The emissary veins of the bull's penis. G. H Ardalani, Urmia University, P.O. Box 165, Urmia, IRAN.

To understand the relationship of ccp of mature bulls with csp and csg and the action of erection, injection experiments were carried out on 6 excised organs from Friesian bulls aged *2%* to *5%* years with Microfil and latex. Injection shows that the ccp and csg have very fine connections that act as a safety valve during full erection. Microfil was slowly injected with a syringe into the ccp of specimen 6 at the level of the distal bend of the sigmoid flexure, without previous injection of the dorsal artery. The injection resulted in filling of the dorsal artery and also the dorsal venous system. In the cleared specimen, the csg was also found to be injected, with filling of left and right lateral components of dorsal arterial and dorsal venous systems. Sliced preparations from this specimen showed very clearly the intimate connections established between csp and csg at the apex of the organ. Is it possible that some bulls have fine veins draining the ccp distally? Veins draining the distal ccp that run a straight radial course through the tunica albuginea may limit the pressure that can be attained within the ccp. This has been discussed by Rotter and Schurman (1950) acting as "safety valve" mechanisms to prevent over-distension of the tunica albuginea, yet never becoming large enough to cause leakage that would interfere with erection? On general morphological grounds, it would be expected that such veins, connecting a very high pressure cavernous system with a low pressure venous system would behave in accordance with the three postulates of Thoma (Clark, 1971). These postulates are: 1) Increased rate of blood flow produces an increase in the size of the lumen of the blood vessel. 2) Increased tension in the wall of the blood vessel produces an increase in the thickness of its wall. 3) An increase in blood pressure provides a direct stimulus for the formation of new blood vessels.

Plastination. Harmon Bickley, Department of Pathology, School of Medicine, Mercer University, Macon, Georgia, 31207, USA.

Plastination is a process for impregnating a biological specimen or other porous material with a curable polymer, using the difference in vapor pressure between the infiltrating polymer and a volatile intermediate solvent to propel the impregnation. It is protected by paten;*, however the use of plastination to generate specimens for teaching or museum display may be permitted without royalty if a license to do so is obtained in advance from the holder of the patent. Plastination was invented by Dr. Gunther von Hagens at the University of Heidelberg in the early 1970's. The entire process includes four steps: 1) fixation, 2) dehydration, 3) forced impregnation 4) curing. Each of these will be introduced in this paper and discussed in more detail by speakers that follow. Silicone elastomer is the polymer most widely used in plastination. Epoxy and polyester are employed in special techniques. The qualities of specimens plastinated with each of these will be discussed. Many educational institutions throughout the world are now producing plastinated specimens. They are useful in a number of ways, their most common application being in undergraduate, medical, and veterinary medical education. Plastination promises to be a fruitful technique for research, however more development is needed. A plastination laboratory is not difficult to establish or operate and the equipment may be as simple or elaborate as one's budget permits.

Pitfalls and pleasures of plastinatina pathology specimens. Sally Ford, Kingston General Hospital, Kingston, Ontario, CANADA, K7L 2V7.

The rationale for production of pathology specimens for plastination is different from that in anatomy. In the latter, the objective is simple demonstration of normal anatomic structures and relationships. In pathology, anatomy is distorted by pathological processes, and disordered structure and function require novel approaches in dissection. This paper outlines the problems encountered in setting up a pathology museum of plastinated specimens, and highlights some of the rewards.

Starting plastination on a limited budget. Gilles G. Grondin, D6partement de Biologie, Faculty des Sciences, University de Sherbrooke, Sherbrooke, QUEBEC, CANADA, J1K 2R1.

After 3Jj years of efforts, a plastination laboratory was started in Sherbrooke with less than $3,000.00 (Canadian $). Those years of preplastination were used to collect different pieces of equipment for plastination (old vacuum pump, glass desiccator, tubings and connections, needle-valves from a

