knowledgeable in capture/handling techniques and emergency measures should be present whenever possible.
3. Only experienced personnel should handle venomous snakes. They should be familiar with standard emergency procedures that are to be initiated in the event of an accidental bite or contact. A treatment protocol and a supply of the appropriate antivenin should be available at all times. In addition, a physician or medical facility should be made aware of the nature of the studies being undertaken so that proper arrangements can be made for emergency care and examination.
4. Whenever possible, an anaesthetic and/or physical restraint should be used before physical contact with the specimen.

Prolonged, distressful restraint should not occur. Administration of a tranquillizer or sedative to an animal that is physically restrained for longer periods of time may help to prevent injury to both the animal and the handlers. In some circumstances, it is advisable to use general anaesthesia for restraint in the field, particularly for larger or dangerous species. Invasive procedures may require some form of physical restraint initially, but usually require subsequent analgesia and/or chemical immobilization.

**4.2. GENERAL PRINCIPLES OF CHEMICAL RESTRAINT**

The administration of anaesthetics to wild animals for restraint purposes can be accomplished safely; however, the use of chemicals do present risks to both animal and man. Field immobilization is almost always performed in less than ideal conditions. Only rarely can you examine animals prior to anaesthesia, give accurate dosages in a controlled environment, and intensively monitor animals during anaesthesia events.

Although some wildlife projects may involve the use of oral or intravenous agents, most field situations utilizing chemical immobilization require the intramuscular administration of drugs. In some cases these are administered with a hand-held or jab-stick syringe to an animal which is physically restrained or confined. In other instances the drugs are given remotely with a projected syringe or dart. Drugs administered by projectiles can seriously wound or kill the target animal if a vital organ, a major blood vessel or a non-target area of the body is penetrated. Therefore, heavily muscled areas must be targeted when darting wild animals.

Every anaesthetic agent has specific advantages and disadvantages, and there is no single agent that is suitable for the chemical immobilization of all wild species under all circumstances. Safe and effective drug dosages will vary with the species, age, sex and body condition of the animal. In addition, there can be individual and seasonal variations in the response to agents. It should also be realized that drugs used for wildlife immobilization have the potential to seriously affect both animals and humans involved.

The effects of drugs on many avian species have not been determined. When information concerning the effect of an anaesthetic drug on the study species is unavailable, it is recommended that pre-experimental testing using low dosages of the drug is initiated under the supervision of a veterinarian experienced in avian anaesthesia.

You should consider the following when using chemical restraint in birds:

1. Birds tend to have a higher metabolic rate and oxygen consumption relative to mammals. Therefore, birds may have a greater requirement for oxygen supplement and assisted respiration than mammals.
2. Birds have far less functional residual capacity than mammals and therefore, apnea (cessation of breathing), will result in death far more quickly.
3. The avian respiratory system, which consists of a pair of relatively fixed lungs and a group of mobile air sacs, is more efficient at gas exchange than mammals. Therefore, birds will often demonstrate a more rapid response to the effects of inhaled anaesthetics.
4. Inhalation anaesthesia, specifically isoflurane, is presently considered to be the method of choice for most procedures which require general anaesthesia in birds.
5. Because of the large volume of stored gases in air sacs, birds can be inefficient at eliminating inhaled anaesthetics. Recovery from anaesthesia can be facilitated by maintaining the bird in lateral recumbency and turning it every few minutes.
6. In general, injectable anaesthetic agents are a poor choice in birds and are used with limited success. Many agents have an unpredictable duration, a rough and prolonged recovery period and serious metabolic effects when used in birds. Birds have a renal portal system and therefore agents injected into the legs may be excreted or metabolized before reaching the systemic circulation. This may act to increase the variability of response to injectable agents.
7. Debilitated or stressed birds are very susceptible to the effects of hypoglycemia which can complicate an anaesthetic procedure.
8. Birds have a high body surface to volume ratio and this will act to exacerbate hypothermia during an anaesthetic procedure. Surgery and recovery areas should be sufficiently warm to counteract heat losses.

The most reliable indicator of depth of anaesthesia in birds is respiratory rate and character. Heart rate varies inversely with the size of the bird and should also be monitored closely.

**5. TRANSPORT**

It may be necessary to transport animals to move them from capture sites to holding facilities. Transport containers and methods of shipping animals will vary widely from species to species. The live traps that are used for capture are usually adequate for transport over short distances. However, if the animals are large or are to be confined for a longer period, these traps may not be suitable. They must be placed in appropriate holding cages, which can include live traps if those traps are provided with adequate ventilation, food, and a source of moisture, and if they encompass sufficient space with appropriate padding and bedding to ensure the comfort of captive mammals. Live traps also should be positioned to permit drainage of urine produced by captive animals. In general, the containers used in transportation must protect the occupants from injury and allow the individual sufficient space so that it can assume a normal posture and engage in comfort and maintenance activities unimpeded. In most cases, animals should be separated. Mammals in transport should never be subjected to thermal environments that exceed their limits of tolerance. Cages for transporting mammals should be kept out of the sun, wind, and precipitation and at a comfortable temperature. Captives should be checked frequently. The precautions used for the humane transport of household pets should be applied when transporting wild animals. Care also should be taken to minimize psychological stress on certain species by shielding cages from excessive light, noise, and human activities.

In birds, space sufficient to permit flight is not usually advisable because the chances of injury are increased. In addition, it may be necessary to restrain the wings of larger species. Containers should be padded in those instances where excitable animals or species with delicate bone structures are to be shipped. Adequate ventilation must be provided. For longer journeys, water and food should be provided. The inside of containers should be as dark as possible, while still allowing them good ventilation, to find food or water, and to move about. It is recommended that transport vehicles be equipped so that the transported animals are not exposed to excessive noise, movement or temperature extremes. Proper arrangements should be made to ensure that animals arrive at destinations during normal working hours, rather than on weekends or holidays.

**6. MAINTENANCE OF WILD ANIMALS IN CAPTIVITY**

Cages or enclosures to hold wild animals and their offspring should be designed to accommodate salient features of their ecology, morphology, physiology, and behavior. To house certain species (e.g. desert granivores, shrews, and fossorial species such as moles) under conditions prescribed for laboratory rodents is not in the best interest of such species and may amount to inhumane treatment. Desert granivores need fine sand for dust-bathing and caching of seeds. Burrowing species require soil or other suitable substrate in which to construct tunnels.

Methods useful for maintaining mammals that have been bred in captivity for many generations may not be appropriate for wild-caught mammals. For example, allowance should be made for less-frequent cage cleaning and inclusion of more objects (e.g. materials for nest construction and play) in many wild species. Although basic cleanliness and hygiene remain a high priority, wild mammals should be disturbed less often and allowed to accumulate familiar odors, which are important to species that are olfactorily oriented. Furthermore, mammals that are hibernating require different caging and housing than the same individuals when not hibernating. Particularly important is the need to maintain sufficiently high humidity levels and to keep temperatures at optimal levels to minimize energy expenditures. In some cases, this may involve keeping ambient temperatures within only a few degrees of freezing, depending on the thermal optimum during hibernation for each species.

**7. CARE AND FEEDING OF WILD ANIMALS IN CAPTIVITY: TERM PAPER**

|  |  |  |  |
| --- | --- | --- | --- |
| **S/N** | **NAME** | **MATRIC NO.** | **ANIMAL** |
| 1. |  |  | Baboons |
| 2. |  |  | Gorillas |
| 3. |  |  | Apes |
| 4. |  |  | Giraffe |
| 5. |  |  | Wild birds |
| 6. |  |  | African Wild dog |
| 7. |  |  | Antelopes |
| 8. |  |  | Snakes |
| 9. |  |  | Spotted Hyena |
| 10. |  |  | Tortoise & Turtle |
| 11. |  |  | Elephant |
| 12. |  |  | Zebra |
| 13. |  |  | Crocodile & Alligator |
| 14. |  |  | Rhinoceros |
| 15. |  |  | Leopard |
| 16. |  |  | Jaguar |
| 17. |  |  | Lion |
| 18. |  |  | African Buffalo |
| 19. |  |  | Warthog |
| 20. |  |  | Hippopotamus |
| 21. |  |  | Jackal |
| 22. |  |  | Wildebeest |
| 23. |  |  | Ostrich |

**8. PERSONNEL SAFETY AND HEALTH PRECAUTIONS**

**8.1. General Precautions**

All wild animals are potentially dangerous either from traumatic injury due to direct contact or from infectious diseases that are carried by mammals or their parasites. Therefore, when dealing with wild-caught animals, you should work under the assumption that the animals pose some risk to your health and safety. The risk can be substantially reduced by common sense and good personal hygiene (e.g. wash hands often with soap and water). Minimize the chances of being bitten or scratched (e.g. wear leather or fabric gloves) and use latex gloves to prevent unnecessary exposure to blood or other body fluids and faeces, which may contain parasites or pathogens that affect humans. It is important that all humans protect themselves against possible injury or exposure to potentially dangerous procedures, chemicals, animals, or animal fluids and waste. They must ensure that adequate protective measures are implemented for the humans involved during the capture of wild animals. In addition, you must ensure that all workers fully understand the techniques to be used for restraint and handling; get familiarized with known biohazards specific to the species under study and with the methods to avoid transmission of zoonotic diseases and parasites.

**8.2. Drugs and Chemicals**

All drugs and chemicals used in field research should be handled in such a way as to prevent human exposure. You should protect yourself against both respiratory and cutaneous exposure to drugs and chemicals as well as accidental injection. Those utilizing immobilization drugs for restraint of wild animals must have the appropriate training and information available to aid in their medical care should accidental contamination occur.

**8.3. Zoonotic Diseases and Parasites**

You are at risk of exposure to zoonotic diseases, or those diseases and parasites transmitted from animals to humans. The degree of risk varies with the species of animal to be studied, the degree of exposure and the organisms present. Take precautions to avoid exposure to external animal parasites such as ticks, fleas, as well as to animal faeces which may contain internal animal parasite ova or larvae infective to humans.

Wild animals captured and held temporarily in cages and traps, and animals brought into holding facilities should be examined and treated for external and internal parasites which may be transmissible to other animals or humans, or can transmit infectious diseases.

Infectious organisms may be present in wild vertebrates which are a potential hazard to humans (e.g. the discovery of hantaviruses in North America). Therefore, any unusual symptoms observed in investigators, students, or technicians who handle wild vertebrates should immediately be reported to medical authorities knowledgeable about the diseases and parasites associated with wild animals.

