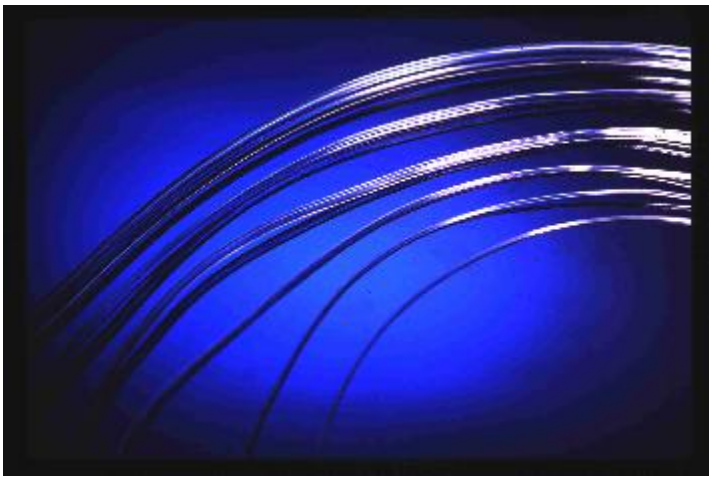
A Guide to Manufacture, Surgical Implantation, Maintenance and Use of Micro-Renathane® Vascular Catheters in Laboratory Animals



By Alfred Sherer, D.V.M. and Lewis B. Kinter, Ph.D.

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I. Introduction

Micro-Renathane® Tubing is precision-made polyurethane micro-tubing offered exclusively by Braintree Scientific, Braintree, MA 02185 (781) 348-0768 or Fax (781) 843-7932. Micro-Renathane® is a natural elastomer; no plasticizers are added to the formulation to preserve its flexibility. Plasticizers used with polyvinyl and polyethylene products are unavoidably 'leached' from the tubing, causing the tubing to become increasingly stiff and brittle with time. Worse, the plasticizers are known toxicants that can accumulate in experimental animals and confound interpretation of results. Micro-Renathane® Tubing is suitable for use in most animal studies. Micro-Renathane® has reduced thrombogenic potential, compared to other plastics and is available in a variety of sizes.

	MRE 010	.010 O.D. x .005 I.D.	50 ft roll only
	MRE 025	.025 O.D. x .012 I.D.	
	MRE 033	.033 O.D. x .014 I.D.	
	MRE 040	.040 O.D. x .025 I.D.	
	MRE 050	.050 O.D. x .040 I.D.	
	MRE 065	.065 O.D. x .030 I.D.	
	MRE 080	.080 O.D. x .040 I.D.	
	MRE 095	.095 O.D. x .066 I.D.	
	MRE 160	.160 O.D. x .091 I.D.	
	MRE COMBO	Make your own kit with your choice of above sizes (excluding MRE010 – minimum 36 ft.	

Micro-Renathane® Tubing has many uses in modern experimental surgery. The following is a compilation of information on the preparation, implantation, maintenance, and use of vascular catheters made from Micro-Renathane® Tubing.

Notice: Braintree Scientific is always willing to attempt to answer customer's inquiries about its products, and promises a 'good faith' effort to provide accurate and balanced information; however, Braintree assumes no responsibility for the ultimate accuracy of the information provided in this document and elsewhere. Braintree assumes that its products will not be used in human studies, that all surgical procedures will be conducted with approved protocols, by qualified, appropriately trained persons, with the

assistance/support of qualified veterinary staff, and in accord with all Federal, State, and Institutional regulations and guidelines.

Warning: Manufacture and implantation of vasculature catheters will expose individuals to some physical, chemical, and biological hazards. All procedures should only be carried out by qualified personnel, in approved work areas and in accord with all Federal, State, and Institutional Safety Procedures.