scrapped freeze-dryer) and to harass the teachers and the administrators to get money and a room. The laboratory that we obtained was 3.5 square meters (2.5m x 1.4m), covered by a glass ceiling to insulate from the lights and avoid the spark hazard. Bringing in ventilation and getting out wall plug, cost $360.00. Buying a household deep freezer and moving the thermostat and compressor from it, cost $870.00. A Bennert manometer, a hydrometer and a gas mask were bought for $700.00. Finally, 40 liters of acetone and 10 kg of Biodur S 10 Silicone resin cost $900.00, and we were ready to start. The dehydration jars were old 4 liter jugs with the top removed and covered by a glass cover. The plastination kettle was a glass desiccator and the curing chamber a plastic bag in an old aquarium. We were able to treat specimens of about the size of a hand but even with this restriction on the size of the specimens, we started plastination. After teachers had seen and touched the plastinated specimens and realized their value, they wanted more and bigger ones. It was then easier to get money to buy a bigger vacuum chamber and possibly move in a bigger room. We realized that starting plastination may be a long term adventure but it is possible to do it on a very limited budget and that perseverance and recycling may replace money. After it is started, you can easily demonstrate that it is worth while and that you can produce any required specimen. Then it is easier to improve your installation.

A simple and inexpensive method for recycling used acetone in plastination laboratories. Gilles G. Grondin and Serge B6rube^, Departements de Biologie et de Chimie^, Faculty des Sciences, University de Sherbrooke, Sherbrooke, QUEBEC, CANADA, J1K 2R1.

Used for the dehydration and defatting of specimens in the plastination process, acetone represents an important part of the operating budget of the laboratories. We have developed a method that is cheap, efficient, and safe for recycling used acetone. This method consists of 3 steps:

1. Freeze separation: This consists of just storing your used acetone in a freezer at -20°C. Fat, water and other contaminants that may be present will form solid particles big enough to be removed by filtration through cheese cloth.

2. Vacuum distillation: This step may seem complicated, but it is very simple. We just use the same equipment as for the impregnation procedure. The contaminated acetone is poured in a vacuum Erlenmeyer flask and warmed to 45 °C. This flask is connected to a second one that is inside the freezer at -20 °C. The second flask is connected to the vacuum pump and enough vacuum is applied to obtain boiling in the first flask. The acetone will boil from the first flask and condense in the second one.

3. Physical extraction of water: We add to the distilled acetone a desiccant (Molecular Sieve) that will complete the extraction of water from it.

We have used this technique for one year and consider it very efficient. The first step is optional and we use it mainly on the residues of the distillation. The second step is the most interesting. We can distill acetone at a rate of 500 to 600 ml/hour and reach up to 97% purity. The third step is also optional as we can use new acetone for the last dehydration bath. Finally, we needed to invest just $300.00 in the new equipment to achieve our objective, which was to completely recycle our acetone. We remove the cost of disposal of the used acetone, buy just a minimum of new solvent and contribute to the protection of the environment.

Plastination at Queen's: What worked and what did not. Blake Gubbins, Department of Pathology, Queen's University, Kingston, Ontario, CANADA K7L3N6.

When establishing a plastination laboratory, most of the obvious problems, that one might face, have been addressed in the literature or can be solved fairly easily. Our experiences uncovered some areas which deserve further investigation. They include: 1) Selection of a fritted-glass, gas-dispersion tube for the gas cure procedure. 2) How to clean the tube when it gets blocked with precipitate from the S 6. 3) The best place to put your vacuum pressure control valve. 4) Is an explosion-proof freezer really best for plastination? 5) Are we being sufficiently safety conscious in plastination? 6) Canadian Safety Standards as they relate to the latest developments in laboratory design.

The standard *S* 10 technique. Robert W. Henry, College of Veterinary Medicine, The University of Tennessee, Knoxville, TN 37901, USA.

The standard S 10 technique, although having been improved over the years, is the classical plastination technique. Specimens produced by this method are quite durable. Almost any type of specimen may be produced using this method,