In addition, you should recognize the risks of contracting diseases that are associated with direct contact with mammals or their parasites. For example, bubonic plague is caused by a bacterium that can be transmitted to humans by fleas that occur on certain rodents, especially sciurids (squirrels), or indirectly by close contact with certain carnivores (e.g. domestic cats). Tularemia is a bacterial disease, primarily of lagomorphs (hares and rabbits), that can be transmitted to humans by arthropods or by handling or eating infected animals. Mammals may also serve as reservoirs for numerous other agents such as relapsing fever, murine typhus, salmonellosis, histoplasmosis, toxoplasmosis, leptospirosis, and pasteurella. The list of pathogens that humans can acquire directly or indirectly from mammals continues to grow, principally because new technologies have become available to detect them.

1. All animal tissues, fluids and excrement should be handled so that the potential for human contact is minimized. You should avoid contaminating skin and clothing with blood, body fluids or excrement. Thoroughly wash your hands and any other contaminated skin surfaces with a germicidal skin cleanser immediately after handling wild animals or their samples. You should also be aware that many amphibians produce toxic skin secretions. The effects of these toxins can range from mild irritation to more severe symptoms. All personnel handling amphibians should practice good hygiene and avoid rubbing their eyes after contact.
2. Appropriate precautions should be taken in order to prevent injuries from bites, scratches and skin punctures from wild animals. Even minor injuries may become infected. Basic first aid and appropriate hygiene can prevent such complications.
3. Where there is a risk from aerosolized pathogens from saliva, faeces or urine, protective gear such as gloves, eye protection, respiratory protection (masks or respirators), foot protection and protective clothing should be used as necessary. You should always wear disposable gloves when handling sick or dead animals.
4. All contaminated equipment should be cleaned and disinfected immediately after use.
5. All drug containers, needles, scalpel blades, suture needles and other sharp instruments should be used and disposed of in a manner which prevents accidental human injury.
6. Those individuals who are exposed to potential vectors of rabies (e.g. skunks, raccoons, foxes, bats or animals with abnormal nervous system symptoms) should immediately report the exposure to medical authorities. Those researchers working with bats and carnivores may wish to consider pre-exposure vaccination. These vaccinations may be given to investigators who routinely handle high-risk species from various sources. All field workers should maintain up-to-date tetanus immunizations. A number of infectious diseases that are transmitted by arthropod vectors may be acquired without direct contact with mammals. Arthropod-borne diseases such as Lyme disease, ehrlichioses, Rocky Mountain spotted fever, the equine encephalitides, dengue fever and malaria are examples of these agents.
7. A key component of safety in the field is common-sense personal hygiene. You should wash your hands, field clothes and any other materials that come in contact with mammals or their blood or body fluids. Take precautions to prevent contamination of food and living areas with droppings and urine.

**8.4. Venomous Snakes**

Only experienced personnel should handle venomous snakes. They should never work alone and be familiar with the emergency procedures that are to be initiated in the event of an accidental bite or contact. A supply of the appropriate antivenin and a posted treatment protocol should be available at all times. In addition, a physician or medical facility should be made aware of the nature of the studies being undertaken so that proper arrangements can be made for emergency care and examination.

**8.5. Allergies**

Individuals with known severe allergies associated with animals, with immune deficiency diseases, or on immunosuppressant therapy, should not engage in studies involving the handling of wild animals.

**8.6. Records**

You should maintain a standardized record of any injuries or illnesses incurred in the field or laboratory. Such information should accompany the individual requiring examination or treatment by a medical practitioner. Researchers should also maintain records and pertinent product information of all immobilization drugs in their possession and their usage.

**CAPTURE, HANDLING AND TRANSPORTATION OF CROCODILES**

**1. INTRODUCTION**

Capture in relation to an animal, includes hunting, shooting, wounding, killing, skinning, poisoning, netting, snaring, spearing, trapping, catching, dredging for, bringing ashore or aboard a boat, pursuing, luring, injuring or harming the animal or attempting to do any of these things. During and after capture, crocodiles are often severely stressed and are easily killed or injured by inappropriate handling. This is particularly true of large crocodiles.

**2. CAPTURE**

Crocodiles in the wild can only be captured/taken by people with skills and training in crocodile capture and handling techniques who are authorised to undertake the activity under a permit or authority issued by the government.

In considering the best capture methods, it must be recognised that:

1. crocodiles usually struggle before they can be restrained

2. metabolism during struggling is anaerobic and acidosis (an increase in lactate and hydrogen ion concentrations in the blood causing an acid base disturbance) can result

3. where live capture is undertaken, crocodiles need to breathe freely and deeply after capture to flush out carbon dioxide and recover from acidosis

4. restraining methods and anaesthetics or immobilising agents, if used, must not impair breathing, especially with large crocodiles (>4.5m)

5. minimising damage to the skin is often a key priority (skin damage is also often a good indicator of poor handling).

There are four main periods where stress is most likely to occur: the capture itself; retention in a capture device; handling; and transportation. Capture methods can be divided into ‘direct’ (where a person actively captures a crocodile, for example through harpooning) and ‘indirect’ (where a person is not present when a crocodile is captured, for example through trapping) methods.

**2.1. Trapping**

All traps and other equipment used for trapping crocodiles must be in sound working order and be free of sharp or abrasive components, which could injure a trapped animal. This equipment must not be used in situations that could cause injuries or unnecessary trauma to crocodiles. For example, a mesh trap must not be set below the high tide level and a floating trap must not be left exposed at low tide. Measures must also be taken to ensure trapped crocodiles do not overheat or dehydrate while they remain in the trap. Capture devices must be checked regularly (daily wherever possible) and the crocodiles removed promptly. Where delays are unavoidable, or to minimise the risk of injury to crocodiles (through struggling) and people, when removing crocodiles from traps or other equipment, veterinary advice may be sought and immobilizing agents may be used.

**2.2. Harpooning**

Harpooning is a quick, efficient method of capturing targeted crocodiles, which reduces the period of struggling (and therefore stress). So as not to damage muscle and other underlying body tissues, all harpoons must be designed and built to ensure that the harpoon barbs penetrate no deeper than just below the skin of a crocodile (barbs must be no longer than 2.5-3cm to prevent unnecessary penetration). Where possible, a harpoon should be placed in the neck area of a crocodile. The skin on the neck does not contain oesteoderms and therefore the harpoon head is less likely to be deflected. The neck is very muscular and is usually the area exposed when the animal is in the water and is therefore the preferred target. The time of struggling must be reduced to a minimum to reduce stress, and the crocodile must be restrained as soon as practical after the initial capture effort. The harpooning of crocodiles greater than 3.5m long is not recommended.

**2.3. Snares**

The use of snares is an effective technique in the capture of crocodiles. The snare should be designed so as to restrain the top jaw of the crocodile and must be adequately tethered to prevent the animals escape. The snare requires a self locking mechanism to secure the jaw effectively and is best constructed of stainless steel wire cable. The cable may be plastic coated to reduce superficial trauma to the crocodile during snaring. Operational snares must be supervised to ensure the crocodile is recovered and restrained as soon after capture as possible to reduce struggling, risk of stress and exposure to potential conspecific aggression.

**2.4. Netting**

Crocodiles inhabiting small waterholes or creeks/rivers may be captured by nets. Nets used for this purpose are of nylon cord construction, usually with a mesh size of 50 to 60mm across the diamond. Seine nets are used as tangle nets, are usually set in one location and must be monitored intensively for signs (float movement) that a crocodile has become entangled. Nets must be monitored constantly and never left alone for any period of time. Nets set in one location must be checked regularly by lifting the lead line to the surface, exposing the entire drop of the net. This allows non-target species to be quickly removed and returned to the water. Drag nets can also be used to target the capture of a specific crocodile in water holes or small impoundments. The dragging of nets may expose the operator to potential interaction with the crocodile and must only be implemented if infrastructure exists to eliminate this risk.

**3. RESTRAINT**

Attempting to restrain large crocodiles by tying ropes at multiple points on the body is rarely effective and can lead to severe injuries if the animal struggles. The most effective method for holding an animal for any length of time is for it to be unrestrained within a specially designed crocodile transport box or floating trap. If this cannot be organised, the animal should be restrained by fixing two top jaw ropes behind the large eye-teeth of the top jaw, the jaws should be tied and/or taped together and the top jaw ropes tied to a swivel on a fixed rope between two stationary anchor points. This allows the animal to move around freely within a defined area. The animal must always be tethered in the shade. The limbs should remain free (see *Limb restraint* below). Great care must be taken to ensure that crocodiles are not exposed to direct sunlight for any length of time. Direct sunlight can kill within hours through overheating. Crocodiles held out of water for more than a day or two must be covered with sacks and watered regularly to prevent sunburn which causes cracking and bleeding between the scales. Care must be taken to ensure the crocodile is not attacked by ants or that moist parts of the body, like eyes and nostrils, or open wounds, do not get fly-blown. The condition of restrained animals must be monitored regularly.

**Limb restraint**

As soon as the jaws are secured, the eyes must be covered with a wet sack to reduce visual stimulation, if any additional tying is to be carried out. If it is essential to restrain the limbs temporarily (to prevent struggling), use only wide webbing or tape (5-10cm wide), tied loosely so as not to restrict the blood circulation. Do not restrain the limbs of crocodiles for longer than two hours. This procedure invariably causes oedema (fluid accumulation) in the feet and can cause severe tissue damage or loss of limbs.

**Rolling or pulling crocodiles**

Lack of care when rolling large crocodiles or lifting them in rope traps can cause dislocation or breakage of limbs. If it is necessary to roll or drag a crocodile for any reason, then ensure the limbs are tucked in parallel to the body and held in place. It is best to drag crocodiles using the top-jaw ropes used to restrain the jaws. In dragging male crocodiles, it is essential to ensure the penis is not extruded, as dragging can cause trauma to this organ. Do not use the limbs as levers or vantage points under any circumstances.

**Cloacal prolapse**

The cloaca of crocodiles, where the reproductive and excretory organs are found, is a delicate and weakly muscled sac. If large crocodiles are lifted or transported without supporting the cloaca, it is possible to cause collapse of these muscles and severe injury or death. In lifting large crocodiles inside or outside traps, it is essential to bind sacking or other support around the base of the tail to prevent cloaca prolapse or trauma to the extruded penis. However, it is critical that any binding used to minimize cloaca prolapse is removed as soon as practicable to allow the animal to urinate and defecate normally.

