The MPIK Surgery Protocols

Preparation of Implants

The first thing to check, when the date of a surgical procedure is determined, is to examine whether the scheduled procedure involves the usage of implants. If implants will be used, then the existence and the status of each implant must be checked before any other preparation.

<u>**Titanium** must be chemically passivated.</u> In fact any unanodized machined implant that will be in contact with living tissue must be passivated. Apparently the machining process produces surface contamination that can eventually cause pitting and corrosion. To passivate place Titanium (NOT anodized) in 20% by volume nitric acid, at 120º-140º F for 30 min. If you use 316L stainless steel (NOT anodized and not electropolished): 20-40% by volume nitric acid at room temperature for 30 min.

Eyecoils, which are made of Teflon-coated, platinum-iridium wire, are assembled on a stringer and placed in a stainless-steel basin for safe keeping until they are sterilized. Sterilization of the eyecoils occurs 45 minutes before they are used in the surgical procedure. This is discussed more completely under Preparation of the Surgical Suite. The connector to be soldered onto the free (non-implanted) end of the eyecoil is sterilized with ethylene oxide (see Ethylene Oxide Sterilization Procedure).

Headposts, made of surgical-grade titanium, are constructed for each monkey individually based on MR images. Prior to surgery, the appropriate headpost is selected, placed in the craniotomy instrument pack and sterilized in the autoclave.

Open and ball-and-socket chambers, made of surgical-grade titanium, are constructed for each monkey individually based on MR images. The two chamber type must be handled differently.

The open chamber, itself, is placed in the craniotomy instrument pack for sterilization in the autoclave. The open chamber cap, and the screws for securing the cap are sealed in a gas-sterilization bag and sterilized with ethylene oxide. The chamber, its cap and screws, and the two insert are packaged in a gas-sterilization bag and sterilized with ethylene oxide.

Open chambers differ only in the angle of the opening that interfaces with the skull (over the craniotomy). The selection of which one to use depends on the location of the desired recording site and the angle with which one intends to approach it. Chambers with a flat bottom afford a roughly perpendicular approach to dura and cortex, while those with an angled interface also result in approaching the dura and cortex at an ever decreasing angle.

The ball-and-socket chamber: The position of the chamber will determine which type of Gimbal (ball-and-socket) chamber to use. Currently there are two kinds that can be distinguished by the base of the chamber. One has a base that is perpendicular to the axis of the guide tube, the other is at an angle. For all of the placements, the chamber remains upright and as close to vertical as possible allowing the greatest amount of the chamber base to come into contact with the skull. In general, medial chamber positions will use the chambers with the flat base and lateral or posterior chambers will require those with an angled base.

The general preparation of a starts with thoroughly cleaning each part of the chamber. The chamber is then reassembled and the ball is positioned at (0,0) and fixed in place. A guide-tube of the appropriate length, which will pass through the center of the ball, is cut from surgical-grade, stainless-steel hypodermic tubing (18 gauge, Thin Wall) and its ends are rounded and smoothed to prevent excessive tissue damage during placement. The guide-tube is passed through the ball and secured in place such that 1.5mm remain above the external surface of the ball. The chamber is then inverted and fixed in a holder. Silicon elastomer is prepared with the appropriate consistency and poured into socket of the chamber, around the guide-tube, until the surface of the silicon is flush with the bottom of the chamber. The elastomer is then allowed to polymerize, forming a seal around the guide-tube and at the junction of the ball and socket. Two guide-tube inserts are also prepared for use during surgery. Both are cut from surgical-grade, stainless-steel tubing (21 gauge), and glass is melted in both ends of both tubes such that they are completely sealed. One of the inserts is straight, with a slight bend to prevent it from slipping while in the guide-tube, and is used during the placement of the chamber. The other, in addition to the slight bend, is bent at a 45° angle 2 to 3mm from one end. This insert is placed in the chamber once it has been secured on the animal's skull and remains there while the chamber is not in use. The chamber, its cap and screws, and the two insert are packaged in a gas-sterilization bag and sterilized with ethylene oxide. Materials Needed and Procedure are outlined in detail below:

- ❑ 18 Ga. thin wall stainless steel tubing (HTX18-TW; Small Parts, Inc.)
- ❑ 21 Ga. stainless steel tubing (HTX21; Small Parts, Inc.)
- ❑ Corning glass powder (7570)
- ❑ Medical Elastomer (6382 RTV Silicone Elastomer; Factor II)
- ❑ Elastomer Catalyst (6382 Catalyst; Factor II)
- ❑ Silicone Fluid (200 Fluid; Dow Corning)
- ❑ Sharp file, round file, whetstone, flat-head pliers

Selection of a chamber: Once a chamber is selected, disassemble it and clean all of its parts. Make sure that the screws are not stripped and are in good condition. Clean all parts of the chamber thoroughly using Nolvasan or Betadine, rinse the parts first with hydrogen peroxide or alcohol, and rinse them a final time with alcohol to insure that they will dry completely. Reassemble the chamber and use the positioner to place the ball at coordinates (0,0). The next step is to make an appropriate guide-tube and secure it in the ball.

Construction of a Guide Tube: The length of the guide tube will be determined by the depth of the structure of interest. Generally, it is desirable to have one end of the guide tube 7 or 8 mm above the recording site and the other end extending 1 - 2 mm above the top of the ball joint (do not forget to take into account the thickness of the skull, generally estimated at 2 mm in adult monkeys). The guide tube is 18 G thin-wall, stainless steel tubing. Cut the tubing by scoring it with a sharp file and bending it rapidly and sharply with flat-head pliers. Make sure the ends are perfectly straight and that there are no burrs on the outside or inside of the end which will contact tissue. Blunt and taper the outside ends of the tube using a fine whetstone or file. Reassemble all of the parts and make sure that travel of the socket is not restricted.

Construction of a Guide Tube Insert: A rod (guide-tube insert) must be inserted into the guide tube to keep tissue and blood from clogging the guide tube. For this we use 21 ga. stainless steel tubing. The piece of tubing used to make the insert should be several millimeters longer that the guide-tube such that it extends just past one end and 3-4mm beyond the other. Two different inserts are required: one for use during position the chamber, and which is left in the chamber once the procedure is finished. The first is straight with the exception of a slight, gradual bend near the middle. The second is not only slightly bent in the middle, but also has a 45-90 degree bend about 3-4 mm from the one end (the end which will not go into the tube or brain). Make the 45-90º bend with a flat-head pliers, being sure not to kink or break the tube. To seal the ends of the tubes, pack them with the Corning glass powder (the same used to make electrodes) and heat them over a flame. When the sharply bent tube is placed in the guide tube the straight end should be flush with, or extend a fraction of a millimeter beyond, the end of the guide tube that will be in the brain. It is a good idea to make an extra bent insert for each prepared chamber to have around should something happen to the original. A second insert is required in any case for positioning the

The Silicone Gasket: The space between the tube and the chamber wall must be sealed with a gasket made of silicone that fills the cavity at the bottom of the socket joint. The primary component of the gasket is silicone elastomer which is thinned with silicone oil and then polymerized using a catalyst. Determine the proper ratio of elastomer/oil/catalyst by mixing up several different batches. In the past a reasonable starting point has been: 1 cc of elastomer, 5 cc of silicone fluid, one small drop of catalyst. This process is necessary only because the ratio does not seem to remain constant over time, due to the age of the materials, and may also be effected by other factors such as temperature. The gasket must be flexible enough to allow the guide tube to move to any position without tearing yet solid enough that it will remain in the cavity. Essentially, the elastomer must be solid but not sticky.