II. General Considerations for Using Micro-Renathane® Tubing for Vascular Catheters

Micro-Renathane® Tubing is more flexible than most polyethylene, polypropylene, and Teflon® tubings, but is less flexible than most silicon rubber-based tubings. Micro-Renathane® Tubing is acceptable for most catheterization techniques and often provides superior results, due to its improved physical, chemical, and hematological characteristics. One general precaution: polyurethane has a small but finite permeability to water. Hence, provided that a sufficient gradient for water transfer exists (e.g. hydrostatic or osmotic pressure gradients), water will move across the wall of the Micro-Renathane® Tubing, either out of or into the lumen, depending upon the direction of the net gradient. Operationally, the significance of this property of polyurethane tubings may be most apparent when the tip of a polyurethane catheter is located in an artery or otherwise exposed to elevated hydrostatic pressure. Unless precautions are taken, water may be lost from the catheter lumen and, with time, blood may enter the lumen from the arterial end of the catheter. To counter-act this property of polyurethane, catheters should be soaked in aqueous solutions for 24 hours prior to implantation and filled (when not being used) with hyper-osmotic solutions (see below) to counteract hydrostatic gradients. Also, the ends of the polyurethane catheters exposed to air can be covered with a variety of materials to reduce water permeability, including silicon rubber, plastic tapes, glues, nail polishes, and waxes.

III. Manufacture of Catheters from Micro-Renathane® Tubing

This Section describes the manufacture of basic arterial and venous catheters from Micro-Renathane® Tubing for use in rats and other small animals. We are unaware of any reason why these procedures cannot be scaled upwards for larger animals or downwards for smaller animals. There are many techniques for manufacture of catheters and no comparative data exists with which to compare the efficacy, longevity, and success of different techniques. Different investigators tend to be satisfied with what has worked for them – so you'll need to discover what works best for your application.

For intravenous or intra-arterial drug administration, central venous and arterial pressure monitoring, and/or intravenous or intra-arterial blood sampling, it is often preferred to insert catheters into the vena cava or abdominal aorta via the femoral vein or artery, respectively. The femoral vessels are easy to locate and isolate, are close to the surface, and are of large diameter (compared to the other peripheral vessels). In addition, the vessels are located in a position in the cardiovascular system such that small thrombi, if inadvertently introduced will be trapped in normally non-critical vascular beds (e.g. hind limbs). The following procedures are scaled for manufacturer and implantation of catheters for insertion via femoral vessels in Sprague-Dawley rats of 250g or larger.

Preparation:

The final length of the desired catheter, including the distance from the point of insertion in the femoral triangle to the intended location of the catheter tip in the aorta or vena cava, the distance from the femoral triangle to the point of exit (often at the nape of the neck), and the length to be left outside of the animal, must be measured and excess allowed for manufacture (20-40%). It is often desirable to measure these distances in several similar sized animals.

Materials:

- ✓ Venous Catheter: 0.025" i.d. x 0.040" o.d. Micro-Renathane® Tubing
- ✓ Arterial Catheter: 0.012" i.d. x 0.025" o.d. or 0.014" i.d. x 0.033" o.d. Micro-Renathane® Tubing
- ✓ #5 or #7 Watchmakers Forceps
- ✓ Fine Scissors and Scalpel
- ✓ Short lengths of polyethylene tubing (approximately 0.043" or 0.038" o.d., respectively)
- ✓ Kitchen matches, butane lighter, or micro-burner
- ✓ Small cutting board (preferably plastic)

Procedure:

Cut several pieces (length plus excess) of the appropriate diameter Micro-Renathane® Tubing. Cut a long (~1cm) temporary bevel on each end.

Count the number of places at which you intend to suture the catheter to fascia or muscle. Cut an equivalent number of 2-4mm pieces of polyethylene tubing (plus a few extra). Using forceps, gently grip each piece in the center and 'flair'. The flared ends can be small or large to suit different purposes (see below). Using the beveled ends of the catheter tubing, thread the appropriate number of polyethylene ('sleeves') and advance to their approximate positions. The polyethylene pieces should grip the Micro-Renathane® tubing firmly, but not occlude the lumen. The tines of fine forceps can be used to stretch

the bore of the polyethylene sleeves, if necessary. Polyethylene sleeves with small flares can be inserted into the lumen of large veins for suturing the vein around the catheter.