**Use of drugs**

Drugs such as Pavulon and Valium have some value in crocodile handling, but can be dangerous in inexperienced hands. All crocodiles can be handled safely and effectively without drugs. Use of some drugs on large crocodiles (more than 3m) under stress can cause acidosis and death. Drugs must not be used except under the supervision of an experienced person or qualified veterinary practitioner. Pavulon or other agents with a respiratory depressant effect must not be used on harpooned crocodiles during or for several hours after capture. Valium is an effective agent for calming very large crocodiles in traps during removal operations. Its effect is short-lived, typically lasting 2-3 hours. As with many drugs, giving repeated ‘top-up’ doses can be dangerous. Complete recovery from the effects of drugs must be confirmed before crocodiles are released to the wild or captive environments with bodies of water. A crocodile compromised by the effects of drugs can potentially submerge in water and drown.

**Dehydration**

Contrary to popular belief, large crocodiles will not dehydrate rapidly if left in shade out of the water for a period of days. Nonetheless, captive animals must be wet regularly and given water to drink. Crocodiles do not drink sea water, but do drink fresh water.

**4. TRANSPORT**

The method of transport used for live crocodiles will generally be determined by the size of the crocodile(s) involved. Care must always be taken to avoid the effects of exposure, including dehydration, overheating (>35°C), excessive cooling (<20°C) and struggling, and to minimise transport time. Smooth interiors for containers and padding around the snout of the crocodile can minimise snout damage, and are recommended. When transporting crocodiles, it is essential to have full support under the head, body and tail base to avoid damage to essential organs. A simple restraining board with webbing straps is effective for crocodiles up to 3m long for short-term transport under supervision. Transporting crocodiles short-term within the confines of a floating mesh trap is often an effective means of reducing handling and stress on wild caught crocodiles, however, due regard must be given to potential exposure and/or cloacal prolapse. Crocodiles may also be transported in a vehicle or vessel for short distances if adequately restrained. A solid ventilated box is necessary for long-distance transport or unsupervised cartage. Ensure that the head is not lower than the body during transport so that any regurgitated fluids can flow back down the oesophagus rather than pool at the opening of the glottis. If the mouth of a crocodile is tied closed, a stick or block must be placed between the teeth to hold the mouth slightly ajar. This will minimise the risk of drowning the animal if it vomits under restraint. Where possible, crocodiles should not be fed for at least three days prior to transport to minimise risks. Where possible, boxes should have smooth material that will limit frictional damage to the skin and claws when the animal moves or struggles. Crocodiles must be regularly doused with water during transport. Despite their size, crocodiles are delicate animals and are easily killed by pounding on hard surfaces during boat or truck transportation. Transport overland across unmade roads must be avoided wherever possible. Suitable cushioning must be used to minimise vibration and shocks where these are unavoidable. Where possible, crocodiles should not be subjected to large public gatherings and display during transport or handling operations. Visual stimulation should be reduced by covering the eyes or keeping the crocodile in a dark container. Captured animals are already in a stressed condition and noise and handling must be kept to a minimum.

If excessive struggling has not occurred, chemical immobilisation with drugs can be considered where necessary. In the specific case where immobilising drugs have been administered to large (>4.5m) crocodiles after prolonged periods of struggling, artificial ventilation of the lungs is sometimes undertaken to enhance the removal of carbon dioxide, and bring about quicker recovery from the effects of capture. As the effects of capture stress may persist for many days, animals must be closely monitored for the first few days after release.

**5. EUTHANASIA**

In certain circumstances, such as when a crocodile has attacked a person, it may be necessary to euthanise the animal. Euthanasia must only be undertaken by trained and experienced personnel.

Crocodiles can be euthanised by shooting, spinal severance or administration of appropriate drugs by a veterinarian or other qualified person. It is important to use the most humane method of killing so the animal is killed instantly or instantaneously rendered insensible to pain until death supervenes. Care must be taken regarding the type of euthanasia chosen should the meat be required for pet or human consumption.

Crocodiles of any size may be shot by rifle through the back or side of the cranial platform, or between the eyes. The use of a captive bolt is considered an acceptable method of euthanasia with reservations. These reservations are based on equipment being fully functional and personnel that are trained and skilled in the procedure to ensure operator safety and animal welfare. Anatomic references for targeting rifle shot or captive bolt gun should be consulted when using these methods.

**Shooting:**

*Large or small crocodiles at Free range*

• Minimum .30 + calibre 150gn

• 12 Gauge Shotgun with Sg or SSG ammunition at very close range.

*Captive or restrained crocodiles*

• up to 2m long – .22 shorts with solid projectiles (low velocity)

• 2-3m long – .22 long rifle or .22 magnum with solid projectiles

• 3m or longer – only high velocity centre-fire rounds.

**Nape-stab:**

Crocodiles less than 2m long that are firmly secured can be killed rapidly and humanely with a hard hammer-blow to a sharp metal chisel positioned between the skull and the first cervical vertebra, just behind the cranial platform. This severs the spine and shocks the brain (thus stunning the animal). The brain must then be destroyed by ‘pithing’ (insertion of a rod into the brain) as soon as possible. Due to the physiology and neural organisation of crocodiles, some reflex activity may be evident after killing – this is not indicative of inhumane or improper methods of killing.

**Pentobarbitone sodium injection:**

For crocodiles less than 1.5m, a pre-euthanasia tranquilisation by heavy sedation or anaesthetic is administered either intravenously or intramuscularly. This is followed by administration of intravenous, intracardiac, or intraperitoneal injection of euthanasia solution.

**CAPTURE AND IMMOBILIZATION OF JAGUARS** (*Panthera onca*)

**1. INTRODUCTION**

Jaguars (*Panthera onca*) are the largest felid species in the New World and the only member of the genus *Panthera*, the roaring cats, that occurs in the Americas. They are the third largest cat species, being outsized only by lions (*P. leo*) and tigers (*P. trigris*). Although not the largest felid, jaguars have the strongest jaw in relation to head size of any of the cats, a fact that should be remembered whenever planning to capture and immobilize these animals. The body weight of jaguars is 90 - 120 kg for males and 60 - 90 kg for females, with a large variation in body size. Jaguars live in a wide variety of tropical habitats, ranging from montane forest and wet savannah to tropical rain forest and deciduous tropical forest. The largest documented jaguars occur in wet savannahs while jaguars that live in more forested regions tend to be smaller in size.

Historically the range of jaguars was the southern USA through Central and South America as far south as southern Argentina. Their current range is limited to a broad belt from central Mexico through Central America to Northern Argentina. The jaguar is known to be rare or extinct in many parts of its former range and it is approximated that 10,000 are left with several subspecies being rare.

Non-infectious problems include a high incidence of neoplasia which may be associated with husbandry in captivity and/or longevity. The infectious agents include protozoan, bacterial and viral pathogens (i.e., canine distemper, feline infectious peritonitis). Additionally, there is serologic evidence of infection with canine distemper and feline immunodeficiency virus. It is also assumed that jaguars are susceptible to the common respiratory disease agents (i.e., *Chlamydia* sp., herpesvirus-1, and calicivirus) in domestic and non-domestic cats.

**2. IMMOBILIZATION PROCEDURE**

*General Principles*

Any person who immobilizes a wild jaguar must remember that she/he is solely responsible for the health of that animal from the time the drug is administered (or from the time the animal is captured or treed) until the animal has fully recovered from the anaesthetic agent(s). It is imperative that anyone engaged in the immobilization of free-ranging jaguars know how to handle the anaesthetized cat, monitor physiologic parameters, and respond to medical emergencies should they arise. Although many anaesthetic agents are relatively safe in felid species, anaesthetic emergencies can and do occur even under the best of circumstances.

Unlike the hospital setting where anaesthesia is more controlled, there are unique problems related to immobilization of free-ranging wildlife in general, and large cats in particular. The capture method may itself result in injuries. Jaguars are aggressive cats and often when trapped will bite on cage material. Free-ranging jaguars have succumbed to tooth root abscesses following fracture of canine teeth from the capture procedure. If chased by dogs and darted while in a tree, the fall itself may cause injury. For this reason, it is best to not dart a jaguar above 5 metre high in a tree to avoid traumatic falls. Jaguars are often highly stressed during capture. The capture team must strive to minimize stress due to the effects that stress may have on physiologic parameters that may compromise the animal once anaesthetized. Lastly, it must always be remembered that capture in the wild of a potentially dangerous animal, such as a jaguar, has inherent risks for the capture team.

*Capture Methods*

Methods that have been used to capture free-ranging jaguars include treeing the animal using dogs, padded foot-hold traps, snares (i.e., Aldrich snares) and cage or box traps. The later two methods may or may not include bait (i.e., live goat or pig) to lure the animal to the trap. Once the jaguar is treed or trapped, it can then be darted. The capture method employed for each jaguar capture should be based on the immobilization team’s previous experience, methods that have been successful in the region (if studies exist), habitat, and current weather conditions. In every capture and immobilization procedure the top priority is for a safe anaesthetic event for both the jaguar and the people involved with the procedure.

*Pre-anaesthetic Management*

Once a jaguar has been captured, it is important to perform the anaesthesia as quickly as possible. When in a cage, the possibility of damaging canine teeth is high and may increase with prolonged time in the cage. A technique to minimize stress includes tranquilizer tablets which are commonly used with padded foot-hold traps, but may be of value with the other capture methods. As is true for field immobilization in general, you should not take a lot of time once you begin your initial approach to dart the captured animal. An approximation of the body weight for the calculation of drug must be done to minimize a drug dose error. It is imperative that you have your entire immobilization equipment ready prior to approaching the cat.

*Anaesthetic Administration*

Anaesthetic agents in a blowpipe, or possibly a pole syringe, may be used for immobilizing jaguars in a cage, foot-hold trap or snare. In all other field situations, it is best to use a rifle or pistol (i.e., Telinject®, Cap-Chur®, Dan-Inject®). You must be familiar with the instrument you choose for use in the field. Darting animals is always associated with some degree of risks. Serious damage to the animal (and human participants) can and does occur if inappropriate instruments are used and/or if instruments are used inappropriately.

Dart and needle selection is also important in preparing for a jaguar immobilization. Darts that are too heavy and needles that are too long/thick can cause serious damage on impact. Damage is also possible if the charge of the dart or the charge of the rifle/pistol is too high. If the jaguar is not adequately immobilized and cannot be restrained, the dart will remain in the animal and may cause problems.