Mount the chamber upside-down in a holder so that the base of the chamber is level. Mix the elastomer well and fill the cavity using a syringe and needle. Fill the cavity slowly and carefully to prevent the formation of bubbles and allow the elastomer to polymerize.

Sterilization of the Chamber: Completed Ball-and-Socket chambers are sterilized with ethylene oxide 2-3 days prior to surgery. Once the sterilization process is finished, the chambers, in their bags, should be set in an open but protected area to air. This is necessary because the silicone gasket absorbs ethylene oxide during sterilization and the gas, which is highly toxic, diffuses out later. Thus, in order to avoid tissue damage and death in the region of the implanted chamber it is good to minimize the amount of EtOX that will diffuse into the sealed region postsurgically.

Preparation of the Stereotaxic Devices

Stereotaxic positioners and selection of the chamber site: Stereotactic positioners are used during the procedure to both determine the position of the craniotomy, and to hold the chamber while it is being secured to the monkey's skull. This procedure should be done prior to sterilization with the chamber to be implanted. All of the stereotaxic equipment required during the procedure must be sterilized with ethylene oxide.

Determination of the Stereotaxic Coordinates: We use the coordinate system established by Horsley and Clark described in "A Stereotaxic Brain Atlas for Macaca Nemestrina" by W. D. Winters, R. T. Kado and W.R. Adey, University of California Press (1969). This system uses a horizontal reference that is 10 mm above the Frankfurt zero for the horizontal plane, defined by points connecting the infraorbital ridge and the middle of the internal auditory meatus. The anteroposterior zero reference is perpendicular to the horizontal plane and passes through the interaural line. The placement of the chamber is critical so be extremely cautious during this procedure. In short, one must determine the carrier values for the Horsley/Clark zero and add the values in three dimensions to reach the desired structure. The dimensions are the three axes, anteroposterior (AP), mediolateral (ML) and dorsoventral (DV) which are normally oriented in orthogonal directions however, these may not be exactly 90 degrees apart if the electrode is not intended to move along just one axis.

Place the L-shaped ear bars into the stereotactic device so that the ends meet at Horsley/Clark zero. Place a carrier (stereotactic positioner) with a pointer (a straight pin) attached on one of arms of the stereotaxic holder. Mark the zero position of the pointer by marking the AP, ML and DV coordinates of the carrier when the tip of the pointer is at Horsley/Clark zero.

Move the position of the pointer to the intended recording site by adding (or subtracting) the values of AP, ML and DV to the values of the carrier zero. The pointer tip should now be in the estimated position of the structure.

Get another carrier, using a U-shaped wire as the pointer, and position it in such a way that the tip of the U-shaped wire meets the tip of the first pointer from underneath. Remove the first carrier and replace it with a carrier holding the chamber (a special holder has been constructed for this). Manipulate the carrier so the tip of the guide tube comes in contact with the end U-shaped pointer, respecting the space that will be occupied by the head of the animal. These are the coordinates that will be used during the surgery. Repeat this procedure to verify that the coordinates are correct. (If this procedure is difficult to follow, use the example in the beginning of the atlas as a practice session.)

The DV coordinates will obviously not be used, however, they are useful for making a secondary check on the placement of the well. Center a monkey skull in the stereotaxic holder leaving the calvaria off. Adjusting the carrier to the coordinates should put the tip of the guide tube in the position of the brain structure. Check this with the internal landmarks in the skull. Raising the guide tube about 9 or 10 mm (depending on the distance between the guide tube and the structure) should put the chamber at its approximate position on the skull. This can be verified by replacing the calvaria and removing the guide tube. The slope of the skull at the point of contact will determine whether to use a flat or angled chamber.

Room Reservation and Equipment Check

Make a reservation of the surgical room before planning a surgery. Although at MPI we have our own private surgical facilities, we still have different investigators, who may decide to operate on an animal the very same day. After making the room reservation, the person responsible for scheduling the surgery must make sure that all of the supplies needed for the procedure are available. All of the items required for a particular surgery are outlined in the **Pick List**.

Room and Equipment Reservation: The surgical suite is reserved for the day of the surgery, but must be available for cleaning the night before the surgery. The autoclave must be reserved for one day before the surgery. Autoclaved packs have a shelf-life of 3 months (when our Aesculap containers with their filters are used), and then they must be resterilized, even if they weren't opened, if the shelf-life is expired. The gas-sterilization equipment (e.g., ethylene oxide) must be reserved for two consecutive days no more than 10 days prior to surgery. Gas-sterilized items have a shelf-life of 7 days and then they must be repackaged and resterilized.

Equipment and Supply Check: Several days prior to the surgery, the surgical gas tanks must be checked to ensure they are functional and capable of delivering the appropriate pressures. This must be done early in case a tank needs to be replaced. All electronic equipment (bipolar and monopolar coagulators, light source, surgical lights) should be checked (powered-on) to ensure that they are in proper working order, and the connections should be cleaned if there is a question as to whether they will make good contact. The bed should be checked to ensure that it is fully operational can be smoothly adjusted. Make sure that all needed stands (2 Mayos, 1 back table, 1 ring stand, 2 prep tables) are available and work smoothly.

Drug Check: Check the availability and expiration date of all required drugs. The drugs most commonly used in or related to surgical procedures are Atropine, Cephatholin, Baytril, Doxapram, Heparin, Ketamine, Naloxone, Oxymorphone, Buprenorphine, Pentobarbital (I), Pentobarbital (M), Lactated Ringer's, Dexamethasone, Lidocaine, Tylenol, etc. See the drug chapter for details concerning their usage.

Administration of Antibiotics

The administration of antibiotic begins three days before, two days before, or the day of surgery and continues for 10, 10, or 7 days depending on the physical status of the animal. Animals of the first class can start taking antibiotics the day of the operation. Dosage and administration is described in the section 'surgical procedure'. Animals of the second class, (which for us are the animals with previous operations or small infections, such as head post infections, infections in the wells etc.), must be administered antibiotics at least three days before the day of the operation. Since in most non human primates intramuscular injections of antibiotics are not effective and IV injections are impossible without chemical restraint of the animals the only effective route of administration is oral. One possibility is to use Bactrim which is available as a pink, cherry flowered pediatric suspension containing in each teaspoonful (5 ml) 40 mg trimethoprim and 200 mg sulfamethoxazole, compounded with 0.3% alcohol. This can be given in a glass of apple or orange juice. Monkeys seem to drink that without problems. Recommended dosage is 1 teaspoonful every 12 hrs. Tribrissen or sulfamethoxazole and trimethoprim suspension is another possibility. We usually administer Tribrissen P.O. at 30 mg/kg. In case of intracranial surgery there is the additional problem of intracranial pressure. This must be kept to a minimum during the operation, for if the surgical activities or the anesthesia cause it to rise, the ultimate disability which may result from the lesion (either because of a guide tube for electrode or because of putting a small local lesion) is considerably increased.

