Short Communication

Bleeding Simulation in Embalmed Cadavers: Bridging the Gap between Simulation and Live Surgery

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Summary

In veterinary medicine, surgical education and training require the development of abilities that can be acquired in practical classes using currently available models such as cadaver training. Limited availability of cadavers, undesirable changes in tissue texture and the absence of bleeding are the main disadvantages of cadaver-based training compared to training in live animals. This study proposes a chemical cadaver preservation method aimed at overcoming the aforementioned limitations. Blood circulation could be reproduced in preserved cadavers, thereby enabling satisfactory simulation-based training of several surgical procedures, from incision to suture and including hemostatic techniques. The model in this study introduces a high-fidelity simulation training alternative to prepare students for the practice of surgery. In this manner, surgical interventions would be restricted to surgical cases and healthy animals would not be submitted to surgical procedures exclusively for learning purposes.

Keywords: surgical education, 3Rs, surgical training, alternative model, chemically preserved cadavers

1 Introduction

Cognitive, visual and manual abilities required for the practice of surgery can be developed and trained using different approaches. While theoretical concepts are an integral part of the learning process, repeated practice of surgical procedures is vital for the acquisition and improvement of surgical dexterity among undergraduates and surgery professionals.

Live healthy animals are still used for surgical training in some veterinary schools despite wide availability of alternative methods. Examples of alternative training methods include inanimate models (Auer, 1994; Olsen et al., 1996; Griffon et al., 2000), virtual reality training (Larsen et al., 2012), videos (Smeak et al., 1994; Jukes and Chiuia, 2003), organ models (Szinicz et al., 1994; Sroka et al., 2012), and cadaver-based training (Carpenter et al., 1991; Silva et al., 2004, 2007; Mathews et al., 2010; Carey et al., 2013). Limited availability, differences in tissue texture and consistency and the lack of bleeding are often seen as limitations of surgical training in cadavers (Blaschko et al., 2007; Reed et al., 2009; Mitchell et al., 2012).

Simulation-based surgical training methods should be able to convey a sense of reality. Ideally, models with similar anatomical characteristics, but capable of replicating individual variations, organoleptic properties and the bleeding ability of live animals should be employed. At the same time, models should be feasible, ethically acceptable and obtained in a humane manner. Simulation-based training is intended to replace live surgery, thereby reducing the number of animals employed for learning purposes while still enabling the acquisition of the level of proficiency required for the practice of surgery, as well as the refinement of surgical skills. However, adequate models for replacement of live animals in surgical training are scarce.

Hemostasis is an important part of surgical training and can be a challenge to the novice surgeon. This project proposes a method of preservation of ethically sourced animal cadavers aimed at close reproduction of live tissue texture and bleeding. The purpose is to offer a high-fidelity simulation method for surgical skills acquisition and improvement.

2 Preparation of the cadaver model

Cadavers

This project was approved by the Ethics Committee for Animal Experimentation of the School of Veterinary Medicine and Animal Science, University of São Paulo – FMVZ-USP

(protocol number 2070/2010). Twenty-three cadavers (3-20 kg) of dogs dead of or euthanized due to non-recoverable and non-infectious causes at the FMVZ-USP Veterinary Hospital were used in this study. Informed owner consent for use in research and education was obtained in all cases. Euthanized animals were heparinized (Hepamax-S 5000 IU – Blausigel®; 5 ml/animal) prior to general anesthesia to minimize blood clot formation. Cadavers were placed in plastic bags and stored in a cold chamber at -8°C.

Cadaver preparation
Cadavers were thawed in water at room temperature for 24 h and washed with neutral detergent prior to use. A rectal enema with tap water was performed to empty the colon. Cadavers were then placed in dorsal recumbence and organoleptic characteristics documented for future evaluation of the preservation method. A skin incision was performed on the right side of the neck. Following subcutaneous tissue dissection and separation of the sternocephalic and sternohyoid muscles, the plexus containing the carotid artery, the internal jugular vein and the vagal sympathetic trunk was identified and vessels were loosely ligated to facilitate future location and manipulation.