With the polyethylene sleeves in their approximate positions, the catheter may be trimmed to final dimensions. The temporary bevels used to thread the sleeves should be removed with the sharpest, cleanest blade possible. A 'rough' cut (w/dull scalpel) or 'crush' cut (w/scissors) will produce microscopic fractures and abrasions which will defeat the anti-thrombogenic properties of the polyurethane. Catheters should be cut on a soft surface. A small polyethylene kitchen cutting board does nicely and can be sterilized. Cut the end of the catheter to be introduced into the blood vessel as 'blunt' (perpendicular to the lumen) as possible to reduce the 'cut' surface area. If a bevel is to be cut, it is useful to mark the other end of the catheter to show the side to which the bevel has been cut.

Finished catheters should be sterilized before being implanted. Ethylene oxide gas or cold liquid sterilization should be used; heat should be avoided. Catheters can be treated with anti-coagulant polymers (e.g. TDMAC heparin) to increase antithrombogenic activity – follow the manufacturer's instructions.

Obturators:

If you intend to plug your catheters after implantation, you will need to prepare obturators (plugs). Traditionally lengths (~1cm) of stainless steel piano wire are cut and the tips polished using a fine grinding wheel. An alternate and much less expensive approach is to use pieces of the appropriate-sized monofilament fishing line. These are available in many diameters, are inexpensive and disposable, do not require polishing and do not 'weight down' the end of the catheter.

Connections:

It is often necessary to temporarily join the end of a catheter to another piece of tubing, a syringe, pressure transducer, or to lengthen the catheter as the animal grows. To temporarily join a catheter to another piece of tubing, segments of hypodermic tubing (thin-walled is best) of appropriate o.d., with both ends carefully polished to prevent punctures, may be used. Hypodermic tubing adapters are available in standard gauges from some manufacturers of hypodermic needles and can be used to join a catheter to a syringe. Alternatively, hypodermic needles may have their tips removed and deburred/polished using an inexpensive micro-lathe. An appropriately sized O-ring compression fitting (Touhy-Borst adapter) can also be used to couple catheter tubing to a syringe.

Alternatively, a connection may be made by joining the ends of two pieces of tubing using a short length of larger-bore material. This method does not constrict the bore of

the joint. A 1 cm piece of thick-walled (high-pressure) polyvinyl tubing with an i.d. of approximately the o.d. of the catheter will do. Short segments of thick-walled silicone rubber tubing may also be used.

For a very snug connection, or for lengthening a catheter in a growing animal, use a 1.5 to 2 cm length of silicone rubber tubing of smaller i.d. than the o.d. of your catheter. 'Relax' the silicon rubber tubing by soaking it in a small volume of xylene for a few seconds. When the bore of the silicone rubber piece is enlarged, wipe off the excess xylene and insert the catheter ends into opposite ends of its bore. Gently 'blow-off' the xylene, or let it evaporate and the silicone rubber will 'shrink fit' over the 2 ends of the tubing and form a tight seal connections.

Tapering Catheters:

Micro-Renathane® Tubing may be tapered using tension, once the tubing has been heated to an appropriate temperature. One technique is to carefully heat a small amount of sesame oil to approximately 200^0C using a stirring bar, an electric hot plate, and an appropriate thermometer. You'll need to adjust the temperature to between 180 and 210^0C to find the optimal temperature. Insert a small loop of Micro-Renathane® Tubing and observe it 'relax' in the hot oil. Remove the loop and gently pull from both ends. When the Micro-Renathane® Tubing is heated appropriately, it 'pulls' like candy or glass. Hold taught for a few seconds to allow the tapered section to cool. Pulling heated Micro-Renathane® Tubing sharply will produce a sharp severe taper; pulling more slowly will produce a more gradual taper. You'll need to experiment to produce the correct taper for your needs. Caution should be exercised when working with the hot oil!