Administration of Steroids

If the surgery will involve a craniotomy, the animal must receive steroids in order to reduce intracranial pressure prior to the operation. At least one day before the operation animals must be administered Dexamethasone. We usually begin administration, via IM injection, 48 hrs before surgery. Decadron Elixir contains 0.5 mg in each 5 ml and can be administered orally. Dosage is 0.25 to 1 ml/kg and the duration of its action is 36 to 72 hrs. Another possibility is intravenous injection of mannitol as soon as the head is opened. Mannitol (a sugar alcohol which is virtually inert metabolically) is an osmotic diuretic. It induces diuresis by elevating the osmolarity of the glomerular filtrate and thereby hinders tubular reabsorption of water. It is indicated for reduction of intracranial pressure and treatment of cerebral edema by reducing brain mass. It must be, however, avoided if intracranial bleeding is suspected. We do not administer Mannitol in cases when Dexamethasone has been administered.

Administration of Analgesics

Administration of the analgesic (Tylenol) begins one day prior to surgery and continues 3 days after surgery or until the animal returns to normal activity levels and does not appear to be experiencing pain related to the surgery. We also administer Tylenol P.O. the day before the surgery as an analgesic since it is more effective when taken over a period of time.

Restriction of the Animal's diet

The day before surgery, food should be provided at the regular feeding times. Food should be withheld overnight but the animal may have water ad libitum. Water must be withheld 2-3 hrs. before the anesthesia. If, however, it is impossible to water-deprive the animal just 2-3 hrs. before surgery, then it should be deprived overnight .

Disinfection of the Operating and the Prep Room

The night before a surgery all of the equipment is removed from the OR and the Prep Room, and the floor and all fixed, exposed horizontal surfaces are disinfected using … . The disinfectant is wiped onto the surfaces and allowed to dry. The floor is then mopped, using the same disinfectant, and allowed to dry. The person mopping the floor, and all persons entering the OR after it has been disinfected are required to be appropriately dressed (surgical scrubs, shoe covers, mask, head cover, gloves). When the floor is dry, the equipment is placed back in the OR and the OR is marked with a sign to indicate that it is now ready for surgery.

Pulling for the case

The scrub nurse and circulator are responsible for selecting and assembling all items required for a particular surgical procedure according to the *pick-list* for that procedure. The following items are included in the list and should be handled according to the instructions given. All of the EtOX sterilized items should be placed in a clean container which can be taken into the OR during setup.

Drapes/Gowns

All of the required drapes and gowns should be pulled and placed in a container that will be carried into the OR for setup

- ❑ Back Table Cover
- ❑ Mayo Stand Covers
- ❑ Barrier-Flex Transparent Drape (Mud flaps)
- ❑ Incision Drapes
    - ♦ Adherent window w/ fenestration
    - ♦ Adherent window w/o fenestration
    - ♦ Fenestration
- ❑ Fanfold Sheet
- ❑ Utility Drapes

Prep Kit

The prep kit can be put in the container with the gowns and drapes.

Syringes/Needles

The syringes and needles should be collected into a bag labeled **Syringes/Needles** that will also be taken into the OR for setup.

- ❑ Prep Tray Skin Scrub w/Solutions
- ❑ Syringe 1cc

- Syringe 3cc
- Syringe 10cc
- Syringe 30cc
- Needle 22x1-1/2ga
- Needle 20x1-1/2ga
- Catheter 24 ga

Knife, Blades, and Sponges

Knife blades and sponges should be collected into a bag labeled **Knife, Blades & Sponges**

- 1 Blade Scalpel #10
- 1 Blade Scalpel #11
- 1 Blade Scalpel #12
- 1 Blade Scalped #15 1215
- 2 Sponge x-ray 92516
- 1 Cottonoid 1/8 x 6 801449
- 1 Cottonoid ¼ x 6 801450
- 1 Cottonoid ½ x 6 801451

Sutures/Needles

Sutures and needles should be collected into a bag labeled **Sutures/Needles.**

- 1 Stapler 35 Regular 316L
- 1 Stapler Remover
- 1 Chromic 4-0 C566T
- 1 Vicryl 3-0 P-1 J497G
- 1 Opthalmic 6-0 J590G
- 1 Opthalmic 5-0 J591G
- 1 Opthalmic 8-0 J975G
- 1 Chromic 5-0 K895H
- 1 Bone Wax BW102

Miscellaneous/Sterile

These items should be collected into a larger bag labeled **Miscellaneous/Sterile**. The smaller bags (**Syringes/Needles**, **Blades/Sponges**, **Blades/Sponges**) should be placed in the larger bag for convenience.

- 2 Raney Clips 20-1035
- 2 Tubing Suction Connect 284521
- 1 Light Handle Gloves DE3613

- ❑ 1 Pencil Electrosurgical E2515H
- ❑ 1 Polisher for Cautery Tips DE3200
- ❑ 1 Needle Counter 10 count 1812
- ❑ 2 Pen Skinmarker 50-1166
- ❑ 1 Surgicel 1951
- ❑ 4 Eye Spears
- ❑ 5 500cc .9% NaCl

Miscellaneous/Nonsterile

- ❑ 2 Suction Tubing 253-4233
- ❑ 2 Suction Canister 494410
- ❑ 1 Pediatric Bovie Grounding pad
- ❑ 1 Medical Adhesive spray
- ❑ 1 Silk Tape 3 in
- ❑ 1 Solder Iron
- ❑ 1 Solder Material

Instruments/Basins

- ❑ 1 Basin Set
- ❑ 1 Instrument Set Craniotomy Pan
- ❑ 1 Instrument Set Ophthalmic Pan
- ❑ 1 Instrument Set for the Eye Coil operation

Implants

- ❑ 1 DeBakey Felt 8972
- ❑ 1 Eye Coil Set
- ❑ 2 Headposts or Chambers (depending on the procedure)

Equipment

- ❑ 1 Bovie Unit
- ❑ 1 Bipolar Unit
- ❑ 1 Headlight
- ❑ 1 Light Source
- ❑ 1 Clippers
- ❑ 1 Loupes
- ❑ 1 Heating pad
- ❑ 1 Safety Glasses

Remaining items

Collect additional instruments and supplies according to procedural need, including drugs and special items (specialty cart).

Collect positioning devices (stereotaxic device, eyebars, earbars, mouthpiece), positional aids, and special equipment, the microscope (if needed).

Collect the irrigation fluids needed. We usually have one bottle of injectable saline, one of injectable water, and 5 500ml bottles of saline for irrigation.

To avoid time loss, all supplies and equipment should be gathered at one time and re-checked for accuracy by the scrub and circulating persons. Positioning devices, special table pads, suction canisters, connecting tubes, and medications to be administered at the sterile field—all these items are needed during the procedure and must be a part of the initial preparation phase, in order to avoid leaving the room to collect them during the procedure.