The use of 1.5 x 30 mm (18 gauge x 1 - 1/4 inch) collared needles is recommended for immobilizing free-ranging adult jaguars. However, if the jaguar is treed or trapped prior to darting, a non-barbed (plain) needle can be used. Non-barbed (plain) needles cause less trauma to the tissues but often do not remain in the animal as long as collared needles and thus may not inject all the drug prior to falling from the animal. When darting a jaguar, it is safest to aim for the proximal region of a rear limb. Serious harm can be inflicted on the cat if the dart hits the thoracic region or head. When aiming for the rear limb, darts should be placed in the caudal most aspect of the muscle mass to avoid the femoral bone and the sciatic nerve. Needles and darts must be disinfected prior to use on the next animal to prevent the spread of diseases.

**Anaesthesia**

The following anaesthetic regimen is recommended for use by field personnel with little experience in immobilizing free-ranging jaguars. This regimen should provide an adequate plane of anaesthesia for short-term work on the jaguar (i.e., radio-collar application, morphometric measurements, biomaterial collections) while requiring a minimal level of technical skill in anaesthesiology.

Telazol (6 - 10 mg/kg) IM as the dose for immobilization in free-ranging jaguars. The darter has the option to include 150 mg ketamine in the initial dart. Supplemental ketamine at a dose of 1 - 1.5 mg/kg, IV or 1 - 2 mg/kg IM, as needed to maintain an adequate level of anaesthesia. (No supplemental ketamine should be delivered for at least 10 minutes after the initial dart containing telazol.) Atropine at a single dose of 0.04 mg/kg either SC or IM may also be administered if the cat has excessive salivation.

(NB: There have been anaesthesia related problems with telazol use in large cat species, in particular tigers). You should be prepared to deal with unexpected reactions.

**Antagonists**

Flumazenil is the antagonist for zolazepam (the benzodiazepine component of telazol) and can be administered, once all procedures are completed, at an IM dose of 1.0 mg of flumazenil for each 40 mg of telazol used. Flumazenil should not be administered for a minimum of 30 minutes after the initial dose of telazol was delivered to ensure the tiletamine component of telazol is nearly completely metabolized. There should also be at least 30 minutes between the administration of any supplemental ketamine administration and flumazenil.

Yohimbine is the antagonist for xylazine and should be administered at 0.125 mg/kg IM and should only be delivered once the procedure is completed and at least 30 minutes after the last dose of the cyclohexane (ketamine) was given.

Atipamezole is the antagonist for medetomidine and can be administered once all procedures are completed, at a dose of 4 - 5 x the medetomidine dose. For example, if 40 ug/kg of medetomidine was used for immobilization, reversal with atipamezole should be at a dose of 160 - 200 u/kg. This should be delivered IM. Atipamezole should not be administered for a minimum of 30 minutes after the last dose of cyclohexane (ketamine) was given.

**Supplemental Drugs**

There will be occasions when the initial anaesthetic agent(s) does not provide adequate immobilization or when the effect of the anaesthetic agent(s) begins to wane (i.e., increased animal movements, increased respiration and heart rate) prior to all procedures (i.e., radiocollar application, sample collection) being completed. In these cases, it may be necessary to administer supplemental drugs for adequate anaesthesia to allow safe handling. The following should be kept in mind if one is faced with either of these situations.

Ketamine at a dose of 1 - 1.5 mg/kg IV or 1 - 2 mg/kg IM, as needed to maintain an adequate level of anaesthesia should be a safe dose in adult jaguars.

Diazepam (valium) at the dose of 5 - 10 mg/jaguar should be administered slowly IV to any jaguar with extreme muscle rigidity, muscle tremors, and/or seizures. Diazepam can be administered again IV after 3 minutes if there is no response to the initial injection. If the jaguar still does not respond following the second injection, another cause of the seizure activity should be considered. If a vein cannot be located (i.e., moving animal), diazepam can be injected IM. Caution should be exercised in administering a second dose of diazepam following an IM injection due to a potentially slower rate of metabolism with IM injections.

Never use telazol as the supplemental drug. If telazol is the initial immobilizing agent and it has not provided adequate anaesthesia or if its anaesthetic effects have worn off, it is best to supplement with ketamine either IV or IM. The dose of ketamine will depend on the plane of anaesthesia prior to supplementation. 25 - 50 mg IV or 50 - 100 IM mg of ketamine total per jaguar should be a safe dose in adult jaguars.

Never use xylazine, medetomidine and midazolam as the supplemental drug. They should only be administered in combination with another drug (i.e., ketamine) for induction of anaesthesia. It is best to supplement with ketamine either IV or IM. The dose of ketamine to deliver will depend on the plane of anaesthesia prior to supplementation. 25 - 50 mg IV or 50 - 100 mg IM of ketamine should be a safe dose in adult jaguars.

If you are not sure of how much of the original drug(s) was successfully administered (i.e., poor dart placement, dart bounced in and out quickly), you should wait at least 15 minutes following the initial dart prior to administering any additional agents.

**Anticholinergics**

Some authors recommend the addition of atropine to the anaesthetic protocol for the anticholinergic property of decreasing salivary secretions. However, atropine can be associated with negative side effects, most commonly on the heart and gastrointestinal tract. In field situations it may be more appropriate to administer atropine only to those cats that are displaying excessive salivation during the immobilization procedure. A single dose should be administered:

Atropine - 0.04 mg/kg SC or IM.

**Animal Handling and Monitoring**

Standard equipment for handling and monitoring the anaesthetized jaguar should include those listed below in Table 1.

All handling equipment (i.e., towels, non-disposable gloves, veterinary supplies) should be disinfected prior to use on another animal to prevent the spread of diseases. Immediately after the animal is darted and an initial assessment of the respiratory rate (RR; 8 - 24 breathes/minute) and heart rate (HR; 70 - 140 beats/minute) are deemed within normal limits, the dart should be collected (avoid handling the needle) and put in a safe place. It is best to have one person immediately take the physiologic parameters while a second person is in charge of the dart. The dart site on the animal should not be touched to avoid contact with drug residues and blood. People who will have contact with the immobilized animal should wear latex gloves during the immobilization procedure to avoid the transmission of infectious diseases between the animal and him/herself, as well as to minimize contact with drug residues at the injection site. The animal should be placed in a position that allows it to breathe easily; preferably, the jaguar should be placed in lateral recumbency. The head and neck should be placed in a position that allows air to flow through the mouth and trachea. The mouth should be kept lower than the back of the throat and neck so saliva flows out of the mouth and not into the trachea.

Once the animal is anaesthesized and placed in the proper position, the eyes must be protected. A triple antibiotic eye ointment (i.e., Trioptic-P®) should be applied in both eyes to prevent them from drying due to the lack of the normal blink response which is often the case when using ketamine and telazol anaesthetics. A towel (nonabrasive material preferably) should then be placed over the eyes to protect them from the sun and dirt, as well as to minimize stressful stimulus to the animal. Cotton balls may be placed in the outer ear canal to minimize auditory stimulus. However, should one choose to use these, one must remember to remove them at the completion of the immobilization procedure.

**Table 1. Standard equipment for handling and monitoring the anaesthetized jaguar**

|  |  |
| --- | --- |
| **Monitoring Equipment** | **Emergency Equipment** |
| Stethoscope | Laryngoscope |
| Thermometer | Endotracheal tubes |
| Pulse oximeter | Ambu bag or Oxygen tank |
|  | Anaesthetic reversal agents |
|  | Emergency drugs |
|  | Portable ice packs |
|  | Dental repair kit |
|  | Surgical pack |
|  | Bandage material |

It is important to reduce the risk of wound infection by screwworm (*Cochliomyia hominivorax*). Topical betadine and a flystrike ointment can be applied to the dart site, and to any abrasions that occur during the procedure, to protect against screwworm.

During all jaguar immobilizations, the physiological parameters (i.e., respiratory rate, heart rate, and temperature) MUST be monitored. If these values fall outside the normal range, the immobilization team should be alerted to a potential impending emergency and be ready to respond in the appropriate manner. The normal physiologic parameters for an immobilized free-ranging jaguar are the following:

Temperature: 37 - 39.50C (98.6 - 103.10F)

Respiratory Rate: 8 - 24 breathes/minute

Heart Rate: 70 - 140 beats/minute

**Both respiratory rate and heart rate should be monitored every 5 minutes and the temperature should be taken every 10 minutes.**

Monitoring these parameters can best be done by use of a thermometer, visual observation of chest wall expansion, and either palpation of the femoral pulse or use of a stethoscope. A rectal thermometer should be placed in the anus (digital thermometers are the best and easiest to use in the field) and the temperature monitored at 10 minute intervals during anaesthesia. Respiration can be monitored by watching the thorax move when the animal breathes. The easiest way to determine the respiratory rate per minute, is to count the thoracic movements during 15 seconds and then multiply this number by four. If one does not have a stethoscope in the field, then light digital pressure over the femoral artery will provide a measure of the heart rate. Alternatively, a stethoscope can be used to auscultate the heart directly over the lateral aspect of the cranial thorax.

The recognition of what are normal jaguar responses to anaesthetic agents is also imperative. Jaguars immobilized with telazol and ketamine usually will have increased salivation, open eyelids, whole body muscle rigidity (including jaw tone), and intact reflexes (i.e., corneal, pedal). Jaguars should maintain swallowing and coughing reflexes with these agents, but should not have muscle tremors and seizure-like activity. In addition to the drugs necessary for safe and effective anaesthesia (which includes a triple antibiotic eye ointment), a number of supportive medications are valuable for field work. Ivermectin (200 mcg/kg SC) should be administered to prevent screwworm infestation. Fluid therapy with Lactated Ringer’s solution (10 - 20 ml/kg IV or SC) for rehydration should be provided especially if the jaguar was trapped for an extended period and/or was highly stressed and hyperthermic. A long-acting antibiotic such as penicillin G benzathine (40,000 IU/kg IM) should be administered, especially for jaguars that have sustained significant trauma from the dart or a fractured tooth, had vomited during the procedure, or had active lesions at the time of immobilization. Both topical fly-strike and triple antibiotic ointments should be placed on the dart site as well as any active skin lesions.

**Troubleshooting Common Anaesthetic Emergencies in the Field**

Any person who immobilizes a wild jaguar must remember that he/she is solely responsible for the health of that animal from the time the drug is administered (or from the time the animal is captured or treed) until the animal has fully recovered from the anaesthetic agent(s). It is imperative that anyone engaged in the immobilization of free-ranging jaguars know how to handle the anaesthetized cat, monitor physiologic parameters, and respond to medical emergencies should they arise. Although many anaesthetic agents are relatively safe in felid species, anaesthetic emergencies can and do occur even under the best circumstances.

The most common anaesthesia emergencies in free-ranging jaguars are respiratory depression and arrest, cardiac arrest, seizures, hyperthermia, and wounds including canine tooth fractures. Additional problems include vomiting and aspiration, shock, capture myopathy, and dehydration.