Swaging needles onto catheters (Barger-Herd catheter; Herd and Barger, 1964):

Sharpened hypodermic needles or tubing can be attached (swaged) to Micro-Renathane® Tubing. First, select Micro-Renathane® Tubing and hypodermic needle (or tubing) of approximately the same o.d. Note that the hypodermic tubing should not be of larger o.d. than the Micro-Renathane® Tubing. Next, taper the ends of the Micro-Renathane® Tubing to which hypodermic needles (or tubing) are to be affixed (see above). The Micro-Renathane® Tubing should be tapered to a diameter that will slide easily through the bore of the hypodermic tubing. Cut sections of hypodermic needles or tubing no longer than necessary. If hypodermic needles are cut or otherwise removed from their hubs, carefully inspect both ends to be sure they are not occluded or burred. Polish if necessary. The tine of a fine forcep may be used to de-burr or polish the i.d. of the metal tubing.

Next, carefully thread the tapered Micro-Renathane® Tubing through the bore of the hypodermic tubing (back to front). This is tedious and may require magnifying glasses.

When the tapered Micro-Renathane® Tubing is threaded through the hypodermic tubing, grip and stretch the tapered Micro-Renathane® Tubing as much as possible, without breaking and slide the hypodermic tubing back along the taper until it snugs up firmly on the shoulder of the tubing. Quickly release the tension to affix the back of the hypodermic tubing to the shoulder of the taper. Stretch the end of the tapered tubing protruding from the front end of the hypodermic tubing. If a needle is used, be careful not to burr or blunt the needle tip. Use a pair of hemostats or opthamalic needle drivers to bend the swaged needle into a curve (the needle bevel towards the inside), if desired.

Storage and sterilization of catheters:

Store swaged catheters carefully to prevent damage to needle tips. Sterilize catheters using ethylene gas or cold sterilization fluids. Flush lumens and soak overnight in 0.9% saline to hydrate the plastic prior to surgery.

IV. Implantation Techniques

General:

Implantation of vascular catheters requires survival surgical techniques and should only be attempted by qualified individuals, with appropriate professional veterinary support, and with strict adherence with Institutional, State and Federal regulations and guidelines. An appropriate area in which to perform small animal survival surgery and post surgical recovery is necessary. All equipment to be used should be appropriately disinfected or sterilized, and the work area decontaminated prior to and following use.

Anesthesia:

In general, vascular catheterizations can be accomplished within approximately 30 minutes. A variety of anesthesia regimens are suitable; 3 representative regimens are outlined below. Professional veterinary staff may have additional recommendations and should be consulted prior to any attempt to anesthetize an animal. Remember, substantial variations in inter-individual-strain-species responses to anesthetic agents may occur. Be prepared to administer antidotes (if available) and to provide critical care support.

Isofluorane (inhalation) – Isofluorane is a volatile anesthetic agent and is administered by inhalation. Equipment with which to mix isofluorane with air/oxygen, and to trap expired gas is required. Rats are initially anesthetized in a sealed box flushed with isofluorane/O2 gas mixture. Anesthetized rats may be maintained by allowing them to breath isofluorane/O2 spontaneously from a gas cone. A condom may be used to make a near-airtight seal around the rat's head and gas cone. Isoflourane provides exquisite control and the most rapid and predictable recovery following completion of surgery.

Fentanyl/Isofluorane (intramuscular/inhalation) – Fentanyl is an excellent opiate analgesic but does not sufficiently relax muscle tone for surgical anesthesia. The

principal advantage of its use is that fentanyl may be rapidly antagonized at the end of surgery with naloxone. Administer fentanyl (o.4 mg/kg) intramuscularly and top off with isofluorane (by inhalation) to achieve an appropriate surgical preparation. Braintree Scientific Decapicones® can easily restrain rats for intramuscular injections. Isofluorane may be administered using inhalation apparatus (as above), or in a cone. For the latter, place isofluorane-soaked gauze in a 50 ml centrifuge tube and place the rat's nose in the open end of the tube and monitor for effect. Cap the cone tightly when not in use. In using isofluorane without inhalation apparatus, surgery should be conducted in a hood, or other arrangements made to collect and trap/exhaust isofluorane. When surgery is completed, administer naloxone (0.4 mg/kg) intramuscularly to reverse the fentanyl. Provide a warm clean environment for recovery from anesthesia, monitor/maintain body temperature at approximately 37^0C until animals are recovered from anesthesia.