ToDo's The Day of the Surgery

Examination and Premedication

Preparation for the actual surgical procedure begins two days (headpost-eyecoil or open-chamber) or three days (ball-and-socket) before the surgery with the preparation of the animal as well as the instruments and equipment required for the procedure.

Before any survival surgery the physical status of animal must be taken in to consideration and appropriate preoperative treatment must be applied. In the Textbook Of Small Animal Surgery (Slatter, 1985) there is a detailed description of the status classification and the corresponding treatment. The following list summarizes this classification.

- Excellent - Usually young healthy animal which have never undergone an operation before.
- Good - Minor fractions, slight dehydration, obesity, prior operation, small infections.
- Fair - Chronic heart disease, anemia open or severe fracture subsequent to trauma, mild pneumothorax.
- Poor - Extreme systemic disturbance, ruptures of urinary bladder, internal hemorrhage, severe airway problems, etc.
- Critical - Animals in moribund state.
- Emergency - This holds for any animal in one of the previous classes that is operated upon as an emergency.

We usually deal with animals of the first or second class, however, prior to undergoing surgery every monkey is prepared according to a standard protocol. This includes the administration of drugs and the restriction of the animal's diet. All animals receive antibiotics and analgesics prior to surgery. If the surgery will require that a craniotomy be made, the animal also receives a steroid to reduce intracranial pressure. The drug chart in this document lists the name, concentrations, and dosages for all the drugs we use.

Psychological and Physiological Considerations

Of particular concern when handling an animal prior to a procedure is the possible effect of stress on the physiological state of the animal. Macaques and other Old World Monkeys appear to be fairly resistant to the stress of handling. Rhesus monkeys are potentially dangerous and at least two people should be present for manipulations. Generally animals should be handled gently and by as few people as necessary. Physiological effects of restraint range from increased heart rate, respiratory rate, and body temperature to metabolic acidosis and even death. The discussion of acute death related to restraint can be divided into three categories: Drug Induced Death, Hypoglycemia, Fatal syncope.

Drug Induced Death

All of the drugs which are used for restraint are respiratory depressants, most are circulatory depressants, and many produce cardiac arrythmias. Restraint in wild animals is often associated with a great deal of excitement on the part of the restrainers and restrainees. There is often great muscular activity prior to and during attempts at restraint. The muscle activity increases oxygen consumption, the increase in activity also increases body temperature which increases oxygen consumption, there is also an imbalance of normal autonomic nervous function. All of these factors place the animal in a precarious position when a depressant drug is added. The animals can die from any of the following: hypoxia, acidosis, hypotension, hypertension, fatal cardiac arrhythmia, marked hyperthermia. When using drugs for restraint it is therefore most important to try to administer them before the animal is too upset. If a great deal of activity has gone on before administration of a restraint drug it is more prudent to allow the animal to calm down before drug administration if possible.

Hypoglycemia

Some animals in captivity respond to what often appears as a mild stress with convulsions and death; captivity and its resultant inactivity and decreased food gathering activity may produce a decrease of glycogen stores in the liver and muscle. When a stress demands glucose (increased catecholamine production, epinephrine, and norepinephrine, muscle activity, increased body temperature) the available glucose is rapidly used up, as is that released by glycogenolysis and the animals become hypoglycemic, convulse and may die.

Fatal syncope

Those who are familiar with restraint of wild animals know the violence of the responses which may occur; they also know of the individual animals which suddenly die during restraint. When one thinks about this phenomenon the areas that immediately come to mind are the sympathetic nervous system, increased levels of epinephrine, hypertension and tachycardia. The typical fight or flight response. There are however many examples of animals which start to fight and suddenly stop and often proceed to die. There are few studies which might give some insight to what might be occurring. Monkeys can be restrained in a chair after having various vessels catheterized and ECG leads connected. These animals can then be monitored for such parameters as blood pressure and heart rate. If such monkeys are given an electric shock after hearing a clicking noise, it can be shown that when the clicking noise starts the animals develop bradycardia and hypotension! Another experimental model used to study stress in animals is to place rats in a glass cylinder containing water, thereby forcing the animals to swim continuously. There is a relationship between the temperature of the water and the duration of swimming. There are, however, always a few rats which die when they are being handled or soon after being placed in the water. When such animals have ECG leads attached it is found that after a brief tachycardia there is a bradycardia that proceeds to cardiac standstill. The relatively common occurrence of fainting (syncope) by some humans in distasteful situations may be a similar but less drastic example of this phenomenon. If the restrained wild animals do die from vagal bradycardia which proceeds to cardiac arrest and death, then pretreatment of such animals with atropine when possible might prevent these deaths.

Preparation of the Sterile Fields

Proper preparation of the procedure room before the case is essential to an effective surgical outcome. It is the responsibility of the scrub person, nurse anesthetist, and circulator to see that everything is ready prior to the acceptance of an animal into the OR. This process should begin 30 to 45 minutes before the scheduled start to ensure there is ample time to get everything done. To avoid unnecessary clutter, furniture and equipment not expressly required for the procedure should be removed.

Figure IV-16 shows the first sterile items to be opened in the positions where they will be opened: the basin pack on the ring stand, the back table cover on the back table, and the scrub person's gown and gloves on a Mayo stand. The remaining sterile items, including the bag of miscellaneous sterile items, surgeon and assistant's gown and gloves, and drapes and covers, are left on the bed until they are to be opened.

Figure IV-16 : Ring stand, back table and Mayo stands with first sterile items

The instrument packs are placed one on each prep table. Once the equipment and supplies have been gathered and placed on their respective surfaces, and the procedure room has been properly prepared, the scrub person can begin the task of creating the sterile fields.

Figure IV-17 : Instrument boxes with external indicators

Before any sterile supplies are opened, the integrity of each package must be checked. Packages not meeting acceptable criteria should be placed aside, since they cannot be considered sterile, and new ones chosen. Each package must be opened under strict aseptic technique, thereby guaranteeing the sterility of the sterile field. Remove the tape of the wrapped package and check the indicator tape to be certain the item has been properly exposed to the sterilization process. The edges of an open package or container are considered unsterile, and sterile items must not make contact with them en route to a field. In the case of the instrument packs, at least one of the external indicator rings (which prevent the lid from being opened and removed accidentally) must be intact. After opening a pack, the internal indicators must also be checked to be certain that the pack was sterilized throughout. In the case of ethylene oxide sterilization, the indicator should be checked when the packages are removed from the sterilizer for aeration. At the time of setup both the integrity of the packages and the indicator on each package should be checked again before opening it onto a sterile field.

The first action of the scrub person is the creation of a sterile field on the back table, i.e. draping it. (described above). The second action is the creation of a sterile field on the ring stand (also described above). Once the two fields have been created, the small, sterile items collected into bags according to the *pick-list* are opened into the basin in the ring stand, with the exception of blades and cottonoids which are opened onto the back table. Next, all of the sterile supplies not in the miscellaneous bag (drapes, gowns, gloves, towels, etc.) that had been assembled on the OR bed, are opened onto the back table. The second to last items to be opened before the scrub person leaves the room to scrub are the scrub person’s gown and gloves.

The gown is opened on a Mayo stand, opening away first, and being careful not to contaminate it. The gloves are then opened and the internal package is flipped onto the sterile field, while maintaining a safe distance from the field. The last items to be opened before the scrub person leaves the room are the instrument packs.