including: whole organs, entire bodies or prosections of portions of the body, whole body slices or brain slices. The steps include: 1) SPECIMEN PREPARATION, 2) MINIMAL FIXATION of the tissue with subsequent removal of the fixative, 3) DEHYDRATION, 4) INTERMEDIARY SOLVENT, 5) IMPREGNATION, 6) CURING or HARDENING. 1) SPECIMEN PREPARATION: An extremely important step as unattractive or inappropriately prosected specimens will remain just as unattractive after plastination. Attractive, useful specimens take time and expertise to prepare. 2) FIXATION: Minimal formaldehyde fixation is the standard. Fixative solutions which are designed to preserve color may be used. The specimen should be fixed in the conformation of the desired final product. Once a specimen is fixed, it is difficult to change its fixed shape or configuration. However, long term fixed specimens may be plastinated. Once fixed, the fixative is rinsed from the specimen using running tap water. 3) DEHYDRATION: is another critical step. Cold acetone (-25 °C) or room temperature ethanol may be used. Cold acetone dehydration, called "Freeze Substitution", has become the accepted method. Dehydration must be complete to avoid shrinkage and provide complete impregnation of the specimen. 4) INTERMEDIARY SOLVENT: Is a volatile solvent whose vapor pressure is such that it can be extracted slowly as pressure is decreased allowing the polymer to be drawn into the tissue as it leaves. Acetone is an intermediary solvent, therefore, this step is accomplished as the specimen is dehydrated. Ethanol's vapor pressure is too low (similar to water) to serve as an intermediary solvent. Therefore, if ethanol was used as the dehydrant, it must be replaced with an intermediary solvent, methylene chloride or acetone. The specific gravity of ethanol and acetone are similar, therefore, the determination of % content of either solution via a hydrometer is not possible. 5) IMPREGNATION: Is the replacement of the intermediary solvent with the curable polymer mixture (Biodur S10/S3). Classically, impregnation is carried out inside a vacuum chamber which is located inside a deep freezer. The cold retards curing (linkage of the polymer molecules). Impregnation may be done at room temperature, but linkage of molecules is accelerated and the pot-life of the polymer reaction mixture is greatly reduced. Impregnation is carried out over a 2 to 4 week period, as absolute pressure is slowly decreased nearly one atmosphere to a final pressure of 1 cm Hg or less. Impregnation is monitored by observing the bubble formation, created as the intermediary solvent is volatilized by the decrease in absolute pressure (increasing vacuum). Impregnation is complete when the bubbling ceases or the absolute pressure is less than 1 cm of Hg. 6) CURING or HARDENING: Is carried out at room temperature after the excess polymer has been drained from the specimen. At room temperature and over a several month period, the S10/S3 mixture will slowly harden via end to end linkage of the polymer molecules. To speed up the curing process, the impregnated specimen is exposed to another hardening agent, Biodur S6, which commences side to side linkage of the polymer and hence seals the surface in 36 to 48 hours. After exposure to S6, the specimen cures from the outer surface inward over a period of several days to weeks.

Vacuum, vacuum gauges and manometers. Robert W. Henry and James R. Thompson, Jr.[1], College of Veterinary Medicine and [1] Department of Physics, The University of Tennessee, Knoxville, TN 37901, USA.

Vacuum is recorded from two perspectives: 1) A decrease in absolute pressure (AP) or 2) An increase in vacuum. These values can be recorded as a fraction of an atmosphere, e.g., 1/3 atmosphere (20 inches or 50.8 cm of Hg, AP), 1/2 atmosphere (15 inches or 38.1 cm Hg), or 2/3 atmosphere (10 inches or 25.4 cm Hg, AP). A column of Hg or vacuum gauge yields a progressively higher reading as absolute pressure is decreased. This is referred to as gauge pressure. However, a manometer yields a progressively lower number and is read as AP. This is because the manometer is reading the difference in two columns of Hg. The vacuum gauge or column of Hg is using atmospheric pressure as point zero, while the manometer is using total vacuum as point zero. Because most manometers used in plastination laboratories utilize two columns of Hg whose difference in height is 20 cm or less, only the last 1/3 or less of the change in the pressure in the vacuum chamber can be monitored. Hence, a vacuum gauge or Hg column is necessary to monitor changes in absolute pressure (vacuum) in the earlier stages (first two thirds) of impregnation. The saturated vapor pressure (similar to boiling point) of dichloromethane is greater than that of acetone, 32.5 mm Hg vs 14.8 mm at -25°C or 78.0 mm vs 35.9 mm at - 10°C. Hence methylene chloride will

vaporize at a higher AP and be extracted before acetone. Pressure is proportional to depth. This results in the pressure being greatest the bottom of the polymer than at the surface of the polymer. At - 25 °C, acetone will remain longer in a specimen or portion of which is submerged 15 to 20 cm below the surface of the polymer. The gauge is reading the surface pressure. If the AP is 2 cm of Hg, then the pressure at the lower level of polymer is the surface pressure (0.38 psi) added to the pressure generated by the 15 inches of polymer (0.51 psi) for a total of 0.89 psi. The force generated when absolute pressure is decreased one atmosphere is 15 pounds per square inch (6.45 cm^). At total vacuum, the one foot cube desiccator, used for plastination, has 2,160 pounds (Ibs) of force on each wall, while an 18 inch by 30 inch surface of a medium size vacuum chamber, has 8,000 Ibs of force, and a 20 x 50 inch surface of a chamber has 15,000 Ibs.