The carotid artery was cannulated either with intravenous or urethral catheters and the vascular system infused with warm saline solution (37°C; 10% of body weight). Modified Larssen solution (Silva et al., 2004, 2007) was then injected at a dose corresponding to 5% of body weight or until clean fluid was recovered through a venotomy incision (jugular vein). Finally, the same solution (10% of body weight) was infused through the carotid artery following closure of the venotomy incision.

In cases where only limited volumes of blood could be recovered, further infusion was performed through the femoral arteries and the fluid drained either through the corresponding veins or the carotid arteries and jugular veins. Both sets of vessels can be used bilaterally for efficient lavage and drainage; this procedure is intended to remove blood clots that might interfere with circulation. Leaking of preservation solution into the abdominal cavity occurred throughout the infusion procedure.

Simulation procedures performed in this study involved intra-abdominal organs. Therefore, the peritoneal fluid was drained through two small incisions on the lateral aspect of the abdominal cavity. Consecutive lavage with saline and modified Larssen solution was then performed (300 and 100 ml for cadavers of less than 5 kg, 500 and 300 ml for cadavers weighing between 5 and 10 kg and 500 ml for cadavers above 10 kg, respectively).

Three different artificial blood circuits were created, as follows:
– Cannulation of carotid arteries and jugular veins for procedures in the head
– Cannulation of femoral arteries and veins for procedures in the trunk, pelvic limbs and abdominal cavity;
– Cannulation of the descending thoracic aorta and right atrium for procedures in the trunk, pelvic limbs and abdominal cavity in small-sized cadavers (less than 5 kg).
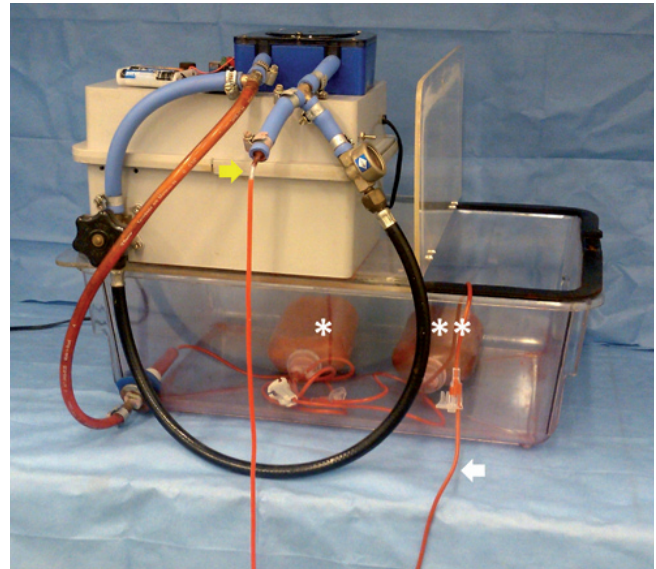

Fig.1: Custom-made electric pulsatile device
Bright red fluid (*) is pumped from the reservoir through an inlet, recovered through an outlet (yellow arrow) and then introduced into the arterial system. A fluid line containing dark red fluid (**) is connected to the venous system (white arrow).

Cannulations were performed as previously described (intravenous or urethral catheters) and catheters held in place with nylon ligatures. Prepared specimens were cleansed and stored in a cold chamber as previously described.

Preparation of blood surrogate solution
Commercial blood mimicking solutions could not be obtained; therefore bright red and dark red surrogate solutions intended to mimic arterial and venous blood, respectively, were prepared using water and water-soluble dyes (Corante líquido Xadrez, Sherwin Williams, Brazil); red and blue shades were mixed to obtain a darker solution as required. Surrogate solutions were stored in standard fluid bags. The fluid obtained is less viscous than blood.

Connection of cannulated vessels to the pumping system
Prepared specimens were thawed and cleansed prior to simulation procedures. Pre-cannulated vessels were then connected to surrogate blood bags and fluid infused into the target area by gravity flow until leaking through a purposely performed mini-incision was obtained (i.e., capillary flow).