The external indicators should be checked, and at least one must be intact for the pack to be considered sterile. After removing the lid, check the internal indicator as well. Figure IV-18 illustrates the position of the instrument packs in the room during setup (foreground, right). The position of the above-mentioned external indicator can be seen in Figure IV-17 (orange fastener through handle).

The scrub person is now ready to scrub in. Figure IV-18 shows the room as it should look just before the scrub person leaves to scrub. Sterile drapes, etc. are open on the back table, the small items have been opened into the double basin, the scrub person’s gown and gloves are open on a Mayo stand, the instrument packs have been opened, and the prep kit is ready.

Figure IV-10 shows the procedure room from above to illustrate the positions of all the equipment just prior to the scrub person scrubbing in. The Bovie unit is at the foot of the bed, the anesthesia machine is on the same side as the ring stand, back table, and Mayo stand (which are now sterile), and the second Mayo, while the IV stand is on the opposite side. The prep kit and the open instrument packs are at the head of the bed. The appropriate waste containers for uncontaminated, contaminated, and linen are also present.

Because several sterile fields have already been setup in the room, it is important that all persons entering the room are aware of the appropriate traffic patterns for preserving the sterility of the room and avoiding contamination of the sterile fields.

When the scrub person returns from scrubbing, they must gown and glove themselves, with assistance from the circulator during gowning. Both gowning and gloving have been detailed above. Once appropriate dressed, the scrub person finishes the set-up of the sterile fields on the Mayos, ring stand, and back table. The post-scrub set-up has been described in detail above (under the section *Creation of Sterile Fields*) and will not be described in detail here. Briefly, the items on the back table are organized an stacked according to order of usage. Next, the Mayo stands are draped. Once sterile fields have been created on the Mayo stands, all items on the back table are transferred to the Mayo stands. Towels are laid down on the back table (according to the procedure outlined in the above-mentioned section – on flat and two folded) to prepare for setup of the working area. The smaller basins, pans, etc., as well as the small sterile items are removed from the large basin and organized on the back table. When the basin is empty, the stack of towels and drapes is moved from the Mayo to the basin. These items remain in the basin until used.

Figure IV-18 : The operationg room just before the scrub person leaves to scrub

At this point the scrub person is ready to unpack the instrument sets and setup the instrument trays. The setup of the instruments has been described in detail in the section entitled *Instrument Trays and the Working Area of the Back Table* and will only be reviewed briefly here. The baskets containing the instruments are removed, placed adjacent to the working area on the back table, and unloaded one by one. The instruments are placed immediately after unloading in their assigned positions and empty, unneeded baskets are placed off of the field. After set-up is complete, the OR is ready to receive the animal.

Following the setup of the sterile fields, items that were wet sterilized (in a formaldehyde solution, for example) should be rinsed and position on the sterile field. This must be done with the help of the circulator. The only item in this lab that is regularly sterilized in this way are eyecoils, because they can neither tolerate heat nor gas sterilization.

- Rinsing the eyecoils: If eyecoils are being sterilized, and they have been in Cidex for the required 45 minutes, now is a good time to rinse them. The scrub person, using a forceps, should grab the stringer, and, being careful not to touch (or let the eyecoils touch) any nonsterile region of the container, remove them.
- Placing them in the middle working field bowl on the back table. This should be done away from the working field to avoid dripping on it.
- Rinsed them well in at least 3 changes of saline. The waste can be dumped into the largest basin.
- When working with irrigation solution or fluid contained in a pour bottle, careful attention must be used to avoid "splash-back".
- Once the solution has been poured, the container cannot be recapped for future use, since its contents can no longer be considered sterile. The solution must be completely used or the remaining fluid discarded.
- Once thoroughly rinsed, the bowl and eyecoils can be replaced on the working field. Take care not to splash saline while rinsing the coils. If the drapes become permeated or moist, they must be considered contaminated, and corrective action must be initiated to cover the area in question or to change the drapes.

Gowning and gloving the rest of the team

The scrub person must also help gown and glove the rest of the members of the team after they scrub in. To preserve the sterility of the sterile field, all of the items are removed from the sterile field by the scrub person and handed to the person gowning and gloving to avoid having them reach over a sterile surface. The scrub person first offers the surgeon a sterile towel. After the surgeon has dried off, the gown is held up in front of the surgeon in a position convenient for the surgeon to slip into it. The circulator must snap the gown and tie the secondary tie, however, the primary tie can to done by the scrub person. The surgeon is then gloved using the open glove method, and moves away from the gowning site so that the procedure can be repeated with the assistant.

Preparation of the Animal

Shortly after the scrub nurse has commenced setting up the OR, the animal can be restrained and the procedure for prepping the animal can begin. The anesthesiologist, assistant, and surgeon begin the preparation of the animal and bring it into the OR where the circulator takes over after the animal is in the stereotaxic device. This way the circulator is available to assist the scrub nurse during setup, if needed. The job of the anesthesiologist begins with the restraining of the animal. For the purposes of this description, we will assume the surgeon is performing the prep and the surgical assistant is assisting. From the time of the first injection, all actions and vital signs must be recorded an monitored.

Catheter Placement

The catheter is placed in the saphenous vein which runs along the center of the back of the lower leg. It is a good practice to shave only the back 1/3 of the leg or so at first in order to see if a vein will be easily accessed in that particular leg. If not, the back of the other leg should be shaved for the same reason. This way, if no vein can be visualized, one can resort to the arms without shaving all the hair off both lower legs. If one does have to resort to an arm, one should be very aware of the fact that both a ground pad for the Bovie and an arterial line will need to be place on or in an arm as well. The Bovie pad can, however, be placed on a leg as well. It is usually the case, however, that one finds a suitable vein in one of the legs. At that point the lower leg of choice is shaved clean all the way to the knee, and the intended site of penetration is wiped down well with alcohol.

- While the assistant fills a vein cap with heparinized saline, the surgeon removes a 22 gauge catheter from its package and slides the sheath several millimeters back and forth along the needle to ensure that it can be placed smoothly.
- When these two items are ready, the assisting person grabs the leg just below the knee and squeezes tightly, creating a tourniquet with their hand. The surgeon snaps the leg (on top of the vein) several times with their finger/s to help expose the vein. Pumping the foot also helps achieve this**.**
- *Do not begin inserting the needle without first fully visualizing the target vein.* (You must know exactly where you are going with needle before puncturing the skin. Your target is not as big as it appears to be on the surface, because it is covered with skin, fat, connective tissue, etc. which all accentuate its size.
- Your first attempt should always be relatively low on the leg. (If you blow the vein, you can always move up a little and try again. You cannot, however, move down from a failed sited.) .
- Hold the needle at a 25-40 degree angle with the tip a couple mm from intended point of entry. Insert the needle relatively quickly at first, for 2-3 mm, then slow down and decrease the angle. The initial movement must be steeper and faster because veins will roll away from your needle if not penetrated.
- They are not, however, very wide, and you must slow down and decrease the angle to avoid exiting the other side. If you are in the vein when you level off, you should see that blood is flowing up the catheter. If no blood has entered the catheter, you may need to go a little deeper, or to one side or the other, depending on the current relationship between the vein and the needle.
- Once the needle is in the vein, push it as directly as you can up the vein for another 3-5mm, then, while holding the catheter needle with one hand, advance the sheath 5mm or so with the other. This will protect the vein while you advance the whole catheter farther into the vein. Advance both another 5mm using the sheath (luer lock), not the needle insert.
- Pull the needle out a little more while holding the sheath in place, and repeat this until the sheath is in to its hilt.
- Holding the sheath in place, remove the needle, discard it in the sharps box, and cap the catheter with the filled vein cap. Hold the catheter securely at all times. Figure IV-19 illustrates the final placement of a catheter before it is wrapped and secured in place.
- While holding the vein cap, as shown in the figure, flush the catheter with heparinized saline, watching for bulging (fluid accumulation) around the insertion point, indicating a blown vein. The catheter is flushed with heparinized saline after each use to prevent the accumulation of