Ketamine/Xylazine (intraperitoneally) – Ketamine/Xylazine is a combined phencyclidine/xylazine anesthetic regimen and may be admistered intraperitoneally. The dose for rats is approximately 55-65/5.0-7.5 mg/kg. The two drugs may be mixed and stored together. Administer ketamine/xylazine intraperitoneally. Braintree Scientific Decapicones® can be used to easily restrain rats for intraperitoneal injections. Simply place the rat in the cone (head first) and close the large open end about the tail. Palpate the rat's abdomen and inject through the plastic.

These regimens provide approximately 30 minutes of surgical anesthesia and may be supplemented, as needed. Short-acting (methohexital) and long-acting (pentobarbital) barbiturates are not recommended.

Checking the plane of surgical anesthesia:

Before and during surgery, the state of surgical anesthesia should be checked frequently by assessing toe pinch and ocular reflexes. See P.A. Flecknell's Laboratory Animal Anesthesia (Academic Press, 1987 – available from Braintree Scientific) for more detail.

V. Catheter Implantation Techniques

Once an appropriate and stable plain of surgical anesthesia has been induced, the rat is moved to the surgical area and the incision site shaved and disinfected. The surgical field should be draped. Provisions should be taken to monitor and to support the body temperature of the rat (procedure >30 min.) and/or to provide body temperature support during recovery from surgery. Braintree Scientific Deltaphase® Pads are an ideal solution for both purposes.

Equipment and materials:

- ✓ fine animal hair clippers and skin disinfectant (Betadine)
- ✓ ethanol
- ✓ sterile drape for rat
- ✓ scalpel, fine scissors, iris scissors or micro-scissors
- ✓ 000 or 0000 suture
- ✓ mosquito hemostats (2 pairs, straight and curved)
- ✓ #5 and #7 fine forceps
- ✓ trochar
- ✓ hypodermic needles (various sizes, 18-25)
- ✓ procaine or xylocaine
- ✓ wound clips or suture needles
- ✓ tissue adhesive

Procedures:

Once the femoral triangle and back are shaved, disinfected, and draped, and the anesthesia state re-checked, a sharp scalpel is used to make an incision (1-2 cm) through the skin over the femoral vessels, parallel to the external oblique. Hemostats or forceps are used to open the fascia and to expose the femoral sheath. The sheath is flushed with procaine or xylocaine solution. Using the curved (#7) forceps, with sharp deburred tips, carefully separate the artery, vein, and nerve sheath. Beware of collateral vessels entering the artery or vein from below! Place pairs of sutures (s9ze 000 or 0000) to retract the vessel selected to accept the catheter. Flush the area with procaine or xylocaine. Gently retract the vessel, first 'upstream' then 'downstream' to engorge the area into which the catheter will be introduced.

The catheter is connected to a syringe filled with heparinized saline (~1000 IU/ml) and the catheter lumen filled.

Techniques:

Several techniques may be used to introduce the catheter tip into the vessel lumen. Three are described below.

General Method – Mount a 22-25g 1cm hypodermic needle on a 1cc syringe filled with heparinized saline. Using hemostats, bend the needle tip (~0.5cm from the tip) to an angle of ~100 degrees (with respect to the axis of the syringe), keeping the needle bevel up (within the 100 degree angle). Flush the needle tip with heparinized saline. Insert the needle tip into the engorged isolated section of the retracted blood vessel. Alternatively, a pair of iris scissors or micro scissors may be used to cut a hole in the vessel – in this case, grasp the top of the vessel with fine forceps and lift

gently to produce a 'tent' and cut with scissors into the back of the 'tent' to the lumen of the vessel. Using the straight (#5) forceps, grasp the lip of the punctured vessel from the bevel of the needle. Withdraw the needle, while retaining grasp of the puncture site with the forceps. Insert the catheter tip (cut and trimmed as described above) into the puncture while lifting the upper lip of the puncture gently with the forceps tips. Relax the proximal retraction and advance the catheter tip into the vena cava or abdominal aorta.