Bright red and dark red surrogate solutions were infused through the arterial and venous system, respectively. Cannulated arteries were connected to a pump to replicate the arterial pulsatile flow. Cannulated veins were connected to the dark red surrogate fluid bag only. The pump employed in this study consisted of a custom-made electric pulsatile device. In this system, bright red fluid is pumped from the reservoir through an inlet and recovered through an outlet, with manually controlled pulsing pressure ranging from 0 to 280 mm Hg (Fig. 1).

3 Results

Cadaver preservation
Specimens remained free from unpleasant odor or signs of decomposition throughout the study; the skin remained intact in all cases. Changes in internal organ color and texture were observed in 3 out of 23 (13%) cadavers, albeit with no negative impact on surgical training.

Considering each step of preparation and use in simulations, all specimens were frozen and submitted to at least three thawing cycles, one (first) for cadaver preparation and the subsequent ones for simulation of surgical procedures. Depending on size, primary cause of death, state of preservation and previous surgical interventions (e.g., previous ovariohysterectomy would obviously preclude training of this technique), the same cadaver could be used for several procedures.

Surgical procedures
Destruction of local blood vessels precluded repeated surgical interventions at the same site; therefore, specimens were allocated to different surgical procedures by area.

The following procedures were performed in the head: enucleation, excision of eyelid masses, blepharoplasty, dental extractions, mucosal flaps, lateral ear canal resection and excision of cutaneous neoplastic masses. Tissue color and elasticity were preserved. Artificial bleeding requiring hemostasis occurred in all simulated procedures (Fig. 2).

Reconstructive surgical techniques were trained in the trunk area. Changes in tissue color and texture were observed at this site. Inconsistent bleeding patterns between specimens were also noted, with less than expected bleeding in some cases.

Knee arthroplasty, surgical approaches to long pelvic limb bones and femoral osteosynthesis were trained in pelvic limbs. Joint flexibility was maintained in all cases, along with satisfactory preservation of tissue texture and bleeding.

Simulated intra-abdominal procedures included gastrotomy, enterotomy, intestinal resection and anastomosis, nefrectomy, splenectomy, cistotomy and ovariohysterectomy. Tissues retained life-like texture and color. Artificial bleeding allowed proper identification and isolation of bleeding vessels, along with adequate training of preventive and control hemostatic techniques (Fig. 3). Surrogate blood leaking into the abdominal cavity was suctioned as necessary using a surgical aspirator to enable continuation of intra-abdominal procedures.

Urethrotomy, urethrostomy and orchiectomy (Fig. 4) could also be successfully simulated. Satisfactory artificial bleeding was achieved during surgical incision and manipulation with good preservation of original tissue texture and color.
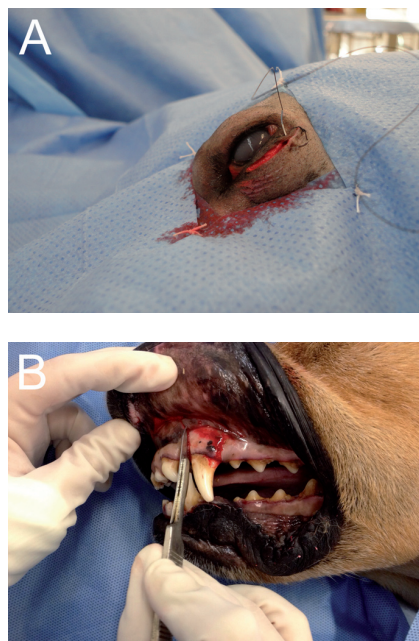

Fig. 2: Artificial bleeding during simulated procedures performed in the head
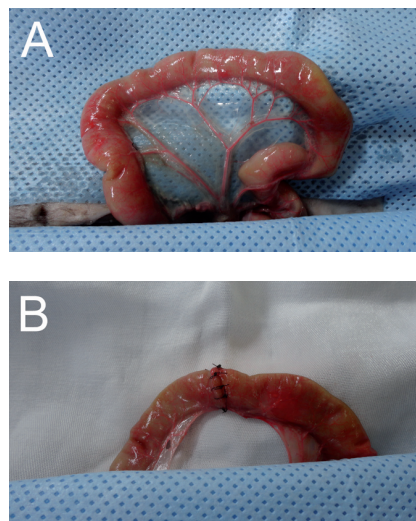(A) Blepharoplasty. (B) Mucosal flap.