**Respiratory Depression and Arrest**

This results in tissue hypoxia caused by inadequate oxygenation of blood haemoglobin and is probably the number one anaesthetic emergency encountered in the field. Diagnosis of respiratory depression/arrest is based on:

The jaguar taking few or no breathes (i.e., less than 4; no chest expansion) per minute;

Blue/gray mucous membrane (mm; gums);

Oxygen saturation is < 80% on pulse oximetry (if available).

During field immobilization there are a number of causes for respiratory depression/arrest including: 1) drug-induced depression of the respiratory centre; 2) airway obstruction due to malpositioning, excessive salivation or regurgitation, laryngeal edema; 3) pressure on the diaphragm from gastrointestinal contents; 4) excessive build - up of carbon dioxide which alters normal respiration; and 5) inapparent underlying disease process.

Treatment of respiratory depression / arrest should include the following:

1. Do not panic (this is true for all anaesthetic emergencies).

2. Do not administer any additional immobilization drugs.

3. Be sure the head and neck are in good positions (extended with no objects compressing them) so air can move through the mouth and trachea. Be sure there is no vomit or foreign objects blocking the trachea.

4. Intubate immediately if an endotracheal tube (ETT) is available. Administer oxygen through the ETT using an ambu bag, your own breath, or an oxygen tank (if available).

5. If no ETT or supplemental source of oxygen is available, use intermittent pressure on the chest to attempt to move air through the lungs. The jaguar should already be in lateral recumbency. Push down firmly on the chest at regular intervals (i.e., press for 1 second, wait for 1 second, press for 1 second and so on). Alternatively, you may attempt mouth-to-mouth or mouth-to-nose resuscitation. Exhale into the jaguar’s mouth or nose for a count of 2 sec and then inhale away from the cat’s mouth/nose for a count of 2 sec.

6. Administer 1 - 2 mg/kg doxapram IV (or IM in the tongue muscle if one cannot quickly find a vein). Note: Doxapram can cause arousal, especially in a cat immobilized with telazol, and caution for human safety must be considered if one elects to use this drug as a respiratory stimulant. Some veterinary anaesthesiologist no longer recommend the use of this drug. If respiratory arrest is not corrected with steps 1 - 5 above, use doxapram as a last attempt for resuscitation. If you must inject the drug into the tongue, you should be very careful not to traumatize the oral cavity.

7. Administer appropriate anaesthestic antagonist if available (i.e., flumazenil, yohimbine, atipamezole). However, do this cautiously as the antagonist will only reverse the drug it antagonizes and the jaguar may be semi-anaesthetized and difficult to handle after the antagonist is administered.

**Cardiac Arrest**

This is usually preceded by respiratory arrest and is defined as the loss of effective cardiac function resulting in cessation of circulation. This is the most serious anaesthetic emergency encountered during field immobilization.

Diagnosis of cardiac arrest is based on:

Weak or absent pulse or heart sounds;

Blue/gray mucous membranes (gums);

Poor capillary refill time measured by applying digital pressure to the mucous membrane until the mucous membrane (mm) turns pale and then releasing the pressure and monitoring the seconds it takes until the mm color returns to normal (this value should be < 2 sec);

Dilated pupils;

Cold extremities;

Loss of consciousness (hard to evaluate if the animal is anaesthetized).

The most common causes of cardiac arrest during field immobilization are 1) drug-induced; 2) respiratory failure leading to hypoxia; 3) acid-base or electrolyte imbalance; and 4) underlying disease process.

Treatment of cardiac arrest should include the following:

1. Do not administer any additional immobilization drugs.

2. Be sure the animal can breathe prior to starting cardiac massage.

3. Begin external cardiac massage. The jaguar should already be in lateral recumbency.

Apply firm pressure downward over the heart. Compression of the heart should be for a count of 1 and release for a count of 1 with 60 - 100 cycles/minute. If an assistant is available he/she should palpate the femoral pulse to ensure adequate pressure, to circulate blood, is being applied during cardiac compressions.

4. Administer 0.02 mg/kg of 1:1000 (1.0 mg/ml) epinephrine IV or intracardially and continue with external cardiac massage. This dose is approximately 1.6 mg (1.6 ml) per 80 kg adult jaguar.

5. Administer 20 ml/kg cool Lactacted Ringer’s Solution as an IV bolus (i.e., a single rapid infusion).

6. If no response, repeat 4 above at 5 minute intervals indefinitely.

**Seizures**

This is defined as disturbances of cerebral function characterized by a violent, involuntary contraction or series of contractions of the voluntary muscles.

Diagnosis is made based on clinical signs that include the following:

Uncontrolled muscle and/or whole body spasms;

Rigid extension of the limbs.

Causes include 1) drug-induced (i.e., ketamine and tiletamine); 2) trauma; and 3) hypoglycemia.

Treatment includes the following:

1. Administer 10 mg (total) diazepam IV slowly over 10 - 15 seconds.

2. Repeat step above if no improvements within 3 minutes.

3. Monitor body temperature to determine if secondary hyperthermia results from the seizure activity.

**Hyperthermia**

This is defined as an increase in body temperature to a point where oxygen demand exceeds supply due to increased metabolism.

Diagnosis of hyperthermia: easily determined by rectal thermometer.

**Temperatures > 410C (105.80F) are true emergencies.**

Causes of hyperthermia in field immobilization include 1) internal heat production due to excessive physical exertion; 2) external heat absorption; 3) drug-induced compromise of thermoregulation; and 4) inability to use behavioral thermoregulation.

Treatment of hyperthermia includes the following:

1. Do not administer any additional anaesthetic agents.

2. Make sure the jaguar is in the shade.

3. Use portable "cold" packs that can be placed in the groin, axillae (armpit) and belly of the jaguar.

4. Cool the jaguar by applying water over the body and/or alcohol to the extremities (legs and feet).

5. Administer cold water enema if tubing is available.

6. Administer 20 ml/kg cool Lactacted Ringer’s Solution as an IV bolus (i.e., rapid fluid infusion).

7. Take the temperature every 5 - 10 minutes to determine if the temperature is decreasing. Continue to wet the animal if the temperature remains high.

8. Administer antagonist IV (IM if a vein is not readily identified). However, do this cautiously as the antagonist will only reverse the drug it antagonizes and the jaguar may be semi-anaesthetized and difficult to handle after the antagonist is administered.

9. If it is believed that the hyperthermia is due to muscle rigidity and a light plane of anaesthesia, diazepam at a dose of 5 - 10 mg/jaguar total can be administered slowly IV to decrease muscular activity.

Note - Hypothermia (< 35**0**C = < 95**0**F) - decreased body temperature to point of cellular death - is much less likely under most field conditions in which jaguar will be immobilized. However, this may occur (i.e., high altitude regions) and should be treated by warming the animal.

**Wounds**

This is often associated with the dart site as well as by trap or chase injuries. (Be especially cognizant of any oral lesions and/or broken teeth).

Diagnosis: based on clinical signs. The severity of the wound will dictate the treatment modality chosen:

Physical examination to evaluate for traumatic lacerations and lesions;

Oral examination to evaluate for oral lesions and broken teeth.

Treatment should always include:

1. Clean the wound with a povidone-iodine or 2% chlorhexidine solution. If neither of these is available, use soapy water.

2. If necrotic tissue is present and the field personnel are familiar with veterinary surgical techniques, debride the dead tissue and repeat step.

3. Only suture those wounds that you know are fresh (i.e., caused by the dart) and that require sutures to minimize further tissue damage. Again, only field personnel who are familiar with veterinary surgical techniques should attempt to suture any wounds.

4. Apply topical antibiotic and fly-strike ointment to wound site.

5. Administer long-acting antibiotic IM (i.e., Penicillin G benzathine 40,000 IU/kg IM).

6. Administer ivermectin 200 ug/kg SC (to prevent screw worm infestation at site of broken skin).

Treatment of broken teeth

It is imperative that a fractured tooth (most commonly a canine is broken during jaguar captures and immobilizations) be repaired to minimize pain and infection associated with the tooth. A calcium hydroxide product (i.e., Dycal ®) can be used to cap the tooth pulp. Instructions for application come with tooth repair kits.

**Post-anaesthetic Recovery**

The recovery period is just as important for proper handling and monitoring as the induction and maintenance periods. It is not uncommon for anaesthetic related morbidity and mortality to occur during this period; in fact, most anaesthetic complications occur during induction and recovery. Although there are reversal drugs for the zolazepam component of telazol (flumazenil), xylazine (yohimbine), and medetomidine (atipamezole), jaguar recoveries cannot be completely reversed with one specific antidote as is available for narcotic immobilization agents (i.e., carfentanil, etorphine) commonly used in hoofstock. For this reason, it is important to ensure that the jaguar does not cause injury to itself or to people involved in the immobilization during the recovery period. During recovery, the jaguar should be positioned so that it can breathe easily and will not harm itself on objects near it. The animal should be placed in lateral recumbency with the head and neck extended. Abrasive material should not be under the head due to possible head movements that could lead to corneal abrasions. People in the area remain quiet and should not stimulate the jaguar. It should recover at its own pace as it metabolizes the anaesthetic agent(s). Stimulation will not result in a faster recovery, but it may cause the jaguar to injure itself.

If the jaguar was originally captured in a box trap, it may be beneficial to let the animal recover in the cage where it is dark and quiet. However, it must be remembered that when the cat is awake enough for release, the danger to field personnel may be significant when opening the cage. While in the cage and recovering, the animal may also be aggressive and cause harm to itself. Thus, if one is to use a box trap for recovery, it requires judgement to be sure the jaguar is awake enough prior to release, but does not cause harm to itself while still in the cage. Alternatively, when no trap is available (i.e., treed by dogs or darted from a blind), the animal can be placed in a quiet, padded (i.e., with leaf litter), and protected (i.e., not near ledges, hard structures) area to recover on its own. Risks are involved with both recovery methods.

**WILDLIFE AND FISHERIES MANAGEMENT MEDICINE**

**AQUACULTURE PRODUCTION SYSTEMS**

Aquaculture production usually involves three major systems namely: Broodstock systems, Fish seed production systems and Grow-out or table fish production systems.

**BROODSTOCK PRODUCTION:** Fish broodstock are the sexually matured male and female adult fish which are reserved and used for breeding. They are the parent stock which can be selected from the harvested table fish and reared for one year or more depending on the age at maturity of the species. They can also be sourced from the wild but care must be taken to collect healthy mature ones. The source of the broodfish must be known with no previous disease record. Broodstock are stocked at very low stocking densities and fed twice daily with high quality fish feed (= 40% Crude protein) at a feeding rate of 1- 2% body weight. The holding facilities are cleaned regularly and good water quality maintained so as to ensure good health condition and gamete viability.