drugs in it, as well as to keep the line clear. It is important that the placement of the catheter proceed carefully but quickly as it becomes increasingly difficult if the animal starts coming up from the Ketamine and begins moving. Therefore, before the catheter is secure, anesthesia is induced with an initial dose of pentobarbital.

Induction of anesthesia

Because Ketamine is already on-board, a reduced dose of pentobarbital is used to induce anesthesia. The induction dose for pentobarbital is 0.16 mg/kg, however, with Ketamine on-board, it is reduced to 2/3 of that. The dose of pentobarbital is injected into the catheter, and the catheter is flushed with heparinized saline. Pentobarbital is extremely irritating to muscle tissue, so once again it is important to watch the area carefully and be certain that the integrity of the vein and catheter have been maintained. Shortly after the injection of the pentobarbital, one can witness the onset of its action first in the animal breathing. The animal should take a noticeably deeper breath about 10 seconds after the injection, and then breathing should return to a near normal pace. At this point the catheter should be secured into position.

Securing the catheter:

The most likely source of infection where the catheter is concerned is the insertion point, so, before covering it, place a couple drops of betadine around it. Holding the catheter securely in position, elevate it slightly, slip a short (3 inches, 6-8cm) piece of tape under it (sticky side up), and fold each in turn back over the catheter and onto the leg to complete what is called a butterfly. The catheter and cap are then taped down using a transparent dressing (Bio-clusive transparent dressing). Figure IV-20 shows the catheter after it has been secured.

Shaving the Incision and Grounding Pad Areas

One arm or inner thigh will need to be shaved for the grounding pad. Clean away all hair and wipe the area down well with alcohol to clean it and improve contact. The area should also be cleaned with degreaser prior to applying the pad. Depending on the procedure to be done, more or less of the animal's head will need to be shaved. If an eyecoil is being placed, the eyebrows and hair around the eye also need to be shaved. Cut the hair as short as possible using a clippers followed by a razor if necessary. In the case of using a razor, shaving cream should also be used to lubricate the area and minimize nicks and cuts which are also a source of infection. When shaving is finished, use silk tape to remove hair from the area. Expose several inches of the sticky surface, press it down onto the skin, pull it off, and expose some more of the sticky surface. Repeat this until no more hair is picked up. Always be very careful when doing anything around the eyes. Do not let excess fluid of any type run into the eyes, and make sure they are closed when cutting and cleaning up hair. At least one of the eyes, both if the surgery does not involve the implantation of an eyecoil, is closed by placing a lubricant or antibiotic ointment in it and pressing it closed. This protects the eye/s from drying out as well as from the entry of foreign substances. At this point the animal is taken into the OR where the preparation procedure will be completed.

Intubation:

The animal is turned carefully so the mouth can be accessed easily. The mouth is held open with two long pieces of gauze running behind the canines. In the case of smaller animals that have no canines, much thinner pieces of gauze or heavy string should be used to pull the jaws apart. The lower piece should be pulled directly down the sternum, and the upper is used to adjust the angle of the head. The area is first mildly anesthetized.

While the mouth is held open by the assistant, the surgeon places the blade of the laryngoscope on the animal's tongue about ¾ or more of the way back and presses down to reveal the opening of the trachea. A size 0 or 1 curved blade is recommended for the small animals. The opening appears as relatively white or yellowish lips, roughly in the shape of a diamond, on a pink/red background. This view can be seen in Figure IV-21 . When the opening is in view, it is sprayed with cetacaine. The blade is immediately withdrawn and the gauze released to allow the animal to react (cough) freely.

- The appropriate size intubation tube is selected and a conservative amount of Xylocaine is applied to the region of the tip and inflatable cuff. **Always check the tube, using a 10cc syringe, to make sure that it is fully functional and not occluded.** If the animal is stable, proceed.
- Repeat the above procedure for visualizing the opening of the trachea, and be sure you are confident about what you see before proceeding. Injury of the trachea can cause it to become swollen and tight making the placement of a tube very difficult, if not impossible.
- Once the opening is in full view, position the tube along the blade, matching the curvature of the tube to that of the blade. Advance the tube down the blade, and slide it smoothly into the opening without pausing. Push the tube in the appropriate distance (usually roughly to the bifurcation of the small tube leading to the inflatable cuff).
- Holding a dental mirror near the end of the tube, look for fogging of the mirror indicating the tube is in the trachea and not the esophagus. Using the stethoscope, listen to the breathing to be sure it sounds clear and normal. If everything sounds fine, the cuff is inflated and the tube secured.
- Fill the cuff using a 3cc syringe. Do not overfill it. Overfilling damages the trachea and causes a bad soar throat which may effect eating post-operatively. However, the tube should not slide freely when the cuff is inflated. Cap the end of the inflation tube, fold it over to kink it, and clamp it using a plastic, rolling PE-tube clamp. ***From this point on be very careful not to occlude the end of the intubation tube as this is now its only source of air.*** Butterfly the tube to the lower jaw, and then tape down the ends of the butterfly with a piece that goes all the around the animal's neck. The plastic clamp is also secured to the animal's neck with a piece of tape that goes all the way around the animals neck, overlapping itself by several inches. Figure IV-22 and IV-23 show the final, secured position of the intubation tube.

Placement of the animal in the stereotaxic device

This procedure usually requires two people to maneuver the animal into the appropriate position.

- While the animal is still lying on the bed, take one of the earbars and establish how deep is will go in and what its will look like when it is positioned appropriately. One person should help position and support the body while the other grips the head from above, placing the fingertips in the temples, positions the head. The first earbar should be inserted into the ear first. The other end should then be placed into the channel in the stereotaxi and the screw tightened. At this point establish that the first bar is appropriately placed.
- Find the zygomatic ridge with your finger. Following it to the ear should lead you directly to the inserted earbar. If the earbar is in, hold the animals head securely in place while the other earbar is inserted and secured. After the second bar is securely in place, the animal's

head-holder used to immobilize the head durin

Starting gas anesthesia and reaching surgical depth

The animal is now connected to the anesthesia machine, and the depth of anesthesia is increased to surgical levels.