Cadaver preservation methods employed in India: is plastination a viable alternative? Sylvia Kamath. Department of Anatomy, St. John's Medical College, Bangalore, 560034, INDIA.

Cadavers, require to be preserved for various purposes such as study, display, transport, and delaying the last rites. This is achieved by embalming. The body tissues are permeated with an embalming fluid which preserves, hardens and produces asepsis. The main chemicals employed in embalming fluid are formalin, carbolic acid and glycerin. Fluid is run into arteries under pressure, using various techniques. We use a simple machine designed and fabricated in our institution. Embalming techniques are modified under special circumstances i.e., autopsied bodies, post surgery bodies, infected bodies for example AIDS, infective hepatitis, rabies, and in unnatural deaths. Here local injection or local injection combined with evisceration and packing may have to be employed. In the Department of Anatomy at St. John's Medical College, we have devised certain techniques for effective museum display of thin sections (150 /zm) and thick sections (3 mm). Whole organs or parts may serially sectioned and displayed and provide useful models for the understanding and interpretation of CAT scans and MRI pictures.

Freeze-drving and plastination: Useful tools for keeping the bugs out. Wayne Lyons, Department of Anatomy, Queen's University, Kingston, Ontario, CANADA, K7L 3N6.

Over the years, despite the use of standard protocols and procedures, fixation has posed a problem in the preparation of anatomical and anthropological materials. Use of dangerous chemicals, shrinkage and durability of specimens have all plagued previous techniques. Only recently, with the advent of new methods such as freeze-drying and plastination, has the use of harmful chemicals such as formaldehyde and phenol been limited. The ability to preserve fresh, frozen specimens, as well as, pre-treated specimens has not resulted in the production of specimens of inferior quality. Specimens produced in this manner have not, to date, undergone deterioration due to the invasion of "bugs" (microbes or museum pests). Freeze-drying and plastination can be used separately or in combination to help solve additional problems in the preparation of specimens. When using the S 10 method of plastination, shrinkage of whole-brain specimens has caused a great concern among plastinators, however, by combining this method with the freeze-drying technique, this is overcome. The results have produced a superior quality specimen which can be used for teaching. The examination of anthropological materials (bones, fossils) has often proven difficult due to their fragile nature. Using the S 10 technique of plastination, on a fragile North American Indian skull, we have shown that Biodur plastic has functioned as a consolident to eliminate this problem. These approaches (i.e., freeze-drying and plastination) appear to have eliminated the decomposition and destruction of specimens. These techniques have definitely made great strides in debugging the tasks of preserving and maintaining specimens. It is only with continued research and investigation that this will be substantiated.

The use of plastinated transverse body slices for teaching CT anatomy. P. P. C. Nel, Department of Anatomy and Cell Morphology, University of the OFS, Bloemfontein, SOUTH AFRICA.

With the advent of computerized tomography scans (CAT-scans), gross anatomy courses should expose medical students to cross-sectional anatomy. The more recent introduction and wide spread use of MR as a non-invasive, non-ionizing method of acquiring high resolution, multiplanar images of the human body, is making the study of the cross sectional anatomy of the normal human body even more relevant. Clinical department are expecting

their students to have a knowledge of cross-sectional anatomy. Commercial sources for teaching specimens (e.g., transverse body slices) are nonexistent and academic sources available are prohibitively expensive. In order to by-pass the financial burden of purchasing teaching specimens, the department of Anatomy and Cell Morphology at the University of the OFS have, in conjunction with the department of Diagnostic Radiology, at the same university, embarked on a project to determine what the affect (if any) of plastination is on a cross section through a human body. The aim of the study is to determine whether the department could produce its own plastinated body slices for teaching material. A cadaver was CAT-scanned and then embalmed. The location of each scan was carefully marked on the body with a felt-tipped pen. After embalming, the body was sliced into transverse sections. These sections were cut exactly on the markings of the CAT-scans. All sections were plastinated. A comparison between the original CAT-scans and the plastinated body slices show no significant change in the normal anatomy of the specimen. Therefore, the process of plastination, as we apply it in our department, has no effect on the gross anatomy of transverse body slices and plastinated body slices are an excellent answer to the problem of how to teach transverse anatomy on cadaver material.