**FISH SEED PRODUCTION:** Fish seed production is the method of production of off-springs or young ones from the parents. It ensures the continuation of the species from one generation to the other. It is very important in aquaculture for the stocking of ponds and tanks.

Types of Fish Seeds include: Fertilized eggs - (fusion of male and female gametes), Larvae -larvae are hatchlings with yolk sac, Fry -fry are free swimming small fish.

Fingerling - small, finger-sized fish

Post fingerling / Juvenile - these are the advanced stages of fingerlings

**Methods of Fish Seed Production**:

There are two major methods of fish seed production in Aquaculture

**Natural Breeding**: Fish spawns naturally in ponds of tanks after attaining sexual maturity. Injection is not used. E.g. *Tilapia*, Heterotis, Megalops

**Artificial Breeding**: The stocked fish is sexually mature but cannot breed naturally in the tank or pond. Hormone is injected into fish to induce the fish to release eggs or milt E.g. *Clarias spp*.

***Pituitary:*** The pituitary gland produces, accumulates, and stores the gonadotropic hormone(s) which plays a decisive role in ovulation. Insofar as reproduction is concerned, the role of the pituitary gland is that of an intermediary between the central nervous system and the gonads. The gonadotropic hormone(s) is produced by sexually mature fish and the cyclical changes in its concentration in the pituitary gland are correlated with the reproductive cycle of the fish. Its concentration is maximum during the prespawning period, while it is very low or almost nil during and after spawning. The release of gonadotropin(s) by the pituitary gland is “ordered” by the hypothalamus through the secretion of gonadotropin-releasing hormone (GRH). The gonadotropin(s) is also responsible for inducing spawning migration, during which its concentration in the pituitary gland gradually decreases. The gonadal development during spawning migration is most probably directed by the continuously released gonadotropin(s). The pituitary gland is situated on the ventral side of the brain below the hypothalamus, which is connected to the pituitary gland by a funnel-like structure, the infundibulum. The part of the cranium where the pituitary gland is located is known as the sella turcica. The gland is usually embedded in fatty tissue. When the brain is taken out of the skull, the pituitary gland remains connected to the brain in some fishes, while in most fishes the infundibulum ruptures and the gland is left behind on the base of the skull.

**ARTIFICIAL (SYNTHETIC) HORMONES** Different types of synthetic hormones used in Nigeria include Human chorionic gonadotropin (HCG), Luternizing hormone-Releasing hormone analog (LHRHa), Ovaprim, Ovatide etc. The most commonly used hormone is Ovaprim. The dosage is 0.5ml/kg of female spawner.

**HORMONE INJECTION** Carefully catch the female spawner, Cover the head of the female with a wet towel, and Insert the needle 2-2.5 cm deep towards the tail end. The most common method is intramuscular injection instead of interperitoneal. The fish is injected within the muscle at an angle of 450 below the base of the dorsal fin and just above the lateral line. For scaly fish, do not inject through the scale but under it. After injection, finger-rob the injected area. Return injected fish to the holding tank or container, Record the water temperature. Gravid females are injected preferably late in the evening.

**STRIPPING**

**(a) COLLECTION OF MILT:** Kill a mature male (1kg), Open the belly with a pair of scissors, Locate the two testes and carefully remove them. Puncture the testes and squeeze the milt into 0.9% salt solution in a container. It is preferable to collect the milt about 20 minutes before stripping of eggs

**(b) COLLECTION OF EGGS:** Stripping of females is carried out at the end of latency period. Latency period is the time interval between injection and stripping of eggs and is dependent on water temperature.

|  |  |  |  |  |  |  |
| --- | --- | --- | --- | --- | --- | --- |
| Water temperature C | 25 | 26 | 27 | 28 | 29 | 30 |
| Latency period (hours) | 11 | 10 | 9 | 8 | 7.5 | 7 |

Drastic changes in water temperature will affect latency period and may result in very low hatching rates (5 – 10%). At the end of the latency period --

- Carefully catch the injected female with a net.

- Dry the body and hold it tightly with a towel.

- Strip the eggs onto a dry container. Do not allow water to get in contact with the eggs.

- Stop stripping when blood appears. Do not allow the blood to mix with the eggs.

- Return the spent female to the holding tank.

**FERTILIZATION/INCUBATION OF EGGS:** Artificial fertilization of the eggs occurs when water is added to the mixture of eggs and milt.

- Prepare the incubation trays

- Plastic sieves and mosquito netting can be used as incubation trays. They are placed in tanks containing clean oxygenated water.

- Pour the extracted milt over the eggs.

- Gently shake the bowl to ensure even mixing of the eggs and milt.

- Add clean water and mix gently by shaking the bowl. Decant some of the water.

- Pour the fertilized eggs in a single layer onto the incubation trays.

- Aerate the water to enhance hatching.

**HATCHING**

Incubation can be carried out in either running water or in aerated static water in concrete tanks, fibre–glass troughs, jars, trays or boxes. The fertilized eggs usually hatch out between 18 – 30 hrs after fertilization at a water temperature of about 27 - 30oC. Water temperature below 24oC may result in low hatching rates The larvae remain in the incubation unit for 3 – 4 days (depending on water temp.) until their yolk – sac becomes reabsorbed. The yolk fry is not fed. Dead eggs during incubation will become whitish in colour and should be siphoned out of the incubation system to avoid fungal and bacterial infection.

**Artificial incubation of the eggs of catfish; *Clarias gariepinus***

**FRY MANAGEMENT**

Fry rearing is an important aspect of hatchery management because the end products which are the fingerlings are derived from fry. As a result of their small size and delicate body, they can easily be affected by infections, parasites and poor water quality.

The hatchlings possess yolk sac, which serves as food for the first 3 – 4 days. After yolk sac absorption, the fry will start moving about searching for food. The use of live food as first feed ensures higher fry survival where live food is not available, highly nutritious artificial starter feeds must be used like Cyprico, Catco etc. Use the right particle sizes ranging from 0.2 – 0.3, 0.3 – 0.5, 0.5 – 0.8, 0.8 – 1.2 to 1.2 – 1.5mm as the fry grow. Feeding is carried out four to six times daily and fry is fed to satiation. For satisfactory growth and fry survival, the quantity and quality of the artificial feeds are of great importance. Most fish feed manufacturers have feeding tables which serve as guide for the farmer. After 8 weeks, fry should weigh 5-8g. Frequent grading and sorting of fry to remove shooters will enhance survival rates.

Good water quality is paramount for successful fry rearing. The desirable levels of some of the parameters are: water temperature 28oC – 30oC, dissolved oxygen = 3mg/l, pH of 6.5 – 8.5, ammonia < 0.1mg/l and nitrite < 0.5mg/l.

Cleaning and maintenance of hygienic environment will reduce the risk of infection. Uneaten food, fish excrement, etc must be siphoned out to prevent fouling of water.

The newly hatched larvae, 5-7 mm in size and 1.2 – 3 mg in weight can be kept in the incubator, and do not have to be fed as they rely on the food resource within their yolk sac for the first few days.

The healthy larvae tend to stay in the dark therefore should never be exposed to direct sunlight. In the hatching tank the water inlet should be covered so that the healthy larvae could gather in the dark. The egg remnants crippled and dead larvae are easily removed by siphoning without causing stress to the larvae. After hatching the larvae can stay in the aquarium for 1-2 weeks.

The optimum temperature for larval rearing is 28 - 30oC. Within the next 2-3 days after hatching (48 hours at 28oC) the yolk sac is absorbed and the hatchling is visibly developed into small cat fish and this fry starts to search for food at this stage this fry must be fed on external feed for its further development and survival.

The first feeding is with Artemia nauplii, the success of the intensive production of fingerlings of the African cat fish is greatly dependent on the use of this Artemia as first feed. Shortly before the yolk sac is fully absorbed the larvae are first fed with live Artemia nauplii. The feeding response of these larvae is stimulated by the movement of the nauplii in the water.

The Artemia is administered 6 times per day at regular intervals, this intervals can be adjusted by checking the larval stomach content after every feeding. The introduction of dry feed does not commence until after 3 days of first feeding with artemia. Replacement of these dry feed is usually gradual and commonly accomplished by co feeding with Artemia which is gradually withdrawn and replaced with commercially available dry feed.

**TABLE FISH PRODUCTION**

Table fish production involves the rearing or growing of fingerlings or juveniles to adult fish for human consumption. It lasts for a period of 4 – 6 months depending on the culture system and adoption of good management and adequate feeding protocols

**PROCEDURE**

 Check how much money you have and want to invest

 Decide on the species you want to culture and the culture system to be adopted

 Calculate the daily water requirement for the quantity of fish to be produced.

 Ensure that provision for supply of adequate good quality water is put in place.

 Prepare culture unit e.g. earthen ponds, concrete tank, fibre glass tanks etc. for stocking

 Impound the unit with good quality water

 Stock with right number of fish fingerlings/juveniles of same sizes and density.

**PREPARATION OF PONDS OR TANKS**

For earthen ponds, dry the pond bottom till it cracks. De-silt the bottom if silted. Lime the pond at the rate of 1000kg Agric lime/ ha. Weed the dykes and repair all damaged pond structures. Fill the pond with water and fertilize using a combination of organic manure (poultry) and in-organic manure (NPK). Leave for 3-5 days before stocking

For concrete tanks, wash several times to remove excess cement which will increase pH. Fill tank with water and leave to stand for 3 days. A pH range of 6.5 to 8.5 is ideal for stocking. Check if tank wall or water pipes are leaking and affect all repairs. For plastic of fibre glass tanks, wash thoroughly and fill with water. Check for water leakages

**STOCKING RATE FOR DIFFERENT CULTURE SYSTEM**

|  |  |
| --- | --- |
| **CULTURE SYSTEM** | **STOCKING RATE** |
| Pond system | 5-20 fish/m2 |
| Flow through (partial ) | 50 – 120 fish/m3 |
| Flow through (continuous ) | 200 – 300 fish/m3 |
| Recirculation system | 200 - 400 fish/m3 |

The above rates are to guide the farmer. There may be increase or decrease in the rates.

**Fish Ecology**

This is described as the interaction of fish species with their biotic (living) and abiotic (non living) in a defined natural or artificial environment for mutual and balanced co existence.