Starting the IV

The IV tube should be checked for air-bubbles, which must all be cleared before the line is connected. The needle is simply inserted into the catheter, and the tube is pulled into and "S" shape, without kinking it, and taped to the animal's leg. The flow rate should be adjusted according to the animal's weight.

Scrubbing the animal

The "mud-flaps", small, translucent drapes as seen in Figure IV-25, are first placed around the site to be scrubbed. Be particularly careful to protect the eyes. The prep table is positioned near the animal's head and the prep kit, which is sterile, is opened and prepared. A pair of sterile gloves is on top in the kit. The circulator must use the closed-glove procedure to glove themselves, and avoid contaminating both the gloves and the kit. The towel is moved to the side but remains on the sterile wrapper. The Betadine scrub solution is poured over the sponges that are in one half of the kit, and paint is poured over the sponges with handles in the other half of the kit (see Figure IV-26). The circulator begins with the sponges in the scrub solution. It is a 5 minute scrub procedure, and there are five or six sponges per kit, thus, each should be used for about one minute. Each sponge is used once and then discarded. One always starts in the middle of the area and progresses slowly, in increasingly larger circular motions, toward the edge of the region. Always move from the center outward, from scrubbed to unscrubbed, from clean to dirty, and never the opposite. When the edge is reach, the sponge is discarded and the next is taken. When five minutes has elapsed (and all the sponges are used), dry the area with the sterile towel.

The towel should be unfolded, placed on the region, and the region should be pressed or patted dry (do not rub the towel around). Then grab the towel by the two far corners, lift it cleanly off the head and discard it. Do not drag the towel across the head as this will only drag dirt onto the clean area.

The area is now painted (Figure IV-27) using the sponges with handles that were soaking in Betadine paint. This is not a timed procedure. Take one sponge after the other and repeat the center-outward circular scrubbing described above for the 5 minute scrub. Once all the paintbrushes are used scrubbing is finished and the animal is not touched again until the surgeon dries its head and marks the area.

Draping the animal

The surgeon will complete the preparation of the incision site, including the final drying, marking and draping. The surgeon takes a sterile towel from the scrub nurse and dries the head of the animal as described above.

The surgeon then takes a marking pen in one hand and wraps the forefinger of the other in gauze. Despite scrubbing the area, it is only clean and not sterile, and it is, therefore, a good idea to touch it as little as possible prior to opening. After the incision site is marked, the area is sprayed with Hollister spray by the circulator, and then the final drapes are applied.

- First are the utility drapes. These are prepared one by one by the scrub nurse (the adhesive strip is exposed) and handed to the surgeon such that they can be applied without additional manipulation. Without touching the skin of the animal, the drapes are positioned and pressed firmly against the skin.
- Next, the fanfold sheet is unfolded by either the scrub nurse and the circulator or the surgeon and scrub nurse and placed in position, covering the animal completely from just below the margin of the utility drapes.
- The final drape is sticky on the underside and goes directly over the incision site. The adhesive is exposed, the drape is centered over the incision site and pressed firmly down onto the skin. It is then unfolded to create the final layer of the sterile field. The incision site and sterile field are now ready. The scrub nurse positions the Mayo stands over the bed, and pulls the back table and ring stand into position. The surgery can now begin.

Following the draping of the animal (Figure IV-28), the cords of the monopolar and bipolar coagulators, as well as the suction tube, are laid down starting from the head of the animal (with enough slack for maneuvering) and extending to the feet. The end that is to be connected to a device is handed carefully to the circulator to be plugged in. The cords and tubes are then secured on the field (by the scrub person) by clamping the main drape around them using an Alice forceps. This forms a channel through which they freely run. Do not clamp the cords or tubes themselves. The final positions of all equipment in the OR, as well as the instrument Mayos, back table and ring stand are shown in Figure IV-29.

Monitoring During Anesthesia

The state of the animal is monitored continuously during anesthesia and the heart rate, temperature, and respiration rate are recorded every 15 minutes.

Recovery of the Animal in the OR

Removal from stereotaxic device: The animal's head is removed from the stereotaxic device, and the animal is placed on its side, facing the anesthesia machine, with towels supporting its head, in order to ensure the airway remain clear. The animal remains hooked-up to the anesthesia machine until the field is clear and everything is ready for the animal to wake up.

- Discontinuation of A- and IV-lines: The A-line is discontinued and the catheter is removed. The site is then appropriately dressed. The IV is discontinued, but the catheter and vein cap are left in place in case drugs need to be administered during recovery from anesthesia.
- Discontinuation of anesthesia: The anesthetic gas is turned off, and the animal is flushed several times with pure oxygen and allowed to breath pure oxygen for 5-10 minutes. The

bifurcated anesthesia tube is disconnected from the endotracheal tube, and the tape securing the endotracheal tube is removed.

- Topical antibiotic should be applied to surgical wounds. In the case of an eyecoil placement, ophthalmic antibiotic ointment (chloramphenicol ointment) is placed directly in the eye and the eyelid is closed. All epidermal incisions are treated with triple-antibiotic (Panalog). The animal is also given a dose of systemic antibiotic (Baytril). H_2O_2 should be used to clean up dried blood (be careful not to get any in the monkeys eyes, nose, mouth etc.).
- Unclip ECG.
- Continue to check pulse and respiration periodically. There should be a gradual increase in both.
- At the first sign of a recovered gag reflex, he endotracheal tube balloon is then deflated in preparation for tube removal. The tube is quickly but carefully removed from the trachea while the animal is inhaling. In so doing, the animal's first action with a free airway will be to breath out, thereby pushing any fluid outward and not drawing it into the lungs.
- When the monkey begins to regain consciousness and movement, the catheter may be removed. Remove the Bioclusive dressing and tape and carefully expose the catheter. The catheter should be entirely free from the animal before attempting to remove it from the vein. Have a gauze pad ready to apply pressure to the insertion point to stop bleeding.
- Remove the monkey from the surgical table and place the monkey on the floor (or some other location where he can not injure himself if he moves around).
- Make sure the monkey is kept warm. Cover him with towels or use a heat lamp.
- If the monkey has not received fluids during the procedure it is a good idea to hook up a 5% Dextrose in Lactated Ringers drip now if the monkeys seems as though he will be down for a while. If he has been receiving fluids, continue.
- Replace the collar on the animal. When the monkey can sit up without falling over, seat him in his chair for observation.
- An strong (opioid) analgesic may be given after surgical procedures. However, it is necessary to carefully control analgesia to avoid the animal touching and contaminating the wound areas. If a strong analgesic is results in the animal irritating the wound a weaker substitute, such as Tylenol or Paracetamol, may be given. These may be given P.O. with some juice. The monkey may receive fluids, but it is not a good idea to feed him until the following day. Analgesics are usually given for the next few days or for as long as the animal seems to be in pain.
- When the monkey is awake and alert enough to support its own weight, it is placed in its primate cage.

The monkey should be given antibiotics for the next 5-7 days. In this lab, Baytril is the most commonly used postsurgical antibiotic. Tribrissen, administered P.O. in some fruit, or cephalothin have also been used with regularity.