Plastination and the Mac. Robert H. Parmelee, Department of Anatomy, College of Veterinary Medicine, University of California, Davis, 95616, USA.

The cost and legal difficulties of obtaining animals for classroom dissection exercises, coupled with gradually increasing reluctance by students to participate in a high turnover program in animal use, clearly indicates a need for change in what has been standard operational procedures. At the School of Veterinary Medicine at U.C. Davis, we are using plastination as a preservative technique coupled with interactive computer programs that join text, photography, and plastinated specimens and offer a radically new approach to teaching anatomy. This combination has far exceeded our expectations in both student acceptance and as partial substitute for dissected specimens in anatomy teaching labs. The relative ease in developing these "user friendly" programs with specific plastinated materials makes this system desirable both from an economic standpoint and an instructional one. High quality Kodachromes are taken of the plastinated specimens and loaded into the Macintosh II Si using a slide scanner and Adobe Photoshop software. These images are sized and imported into the Mac Supercard program, which allows several windows to be opened on screen at once so that graphics and instruction can be presented simultaneously. This program allows graphics to be scripted so that students can interact with the program which then becomes more of a tool instead of just a display. A discussion of the problems encountered in the production of S10 plastinated specimens and the development of interactive Supercard programs will be presented.

Monitoring the degree of fixation, dehydration, forced impregnation and cure. M. Ripani, A. Bassi, L. Perracchio, V. Panebianco, M. Perez, M.L. Boccia, State University of Rome "La Sapienza", II Department of Human Anatomy, Roma, ITALY.

The possible technical mistakes in plastination have been examined in eleven parenchymal organs (liver, kidney and spleen) treated with S 10 Standard Technique. All of the four phases were considered: fixation, dehydration, forced impregnation and cure. Possible deformation of the sample was not considered because we routinely fix the specimens on stiff supports. We identified inappropriate dehydration and/or forced impregnation procedures as the most important reason of shrinkage and accidental variations of properties. On the other hand, in our experience during the other phases, damage to the specimens are scanty and nearly always reversible. If standard procedures, found in the literature, are thoroughly followed. Two parameters of dehydration and forced impregnation have been studied: degree and duration. Average values for the eleven organs were calculated and plotted in a diagram. These data make it possible to define the most appropriate treatment in order to reduce the lasting of the procedure, the amount of reagents used and to avoid technical artifacts.

Comparative analysis of a plastination specimen and clinical diagnostic images. M. Ripani, A. Bassi, M. L. Boccia, G. Tomaselli, and G. Marinozzi, State University of Rome "La Sapienza", II Department of Human Anatomy, Roma, ITALY.

This specimen is an invasive apocrine carcinoma of the perineal glands involving the anal canal, the vagina, and the vulva. It was taken from a 62 year-old subject. The patient was studied by Computerized Axial Tomography (CAT) before surgery. The specimen was washed in running tap

water and then in formaldehyde solution. It was dipped into acetone at increasing concentrations and then placed in a -20 °C freezer. Some weeks later, the specimen was dipped into Biodur S 10 plus S 3 and the placed in a vacuum chamber. When forced impregnation was completed, the specimen was exposed to fast cure and then to slow cure to be cleaned. Finally, slices of the specimen were cut following CAT images. By comparing CAT images and photographs of anatomical dissections, it was possible to confirm the accuracy of the *in vivo* instrumental study. Moreover, it was possible to explain in detail, structures which were ambiguous on CAT images. Thus, plastination has proven to be an important means for verifying sophisticated diagnostic images.

Sheet plastination. E 12 technique. Wolfgang Weber, Department of Veterinary Anatomy, Iowa State University, Ames, Iowa 50011, USA.