**PLANKTONS**

These are mass of tiny floating organisms usually made up of tiny animals and plants floating in the sea, lakes or ponds usually near the surface, and eaten by fish and other water animals.

**PHYTOPLANKTON**

These are very small free floating plants a typical example is one celled algae found in the planktons. In open waters photosynthesis is performed by phytoplankton with sunlight and nutrient in order to grow. Usually the sunlight for photosynthesis is not a problem however shortage of nutrients especially in epipelagic zone is low in nutrient because organic debris (such as dead animals) sinks to much greater depth. Occasionally some nutrients are brought up from the ocean depth by upwelling, storms and ocean current. In this areas, phytoplankton grow rapidly and can become so numerous that the water turns green from their chlorophyll the pigment that gives land plant their color. These areas are the most productive in water supporting billions of aquatic life

**ZOOPLANKTON**

Microscopic animal present in planktons such as protozoas, phytoplankton are eaten by these zooplankton. The most abundant zooplanktons species are copepods and krills others are tiny crustaceans, jelly fish, larvae of fish marine worms, star fish and other marine organism. These zooplankton are however consumed by a huge variety of other animal in water. They range from small fish like sardines, giant marta rays, whale sharks and some sea birds also feed exclusively on zooplanktons.

**NEKTONS**

These are organisms living in water and these organisms can actively swim against the current typical examples are fish, marine mammals, penquins. Shell fish (lobsters, cray fish, crawal fish, shrimps, oyster, mussels, cockle, whelk etc)

**BENTHOS**

These are organisms both animal and plant that live on or in the sediment at the bottom of a sea or lake or deep water examples are clams, mussels.

**MACROPHYTE**

These are large water plants on water surfaces these are seen without the aid of a microscope examples water hyacinth etc.

**CHEMICAL PARAMETERS**

**(A) Dissolved Oxygen** (D.O): This is by far the most important chemical parameter in aquaculture. Low levels of D.O. have been responsible for mass fish deaths either directly or indirectly. Levels of D.O. in fish pond water is a function of several factors including temperature, salinity, stocking density, duration of the day with level of phytoplankton in water etc. The higher water temperature and salinity the lower dissolved oxygen.

For fish, D.O. levels between 5ppm and 10ppm or mg/liter is considered safe while between 3ppm and 4ppm is the caution level. D.O. levels below 2ppm are lethal. At 3ppm, catfish with a full grown arborescent organ can survive. When low D.O. occurs, fish begin to come to the surface to “pipe” or go close to a source of fresh in- coming water. D.O. could be increased by using mechanical aeration method like paddle wheels or strippers. D.O. could be measured on site, using D.O. meters that have been calibrated

**(B) pH**: This is a measure of the acidity or alkalinity of water. The pH scale is from 1 to 14. A value of 7 is considered neutral while below 7 is acidic and above it is alkaline. Acceptable range is between 6.5 and 8.5. Different pH levels have their implications on fish growth as shown below

**pH Effect on Fish**

4 Acid death point

4.5 No reproduction

5.0 -6.5 Slow growth

6.5- 8.5 Desirable range for fish production

9 -10 Slow growth

11 and above Alkaline death point

Most cases of hatching failures have been associated with low pH and softness of water from the source. Calibrated pH meters could be used in measuring pH of fish pond water. Low pH could be adjusted, using calcium carbonate or sodium bicarbonate. Very high pH could be adjusted to the normal range, using aluminium sulphate at 1ppm, to remove 1ppm of alkalinity which is also a reflection of the pH.

**(C) Alkalinity**: This is the capacity of water to neutralize acids without an increase in pH. This parameter is majorly a measure of the bicarbonates and carbonates. Alkalinity has a strong influence on productivity of fish ponds. It has been demonstrated that alkalinity of 100-250 mg/litre was the best for optimum productivity in fish earthen ponds.

**(D) Hardness:** This is chiefly a measure of the calcium and magnesium ions in water. A sample of water is considered to be soft when the measure of hardness is below 50 ppm. Most soft water samples are acidic. While those that are hard are alkaline, i.e. with pH above 7. Fish in soft water (very low Ca2+), tend to lose Na+ and K+ and would have to spend some energy to re-absorb these ions back into the body, hence poor weight gain. Calcium carbonate or ground agricultural lime (limestone) could be used in increasing water hardness.

Experience has shown that excessive hardness of water at about 300-400 ppm or more will not support hatching operations, though juveniles of catfish bought from other sources would still thrive on such farms. At the hatchery level, zeolite (hydrated aluminium silicate mineral or volcanic ash) could be used as a means of reducing the level of calcium and magnesium ions.

(E) **Ammonia:** Fish excrete ammonia and less amount of urea into water as waste. Two forms that occur in water are the unionized ammonia (UIA) and the ionized ammonia (IA). Both are referred to as total ammonia nitrogen (TAN). Temperature and pH do affect the proportion of ammonia that is toxic (IA), and here the lower the pH, the better. The UIA concentration of 0.4 to 3.1 ppm within 96 hours has been shown to be toxic to catfish, while lower concentrations depress growth rates. High ammonia destroys fish gill tissues before leading to death.

Up to 25 times the water concentration of UIA can be found in fish tissue because of high rate of absorption. Build-up of ammonia is more of a problem in intensive or super-intensive culture system. To prevent ammonia build-up, there is need to avoid over-stocking, over-feeding and ensure there is proper oxygenation of the system. This will help convert ammonia to nitrite (which is toxic) and ultimately to nitrate which rarely causes problems in catfish at concentration below 300ppm.

Where level of ammonia is high from source, such water sample should be passed through filters containing activated charcoal for proper adsorption before use in fish hatchery

(F) **Nitrite:** By the process of biofilteration, some bacteria convert ammonia to nitrite (which is toxic) and further convert nitrite to nitrate. Nitrite poisoning in fish is very lethal as the nitrite combines with haemoglobin to form methaemoglobin which cannot take up oxygen .This leads to anoxia and death. This condition is called “Brown blood disease”. Where the sign begins to show in stressed fish, it starts “piping”. The first step to correct this condition is an immediate water change before the use of salt (NaCl) based on the principle of competitive inhibition of nitrite at the gill epithelium by chloride ions.

**(E) Temperature:** Right from the developmental stage of fish embryo to the adult stage, temperature plays a major role in regulating metabolic processes in fish which is poikliothermic animal. The higher the water temperature, the lower the level of dissolved oxygen. At a lower water temperature, the feed consumption and metabolism equally becomes lower.

**(F) Turbidity:** This is a measure of the absorption of light passing through water. Light penetrates only a short distance in highly turbid waters. A secchi disk is used in measuring turbidity and the measure of transparency is an indicator of the degree of fertilization in earthen ponds.

Phytoplankton (which is vital for oxygen production by photosynthesis) and zooplankton in earthen ponds have their own roles in this system and are measured by different means. However in intensive/super-intensive re-circulatory system these have no place as formulated fish feed pellets are consumed by fish and aeration units are available. The biological aspect that is important in the super-intensive re-circulatory system is the microbes like fungi and bacteria. The levels of these organisms could build-up dangerously in a closed system if not checked. This is the reason why U.V.radiation and ozone are used as a means of controlling these.

**FEEDING METHOD**

 In fish culture, provision of well balanced diet for the fish is very important.

 Good quality fish feed must be nutritiously balanced having a crude protein content ranging from 40 – 45% for catfish and 28 -30% for tilapia.

 For pond culture, feeding spots must be identified and adhered to when feeding.

 Always observe the response of the fish when feeding. If the fish is not accepting the feed, stop feeding and determine the cause of low appetite.

 Fish can be fed either manually or using automatic or demand feeders

Hints on how to construct a feeding table:

 Know the average weight of your fish in grams

 Know the total number of fish in the tank

 Use the two above to get your total biomass in kilogram or gram

 Know the recommended percentage body weight you want to feed for the next two weeks. Feeding rates for catfish production ranges from 2 – 6% total body weight

 Adjust the feeding ration every fortnight. Feed 2-3 times daily for pond and flow though system and 4 – 6 times daily for water recirculation system.

 Keep record of quantity of feed given daily for each tank or pond

 Record total feed purchase and total feed given monthly

**FEEDING TABLES**

**Temp** **Fish size (g)**

1-10g 10-25g 25-50g 50-100g 100-300g 300-800g

16 1.0 0.6 0.4 0.3 0.2 0.2

18 3.0 1.6 1.0 0.8 0.6 0.5

20 5.0 3.0 2.0 1.5 1.2 1.0

22 6.8 4.5 3.0 2.4 2.0 1.7

24 8.1 6.0 4.0 3.0 2.5 2.2

26 9.5 6.6 5.0 3.6 3.2 2.8

28 10.0 7.0 5.5 4.0 3.5 3.1

30 9.8 6.8 5.3 3.7 3.2 2.9

32 9.5 6.5 5.0 3.5 3.0 2.8

**FISH SAMPLING AND GROWTH MONITORING**

 This is an integral part of fish production management, the farmer must know when to sample, why he is sampling and what he hopes to achieve by sampling.

 Fish sampling is important in different culture systems and it is dependent on the species of fish involved.

 In the catfish production, sampling is carried out to reduce cannibalism which is occasion by increase or size differential which can lead to the depletion of the standing stock.

 In Tilapia production, sampling is done to separate male and female fry for stocking for increased production.

**Methods of Fish Sampling**

 Weight the fish sample

 Count number of fish in the sample

 Calculate the average weight and total weight

 Calculate daily growth rate (g/day)

 Calculate weight gain

 Calculate food conversion ratio

 Cross – check with a standard table (Coppers or Durante feeding table).

**BASIC HUSBANDRY ON FISH FARM**

 This covers in general terms:

 Handling of stock with minimum stress

 Management of accommodation and environment

 Feeding and prevention of diseases

 Harvesting

 Record keeping

 Appropriate use of equipments

 All the above are essential aid to good management

**SITE OF STOCK**

Must be done in such a way that the risk of disease transmission between generation is minimised e.g fallowing of sites regularly especially at the end of production cycles. This may be difficult to achieve this when resources are limited and considering when the grow-out periods is long. However the benefit of fallowing is enormous considering where infectious diseases are enzootic.

**SOURCE OF WATER**

There are 2 sources of water for fish farming:

Surface water (river lakes etc)

Ground water (well, bore hole)

Surface water from rivers are most common sources of Furunculosis (bacteria infection caused by *Aeromonas salmonicida.* However Ground water supplies are free of this risk but may be a more expensive source because it has to be pumped and breakdown may require a back up and other expenses associated with pumping e.g electricity

 The problem of ground water is the frequency of super-saturation with dissolved gasess (O2, Co2, N2) which can lead to gas bubbles disease in hatched fish.