Fig. 3: Artificial bleeding during simulated intra-abdominal procedures
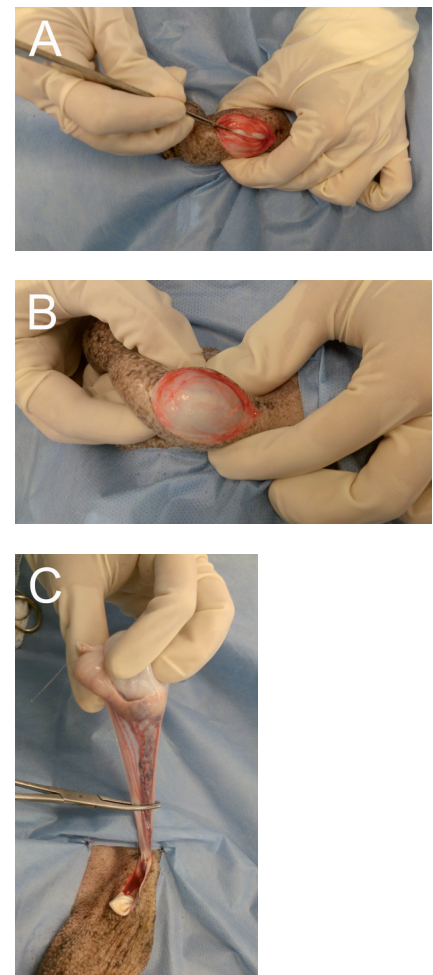(A) Enteric vasculature. (B) Enteric suture.

Fig. 4: Artificial bleeding during simulated orchiectomy
(A) Incision of the vaginal tunic. (B) Testicle excision. (C) Hemostatic clamp placed on the spermatic cord.

4 Discussion

Based on the results of this study, surgical and hemostatic techniques can be practiced in chemically preserved cadavers. In veterinary medicine, such procedures used to be trained independently using cadavers and models, respectively (Carpenter et al., 1991; Bauer, 1993; Olsen et al., 1996; Griffon et al., 2000; Silva et al., 2004, 2007; Mathews et al., 2010). Combined training of the three fundamental surgical principles in a single model yielded satisfactory results; a sense of reality resembling that of an operating room could be conveyed. Cadaver perfusion systems have been previously employed in humans (Garret, 2001; Aboud et al., 2004, 2011).

This study combined a perfusion technique and a well-established cadaver preservation method (Silva et al., 2004, 2007). Tissue texture was maintained and cadavers could be used several times, thereby overcoming some of the limitations of formaldehyde preserved cadavers (e.g., unpleasant odor) and of fresh/cryopreserved specimens (e.g., single use). Overall reduction of the number of specimens required for training and less time-consuming cadaver preparation are further advantages of the combined method.

Cadavers of animals dead or euthanized due to incurable diseases at HOVET-FMVZ/USP were employed; therefore, our sample was not homogeneous. Blood clots within the arterial and venous vascular systems may prevent proper perfusion of the preservation solution and compromise the quality of the model due to lack of proper bleeding simulation. Hence, cadavers in this study were heparinized whenever possible (euthanized animals).

Comparison of outer and inner organoleptic characteristics before and after embalming and following each simulation procedure is vital for critical evaluation of cadaver preservation methods. The preservation method studied proved satisfactory for extra-abdominal procedures. Several factors may have accounted for undesirable changes documented in intra-abdominal organs in 3 out of 23 cadavers studied. Time to preservation and cause of death (Silva et al., 2007), as well as body condition (i.e., muscle mass and obesity) and individual variations may impact the quality of cadavers preserved by identical methods (Jaung et al., 2011).