Sheet plastinated specimens find the highest acceptance of preserved body slices. Cross-sections of the body never were as detailed, never were transparent, never were projectable, never as durable nor as easy to handle. There are two techniques for sheet plastination. Brain sheets are done with a polyester-compound called BIODUR P 35. While, cross sections of the body are done with an epoxy compound called BIODUR E 12. The procedures are similar, but for the sake of clarity, each process will be presented individually.

LIST OF NECESSARY EQUIPMENT: A Vacuum chamber large enough to hold the flat chambers; Ultra-low deep freezer and an additional freezer; Manometer; Biodur Polymers: E 12, E1, AE 10 and AE 30; Meat slicer and/or Band saw with guide stop (refrigerated is preferred); 50*C Oven, Stainless steel (HD 18) or aluminum grids; Polymer mesh (HD 16) or Fly screen; Covered basket (HD 08); Appropriate containers for storage and dehydration of specimens (HD 07). To assemble each flat chamber: 2 Tempered glass plates (5mm) (HS 01 or HS 02), 6mm Gasket (HS 06), 15 large (HS 30) Fold back clamps or 2 inch binder clamps, and Plastic sleeve or hose (to serve as a funnel).

FIXATION OF SPECIMEN: Kaiserling's solution to enhance color of muscle. Fresh specimens with no fixation. Float specimen.

FREEZING THE WHOLE BODY AND PORTIONING: Suspend to minimize flattened areas. Remove and store extremities.

SLICING THE BODY PARTS: Even 2.5 mm slices for best transparency. Band saw with a refrigerated guide stop. Place slices in freezer, scratch saw dust.

ORGANIZATION OF SLICING: Place slice on grid covered with fly screen, in cold acetone (Fly screen-specimen sandwich). Cover with flyscreen and another slice, etc.

DEHYDRATION: Freeze substitution of stacked flyscreen/specimens.

DECREASING: Important for transparent slices. Room temperature acetone or methylene chloride.

IMPREGNATION: Polymer Mixture - E 12 (95 pbw), AE 30 (5 pbw), AE 10 (20 pbw), and E1 (26 pbw). Can not prolong its short life, after mixed. Weight stack of slices to submerge in reaction mixture.

CASTING: 2 tempered glass plates, 6 mm gasket, 5 mm spacer strips. Large fold back clamps.

FILLING THE MOLD: Plastic sleeve (funnel); 600 cc of E 12 (95 pbw), AE 30 (5 pbw), E 1 (26 pbw) mixture per mold.

EVACUATION: in vacuum chamber for 1 hour.

CORRECTING THE POSITION OF SLICES AND REMOVING AIR BUBBLES: Use pieces of stainless steel wire to manipulate air bubbles and the slices.

CLOSING THE MOLDS: The ends of the gasket are used to close the top.

CURING: Molds placed at a 15° angle for 1 to 2 days. 40 °C oven 1 week.

DISMANTLING: Cool to room temperature. Cover with clear plastic wrap.

CUTTING OUT: Mark 1 cm from perimeter of slice and saw on band saw.

SMOOTHING EDGES: Belt sander.

POSTERS

Understanding standard imaging planes for two-dimensional, real-time echocardiographv in the dog aided by plastinated specimens. Janice M. Bright and Robert W. Henry, Department of Animal Science, College of Veterinary Medicine, University of Tennessee, Knoxville, TN 37901 USA.

A valuable method for evaluating structure and function of the heart is ultrasonography. Two-dimensional, real-time echocardiography (2DE) routinely is used to provide images in a single plane. Standard imaging planes have been described for evaluation of the canine heart. Standard imaging planes were obtained from single frame images of real-time, 2DE examination of healthy dogs. To illustrate three-dimensional anatomy relative to the two-dimensional sector, these images are correlated with plastinated specimens which were sectioned along corresponding imaging planes. Labelled photographs of the sectioned specimens are displayed adjacent to the echocardiographic images. Standard imaging planes are designated and transducer positions, used to obtain standard imaging planes, are described. To enhance the student's ability to interpret standard imaging planes, a cadaver