Finally, a heat lamp is positioned in front of the cage, and the animal remains seated in the cage for 24 hours to allow the healing process to begin undisturbed. The monkey is provided with free access to water and is put back on a regular feeding schedule after 24 hours. During the 24 hour period in the cage, the animal's condition is monitored closely. The following morning the animal is again given a dose of systemic antibiotic (Baytril or Cephazolin) as well as an oral analgesic (Tylenol or

Paracetamol) In the afternoon, the animal is returned to a clean cage in the vivarium. The administration of oral analgesic continues twice a day for another 24 to 48 hours, or as deemed necessary by the person responsible for monitoring the animal's recovery. Administration of systemic antibiotic continues twice a day for six days, making a total of treatment time of 10 days.

Treatment and Monitoring in the Recovery Room

Following a surgery to place a head-restraint post and eyecoil or a recording chamber is implanted, the operated animal is allowed to recover in special recovery cage for up to 72 hrs before being transferred back to his cage in the vivarium. There are two very important reasons for doing this. First, by keeping the animal's hands away from his wounds, we can guarantee that he will neither irritate his open wounds or newly operated eye, nor will he contaminate what is otherwise a relatively clean wound perimeter. This benefit can be clearly seen by comparing the condition of animals recovered in their chairs to animals recovered in a cage, both with respect to their eye and head. We have, in the past, had to place monkeys on systemic antibiotic treatments in order to fight eye infections which were caused, at least in part, by self-inflicted irritation of operated eyes. This, of course, is not healthy for the monkey, and delays the onset of our experimental protocols.

Second, by placing the animal in this special recovery cage, it is possible to keep him both clean and to administer appropriate levels of analgesic, including topical medication directly on the eye, without fear that he will injure himself. This allows us to make his recovery as painless as possible without jeopardizing his well-being. In the past, strong analgesics, which seem appropriate following such a major surgery, have often been avoided because on more than one occasion an operated monkey has removed the newly implanted eye coil in the course of rubbing or scratching his eye. We previously explored a number other procedures for aiding the recovery of the animals, none of which were found to be satisfactory.

We attempted to use the oversized collars which isolate the monkey's head from the rest of his body. These collars, however, are not well designed for primates, as they prevent normal self-feeding and because the monkeys are capable of removing them with little effort. In addition, we have considered the possibility of covering the hands of the monkeys with extensive soft bandages to prevent them from touching their wounds. Here again, however, we are sure that the primates will become consumed with the notion of removing the bandages, and resort to violently using their teeth and feet in order to free themselves of such temporary patches. There are, indeed, concerns about leaving an animal in a chair for an extended period of time. We believe we have taken all necessary precautions with regards to these risks.

Under no circumstances has an animal ever been left alone if he shows the slightest indications of discomfort beyond that which is expected following a major surgery. Furthermore, while the animal is in the recovery cage, he is fed regularly and provided with as much liquids as he will drink.

Finally, proper delivery of post-operative medication is administered, and is clearly indicated in the animal's health records. We firmly believe this course of action is the most appropriate method of recovery currently available to us. We are, however, in the process of designing a new recovery system which will provide some freedom of movement for the recovering animal but still prevent him from injuring himself.

We cannot stress enough that it would be easier for us to simply put the animal back into his cage and not bother with this particular procedure. However, our genuine concern for the animal's successful and timely recovery has led us to the conclusion that, unlike the case of human post-operative patients, we must take extra precautions to protect the recovering animal from his natural inclinations to put himself at unnecessarily high risk following surgery. It is often forgotten by those outside the laboratory that our research depends critically upon the good health and cooperation of the animals with which we work. It is our opinion that our standard handling, surgical, and post-operative procedures speak for themselves with respect to our personal feelings about animal care.

Clean Up and Instrument Cleaning Protocols

After the last stitch is in place, the Mayo stands, back table, and double-ring stand are backed away from the bed to allow easy access to the animal. The area is cleaned, antibiotic and the desired dressing are applied by the scrub person and held in place while the drape sheets are removed with assistance from the circulator, rolling the drape sheet inside out toward the foot of the table. The scrub person, with the equipment, should then step back to a position away from the operating table, where the actual termination procedure will take place. Prior to disposing of drape material, it should be checked for instruments, since they may be still attached to the drapes during the dismantling phase. Disposable linen and cloth items are then placed in their respective bags for disposal. Gloves must be worn during this process since blood and fluids may still be present on the drape sheet. All sharps, including cautery tips, blades, and hypodermic needles should be placed in a container (a needle-counter box can serve this purpose) and disposed of according to acceptable protocol.

Any items needed during recovery, for an emergency or otherwise, must be left in the OR in a place accessible to those responsible for monitoring the recovery. These instruments can be collected later. The remainder can be removed and taken to the room will the clean will be done. Gloves must be worn during cleaning as all items in the OR are considered contaminated.

Cleaning of the surgical instruments

The first phase of the decontamination process involves mechanical cleaning. The most common methods of mechanical cleaning are the ultrasonic washer, washer-sterilizer, and the new washer-decontaminator. Wash all surgical instruments (regardless if they were used or not). One way is to put them, in small batches, in the sonicator. Otherwise, do them by hand. Sometimes even when they are sonicated, a film or rust stains remain on them (a result of being in zephiran with the antirust tablets, or from autoclaving repeatedly). If this is the case, an abrasive sponge (the green scrub pads or surgeon's hand brush) must be used to clean them. Lay out to dry on paper towels or white cotton towels. Clean all equipment and instruments used in the OR. Wipe down the heating pad, the stereotaxic frame, and the eye, ear and mouth bars, beakers and pans, and the hand drill (this must be taken apart, cleaned with both water and oil and Q-tips, inside and out) with zephiran and water. The paralytic infusion tube and any other tubes used, if they are not disposable or will be used again, need to be cleaned thoroughly inside and out. The entire cutting head of the clippers should be disassembled, thoroughly cleaned, reassembled, and sprayed with Clippercide Spray (germicidal) while it is running. Wash all the laundry that was taken into the OR, regardless of whether it was used. Do all the white towels or (anything with blood on it) twice : once in cold water, the second in hot water with liquid bleach (to clean first, then to get the blood stains out and make the towels white again). Do the blood-stained towels separate from the clothes.

Terminal Cleaning

After the surgical procedure, all items that have come in contact with the animal or sterile field should be considered contaminated. Interim cleaning of the procedure room is performed at the end of each case, using an established protocol. If the wet-mop method is used, the mop head and disinfectant solution must be changed after each use and properly disposed of according to policy.

Adequate time must be allowed for proper disinfection and set-up of the procedure room. At the conclusion of the surgery, the procedure room, scrub/utility area, corridors, furnishings and equipment should be terminally cleaned.

Restock all surgical supplies, according to the list for general surgery. This lessens the chance of forgetting to order new supplies as well as finding just prior to the next operation that something is missing. If gown or instrument packs need to be made, it is best to assemble them directly after washing the laundry or cleaning the instruments. This way they are ready in plenty of time for the next procedure and are also available in case of an emergency. Immediate clean-up and repacking also insures that things do not lay around with the potential of being misplaced (and generally are not in the way). The packages of materials to be ethylene oxide sterilized can also be assembled and placed in storage such that they can simply be pulled off the shelf and sterilized for the next surgery.