Scissors-Action Forceps Method – This method uses a pair of scissors-action micro-forceps. Individual investigators may wish to hone or grind the tines of these forceps to reduce thickness and to sharpen the tips. The engorged section of the retracted blood vessel may be punctured with a hypodermic needle (see A above), or directly with the sharpened points of the scissors-action forceps. Once the tips of the forceps are inserted into the blood vessel lumen, the tines are gently separated and the catheter tip threaded between the separated tines directly into the vessel lumen. The forceps are then removed and the catheter tip advanced. Relax the proximal retraction and advance the catheter tip into the vena cava or abdominal aorta.

Barger-Hard Technique – This technique is for catheters with swaged needle tips (see above). Attach a syringe (filled with heparinized saline) to the catheter and fill the lumen with heparinized saline (if possible). Bend a gentle curve in the swaged needle, keeping the bevel of the needle towards to concave arc. The blood vessel into which the catheter is to be introduced may or may not be isolated or retracted. Holding the needle (a pair of ophthalmic needle drivers work nicely for this), puncture the blood vessel lumen and advance the needle tip along the lumen for at least 0.5 cm. Repuncture the blood vessel wall with the needle tip (inside to out) and pull the needle and at least 0.5 cm of catheter through the second puncture. Carefully and cleanly cut the catheter behind the needle (see above) and discard the needle. Gently retract the catheter to 'pop' the newly cut tip back through the second puncture into the lumen. Quickly advance the tip past the second puncture into the vena cava or abdominal aorta. Apply gentle pressure with a cotton swab to the second puncture until any bleeding stops. A small piece of surgical 'gel foam' may be used to stop the bleeding at the puncture site.

Securing the implanted catheter:

Once the catheter tip has been advanced to its final position in the vena cava or abdominal aorta and patency has been confirmed, the catheter should be secured with suture or medical-grade adhesive (cyanomethacrolate) at, or as close as possible to, the puncture site. The catheter may be 'glued' to the insertion site. Alternately, the catheter may be secured within the vessel using a 'basket weave' knot around the vessel. The sutures used to retract the vessel may be used for this purpose; care must be taken not to occlude the catheter lumen. Note: This method occludes blood flow around the catheter in the vessel. Finally, the catheter may be sutured to fascia or muscle near the insertion

site, using a polyethylene sleeve (see above). Particular caution must be taken to prevent arterial hydrostatic pressure from 'blowing' the arterial catheter out of the blood vessel. It is generally advisable to secure the catheter at another point approximately 1 cm from the insertion site to prevent direct pulling on the insertion site. Do not suture a 'loop' of catheter as this may kink and occlude the catheter.

Tunneling the Catheter:

The open end of the catheter (not implanted in the vasculature) must be tunneled sub-dermally to an exit point, usually at the nape of the neck. A puncture (14g needle) is made at the exit point. A sterilized trochar (14g) is inserted and maneuvered sub-dermally to the cannulation incision. The obturator of the trochar is removed and the catheter threaded up the bore of the trochar. The trochar is removed carefully from the exit point, threading the catheter along its path and out the exit point. When the distance between the cannulation site (e.g. the femoral region) and the exit site (e.g. the nape of the neck) is long, it is sometimes helpful to tunnel the catheter in two steps (femoral region to mid-back, mid-back to nape). In this case, it is often helpful to suture the catheter to the underlying muscle at the mid-back site before closing the skin incision. Sutures or wound clips should not be placed at the exit site as these promote irritation and will cause the rat to 'groom' this site and potentially damage the catheter. The site should be shaved and disinfected and precautions taken to promote rapid healing.