heart was prepared for plastination. The heart was separated from the lungs by transecting the pulmonary vessels as long as possible. An appropriate size of tubing was used to cannulate a pulmonary vein and the caudal vena cava. The heart was flushed with warm tap water through both cannulas to remove blood. The other large vessels were closed by ligating a bleached cork in place (larger end into vessel). The remaining smaller vessels were closed with hemostatic clips, string, or suture material. The heart was hydrostatically dilated with cold water, dilated-fixed with a 5% Kaiserling's solution, dehydrated in cold (-25 °C) acetone, and impregnated with Biodur using the standard S10-S3 technique. Following impregnation, the heart was drained of the excess polymer-mix and exposed to Biodur S6 (gas curing agent). During curing, the heart was manicured daily to remove excess polymer-mix which accumulated on the surface of the specimen. After curing the heart was sectioned along the planes of standard images and attached to the poster.

The P 35 technique for sheet plastination of the human head. R. T. de Boer-van Huizen, C.J. Cornelissen and H. J. ten Donkelaar, Department of Anatomy and Embryology, Faculty of Medical Sciences, University of Nijmegen, The NETHERLANDS.

Modern imaging techniques such as computed tomography (CT) and magnetic resonance imaging (MRI) require a basic knowledge of cross-sectional anatomy of the human head or central nervous system (CNS). Although excellent volumes on sectional anatomy of the human head and CNS are available as anatomical guides for the analysis of CT and MRI sections, the study of real sections of the human head would be preferable, particularly for teaching purposes. Macrosections of the human head or brain have some disadvantages in that they have to be kept in formalin, fall apart because of handling and the differentiation between white and grey matter of the brain becomes less distinct in time, due to the bleaching effect of the fixative. Plastination, a technique of tissue preservation, introduced by von Hagens offers the possibility to prepare clean, dry and easy-to-handle slices. In the process of plastination, water and lipids are replaced by polymers which are subsequently hardened resulting in dry, odorless and durable specimens. The P 35 plastination technique gives excellent differentiation between grey and white matter of the brain and offers excellent reference material for pre- and postdoctoral training in cross-sectional anatomy. In the present study, the P 35 technique was applied to four mm anatomical slices of a formalin-fixed human head.

A new positioning technique for comparing sectional anatomy of the shoulder with sectional diagnostic modalities: magnetic resonance imaging (MRI). computed tomography (CT) and ultrasound (US). C. A. C. Entius, J. W. KuiperTT w. KoopsJ A. DeGastJ Departments of Anatomy and ^Radiology, ERASMUS University Rotterdam, The NETHERLANDS.

A new positioning technique is presented, based upon MR and CT imaging with surface markers applied to the skin of the specimen. With this technique, the required anatomic section for cryosectioning can be defined accurately. Plastinated slices, of 2 mm thickness, with the correlating MR, CT, and US images of double-oblique coronal imaging of the shoulder are shown. The method is equally applicable to all parts of the body and makes comparison of structures in correlative anatomic studies easier and more accurate.

Special brain preparations to show fiber tracts and vascularization. M. R. Haffajee, G. Mathura, and S. Kassim, University of Durban-Westville, Durban 4000, SOUTH AFRICA.

Although sectioned specimens of brain have reached a high degree of sophistication in plastination for museums, we have found that undergraduates still have difficulty conceptualizing three-dimensional concepts of fiber tracts. We have combined two methods to remedy this difficulty. Fresh brains are injected with a coloured mass to show the arterial tree. The ideal method for showing fiber tracts is by careful dissection following the "freeze-fixation technique". It also enables students to visualize the basal nuclei in three-dimension. Normally, these specimens are kept in formalin but are not amenable to regular use. By plastination using the "S 10" method, although some shrinkage occurs, they are suitable adjuncts to standard sections of the brain for learning.

Plastination of three-dimensional brain sections as a component of a learning module. G. Mathura, M. R. Haffajee, S. Kassim, University of Durban-Westville, Durban 4000, SOUTH AFRICA.

Neuroanatomy is considered to be one of the more difficult areas of study in anatomy.

A special limb plastination using the S 10 technique with roentgenological and angiographic views. G. Mathura, M. R. Haffajee, S. Kassim, University of Durban-Westville, Durban 4000, SOUTH AFRICA.

Using a combination of techniques, a limb preparation method is demonstrated that shows vascularization, and horizontal and longitudinal sections together.