 Water –flow for fishes should be sufficient to meet O2  requirement as well as remove metabolic waste (Faeces and surplus feed)

 The long-term health problems due to poor water quality are rare in areas of rapid flushing but sedimentary conditions on heavily used sea sites in areas of poor water exchange can deteriorate to the point where gas production poses a threat to fish e.g in sea cages, net fouling not be allowed to impede the passage of water. Therefore simple monitoring methods can provide useful warming to potential problems and thus prevention of stock losses

**TANK HYGIENE**

Tanks should be cleaned daily to prevent accumulation of organic materials, dead or dying fish must be removed. Regular examination of fish for ecto-parasites should also be part of the daily routines.

Cleaning is usually done manually but can be supported by flushing. Circular designed tanks usually aid self-cleaning flows with centrally located drains. It should be noted that high water-flow rates stress fish.

**STOCKING DENSITY**

In the real sense there is no “correct” stocking density for commercial fish farmer. The goal of any fish farm is to extract acceptable returns from investment which can be easily calculated, while the biological need is to provide optimal condition for the stock. It may be difficult to reconcile the two needs. Physical condition at the site will also be a factor. From the point of view of husbandry and fish health, the following should be considered in relation to stocking density.

**CONSIDERATION IN RELATION STOCKING DENSITY**

 Water quality and exchange rate

 Current strength

 Spread of infectious diseases

 Total biomass on the farm

 Peer competition for feeding

 Available surface area in relation to number of fish

 Procedures which involve crowding, e.g Rx for lice, grading.

 Water quality and exchange rate

 Current strength

 Spread of infectious diseases

 Total biomass on the farm

 Peer competition for feeding

 Available surface area in relation to number of fish

 Procedures which involve crowding, e.g Rx for lice, grading.

Reducing of stocking density will improve individual fish performance (preferable all things being equal) but beyond a certain point, total yield will fall below commercially acceptable levels.

**GRADING**

This is the most common procedures on a fish farm and is also one of the major causes of stresses on fish. The reasons for grading are:

 To put peer groups into narrow weight ranges.

 To reject deformity or undersized fish.

 To remove fish which are unlikely to grow well

 To thin stock while biomass increases.

 To set up population for transfer to on-going facilities

 To count fish into new tank or cage

 To control numbers.

Grading usually carried out several times, either by hand or machine. Grading involves crowding the fish often at high temperature which may result in large numbers of deaths.

Overcrowding fish during grading should not be done over a length of time. In sea cages fish can be bruised against the net or cages and may experience a period of low oxygen especially if the high temperature. The damages can also be inflicted by equipment not properly constructed therefore the surfaces of equipment should be properly smoothened

In-order to avoid abrasion. Outbreaks of diseases often follow grading, and should be anticipated. To minimise the need for repeated handling and fish farmer should incorporate other husbandry procedures e.g Vaccination, inspection for ecto parasites etc.

**REMOVAL OF MORTALITY**

Frequent removal of mortality is one of the most important measures in disease control in any intensive husbandry system, for the following reasons:

 Early detection of rising mortality

 Removal of source of continuing challenge in an enzootic

 Regular supply of fresh pathological materials for disease diagnosis.

 Indication of the efficacy of disease control measures e.g anti-bacteria, vaccination

 Reduction of self-pollution and the discharges of organic matter into the environment.

 More reliable assessment of stock number.

A special problem may arise especially when fry are 1st introduced to floating pen, they remain on the floor thus making the removal dead fish very difficult. However at the growth stage they spend less time on the bottom and mortality removal becomes easier.

**WAYS OF REMOVING MORTALITY**

Use of divers can guarantee that all mortalities are removed, but there are doubts about the safety of frequent of diving.

**Raising net floors:** The net floor may be raised so that dead fish can be lifted out in a hand net. This method is labour intensive and it crowd the fish. It usually possible to reach all the fish.

**Dead sock:** The net floor incorporated as a central trap or “dead sock” into which most of the dead fish eventually roll. The floor is raised and the dead fish are removed from the

**Sock with hand net:** Sometime in sea net cage tidal movement may distort the net, so that mortalities may accumulate in the corners.

**RECORD KEEPING**

Accurate and complete stock records are essential to good management and should include the following:

 Stock origin

 Stock number

 Mortalities and their causes

 Disease investigation reports

 Growth data and feeding details

 Treatment records and medicine withdrawal periods

 Medicine stock records

 Environmental data, including water quality.

The records constitute a valuable history for the veterinarian who may not have frequent contact with the farm. By careful examination of record, it is

Possible to detect trends before problems become intractable and to target actions most effectively. Close familiarity with husbandry practices on a particular farm is essential if an effective veterinary service is to be provided. The efficacy of therapy should be monitor closely , to avoid unnecessary and wasteful use of medicines.

The major disease condition encountered in aquaculture are caused specifically by changes, or deterioration, in the aquatic environment and many other conditions are precipitated or exacerbated by environmental effects. The majority of disease condition in aquaculture will be significantly reduced if proper attention is paid to good husbandry and to the maintenance of optimum environmental conditions especially water quality.

Diseases condition can be broadly split into 2 categories: Non infectious and infectious.

The non infectious disease includes the direct effect of all environmental factors on the health of the fish.

**DISEASES OF FISH**

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The non infectious disease includes the direct effect of all environmental factors on the health of the fish.

It must be noted that outbreaks of infectious disease may be precipitated by adverse environmental effects, which include any “stress” acting upon as result of change of physical environment or management of fish themselves, including handling, grading, crowding and administration of drugs.

Non infectious causes of fish diseases:

**Direct environmental effects:**

**Temperature**

High temperature above the optimum temperature range of fish result in a fall in dissolved O2 causing respiratory distress, particularly if the respiratory capabilities are already compromised by the presence of established gill disease. This could result to acute mortalities this situation highlight the complex relationship between the environment and manifestation of diseases.

A sudden change in temperature can precipitate outbreak of infectious disease, perhaps because the pathogens adapts more rapidly than the immune system of the fish to the changes in temperature. Similarly sudden temperature changes during egg incubation can result in the developmental abnormalities.

**Direct environmental effects:**

**Oxygen**

Sources of o2 in fish cultures are plant photosynthesis and diffusion from the atmosphere (limited). Minimum level of 5mg/litre is considered ideal however higher amount may be required for hatcheries. In the night photosynthesis in plant does not takes place therefore overnight de-oxygenation and to certain extent clogging of the gills which eventually result in gasping of fish especially at night.

**Carbon dioxide**

This gas is essential for phytoplankton growth and is usually present as a free gas or in bicarbonates, carbonates and organic forms. Sources includes diffusion from atmosphere, inflowing underground water, decomposition of organic matter and respiratory waste of fish and other aquatic organisms. It is eliminated by chemical combination, diffusion

**Carbon dioxide (contd)**

Into the atmosphere, and use in photosynthesis. High level of free Co2 can cause problems which to produce acidic pH due to dissociation reaction of gas in water:

H2 0+ Co2--------- H++ HCo3---------- 2H+ + Co42-

high level of Co2 in water usually interfere with oxygen uptake and can also cause nephrocalcinosis a condition where calcium carbonate is deposited within the kidney tubules and for which there is no treatment.

**Ammonia**

This is the primary nitrogenous metabolic waste products of fish, but is also formed by the decay organic matter. High level is indicative of overstocking or overfeeding. The un-ionized ammonia (the toxic form) will cause primarily cause direct gill epithelial damage with consequent hyperplasia and reduced ability to take up oxygen. Depending on the spp of fish it can

Also cause liver, kidney and brain damage with reduced activity and growth. Low level of ammonia also causes chronic stress. The level of ammonia varies with pH and temperature being minimise by low values of both parameters. Ammonia is usually a problem to fish in culture where plants are present, as it is used as a nitrogen source by plants. Therefore ammonia thus becomes problem where there are too few plants, and where there is insufficient flow to carry away excess. In such situation ammonia removal is very necessary.

Biological filters incorporated bacteria necessary for the conversion of ammonia to nitrates. The recirculatory system contains these filters with large surface areas achieved by use of specially designed mouldings which allow bacteria growth. These bacteria require some to grow in the filters so as to meet up with ammonia load.

**Food Protein**

Food and faecal fish metabolic waste

NH3-----------------------------------🡪 loss to the atmosphere

NH4\_nitrosomonas>>> NO2> nitrobacter NO3> denitificatn N2

plant\_<\_\_\_\_\_\_\_\_\_Assimilation\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_NO3>

**pH**

This the measurement of the level of hydrogen ion (H+) present in water. It is related directly with to the hardness and alkalinity or buffering capacity of the water and should be maintained within limits tolerable to spp of fish. If the pH is allowed to vary significantly, stress-related problems may become apparent. The optimum pH for most spp is between 6.5-8.5 out of this values toxic effects can occur and stress level will be high. The most damaging situation is a sudden change in pH and this can occur especially in areas affected by acid rain where a flush of low pH water enters the aquaculture facility. Heavy metal are more soluble in acid water , and consequently heavy metal toxicity can be associated with acidity problem.

**Infectious Diseases of Fish**

**Viral diseases**

The source of infection is usually from farmed or wild asymptomatic carriers in the watercourse: usually viral shedding and clinical disease may not be seen until the fish become stressed by movement, crowding, temperature rise etc. Virus can be carried by non susceptible spp of fish and other aquatic mammals and birds; movement of these spp, along with movement of susceptible spp between different water-course, play an important role in the epizootiology of viral infection.

Transmission is usually horizontal between fish where the principal routes of infection are skin abrasion, the gill and the gut; vertical through the egg from infected broodstock to their to their offspring.

Most economically important viral disease of fish include:

 Infectious pancreatic necrosis (IPN)

**Infectious pancreatic necrosis (IPN):** A serous disease of first feeding trout fry and it occurs when asymptomatic carrier become stressed from crowding, transportation. (stress mediated IPN). It is cosmopolitan in distribution especially among freshwater and marine fish and also in invertebrates.

Cx signs ; inappetence, darkening ascites and exophthalmia, loss of equilibrium and trailing faecal cast. Mortality usually up to 90% in very young first feeders but the disease is less severe with the increasing age of fish.

Pm: Ascitic fluid present, the gut is usually filled with white exudates; there may be occasional haemorrhages over the viscera.

**Infectious haematopoietic necrosis (IHN)**. This virus is the same group as the viruses causing Viral Haemorrhagic septicaemia (VHS) and spring viraemia of carps (SVC).