VI. Catheter Use

Rats should be observed closely or gently restrained during experimental procedures to minimize accidental catheter damage. Braintree Scientific Experimental Containment Units (ECUs) are an ideal device for use with catheterized rats. A Decapicone® can serve in an emergency situation. Once the rat is secured, the catheter is occluded. A pair of light hemostats or opthalamic needle driver (without teeth or serrations) is a useful tool to occlude catheters. The obturator is removed and blunt hypodermic needle (tubing adapter) attached to a syringe is inserted in the catheter bore. Alternatively, larger bore tubing attached to a syringe may be applied over the catheter end (see above). The content of the catheter is evacuated gently into the syringe. Fresh isotonic vehicle (saline or glucose, usually containing anticoagulant) is flushed through the catheter (2-4 catheter volumes). The catheter may then be connected to monitoring equipment, infusion pumps, or used for injections or blood sampling. It is helpful to keep anticoagulant in the bore of the catheter when blood is not being withdrawn or infused. At the end of use, reflush the catheter and refill with a Loc-solution (see Mann et al., 1987)/ seal with a sterile obturator.

VII. Catheter Maintenance

Catheters should be checked daily for accidental damage. Punctured sections of catheters should be removed or covered with a 'sleeve' of silicon rubber tubing (see above). In long-term studies, it may be necessary to lengthen the catheter as the rat grows; use the catheter bonding techniques described above.

When not in use, catheters should be filled with a Loc solution. Experience suggest that hyper-oncotic solutions of saline (9.0%) or glucose (25-50%) with anticoagulant (heparin, 1000 U/ml) work well. Hyper-osmotic solutions are bacteriostatic and create a large inwardly directed osmotic gradient favoring water entry from the tissues into the catheter. This osmotic gradient more than counteracts the low water permeability of thin-bore tubing, even under arterial pressures, and may create a slow auto-perfusion of the catheter tip.

VIII. Sources

Cold sterilization products – 'Amerse', Calgon, Corp., St. Louis, MO 63133; 'Omni-II', ProChem Co., Cottrell, Ltd., Englewood, CO 80112-9937; 'Gericide', Henry Schein, Inc., Port Washington, NY 11050.
Decapicones® - Braintree Scientific, Inc., Braintree, MA 02185-0929.
Fentanyl – Pitman-Moore, Mundelein, IL 60060.
Hypodermic tubings and Stainless Steel Wires – Braintree Scientific, Inc., Braintree, MA.
Hypodermic needles and tubing adapters – Becton Dickinson & Co., Rutherford, NJ 07070.
Isoflourane chamber and gas scavenging system – Braintree Scientific, Inc., Braintree, MA 02185-0929.
Medical grade adhesive (no. 891) – Dow Corning Corp. Medical Products, Midland, MI 48640.
Microsurgical supplies - Braintree Scientific, Inc., Braintree, MA 02185-0929
Naloxone – Elkins-Sinn, Inc., NJ 08003.
Polyethylene tubing - Braintree Scientific, Inc., Braintree, MA 02185-0929.
Polyvinyl tubing (S-54-HL, MicroBore) – Norton Co., Akron, OH.
Post-surgical antibiotic – 'Polyflex' (Sterile Ampicillin Suspension), Fort Dodge Laboratories, Inc., Fort Dodge, IO 50501.
Rat restrainers and ECU's - Braintree Scientific, Inc., Braintree, MA 02185-0929.
Micro-Renathane® - Braintree Scientific, Inc., Braintree, MA 02185-0929.
Silicon rubber tubing – Braintree Scientific, Inc., Braintree, MA 02185-0929.
Surgical Disinfectant – 'Prepodyne Scrub' WestAgro, Kansas City, MO 64153.
TDMAC-Heparin – Polysciences, Warrington, PA 18976.
VetBond – 3M Animal Care Products, St. Paul, MN 55144-1000.

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