Chemical preservation of intra-abdominal organs is a difficult and time-consuming procedure. Proper preservation of tissue texture in contaminated hollow viscus remains a challenge to date. Intestinal cleansing, drainage of free abdominal fluid and the consecutive infusion of saline and preservation solutions was thought to alleviate the compression of blood vessels by intra-abdominal contents, facilitating tissue perfusion with the preservation solution. Changes in tissue texture, color and elasticity, as well as shedding and foul odor, tend to render the simulation experience unpleasant for trainees. Therefore, good tissue preservation is a vital aspect of cadaver preparation methods.

Surgical techniques involving different body systems and anatomical regions could be satisfactorily executed in the cadavers studied. Artificial bleeding allowed adequate training of hemostatic techniques required in procedures such as ovariohysterectomy and splenectomy, and closely reproduced bleeding-related difficulties inherent in routine practice of open invasive surgery. Adequate artificial bleeding was obtained in bone tissue, mucosae and serous membranes of abdominal organs, with poorer perfusion following epidermal and dermal incision. Dermal/epidermal perfusion tended to increase as vessels in the deeper cutaneous plexus were cut. Vessels in these plexuses are thought to be primarily responsible for intraoperative bleeding (Pavletic, 1991). Small vessel collapse and/or obstruction following death, the effects of cryopreservation and individual variations (Jaung et al., 2011) may have accounted for differences in tissue perfusion in this study.

Suction of surrogate blood accumulated in the abdominal and pelvic cavities was required in some cases. The undesirable, excessive leakage of fluid into the abdominal cavity observed at the time of connection of the specimen to the pump, during surrogate blood infusion by gravity flow and during the fixation process in some cases was thought to result from lack of vascular permeability control post mortem. Similar occurrences were not observed in previous studies, although excessive diffuse artificial bleeding leading to an increased level of difficulty during simulated training of selected techniques has been reported (Aboud et al., 2004, 2011). More viscous blood mimicking fluids should be tested and may help minimize this effect.

Undergraduates trained in cadavers and artificial models that do not mimic live tissue texture and bleeding tend to be less confident in their skills due to limited exposure to the actual challenges inherent to the practice of surgery and may therefore hesitate to operate on live animals (White et al., 1992; Smeak et al., 1994). Artificial bleeding introduces a high-fidelity simulation training method to overcome the lack of familiarity of novices with basic surgical tasks.

Technical errors during simulation are obviously devoid of clinical significance and morbid or lethal consequences. However, the practical experience gained with this method is a feasible alternative for replacement of live surgery in the beginning of the learning curve, and can certainly be applied in the operating room.

Allowing the refinement of surgical competence and proficiency before the first experience on real patients, surgical interventions can be restricted to animals that would really benefit from such procedures and avoid using animals exclusively for learning purposes.

References

Aboud, E., Suarez, C. E., Al-Mefty, O. and Yasargyl, M. G. (2004). New alternative to animal models for surgical training. *Altern Lab Anim 32*, 501-507.

Aboud, E. T., Krisht, A. F., O'Keeffe, T. et al. (2011). Novel simulation for training trauma surgeons. *J Trauma 71*, 1484-1490. http://dx.doi.org/10.1097/TA.0b013e3182396337

Auer, J. A. (1994). Veterinary surgery education performed on models. *ALTEX 11*, 44-46. http://www.altex.ch/All-issues/Issue.50.html?iid=144&aid=8

Bauer, M. S. (1993). A survey of the use of live animals, cadav-

ers, inanimate models, and computers in teaching veterinary surgery. *J Am Vet Med Assoc 203*, 1047-1051.

Blaschko, S. D., Brooks, H. M., Dhuy, S. M. et al. (2007). Coordinated multiple cadaver use for minimally invasive surgical training. *JSLS 11*, 403-407. http://scolaris.beta. semantico.com/media//data/0a/b2/_extracted/JSLS_2007_ Oct-Dec_11(4)_403-407/jsls-11-4-403.pdf

Carey, J. N., Rommer, E., Sheckter, C. et al. (2013). Simulation of plastic surgery and microvascular procedures using perfusedfresh human cadavers. *J Plast Reconstr Aesthet Surg 67*, 42-48. http://dx.doi.org/10.1016/j.bjps.2013.09.026

Carpenter, L. G., Piermattei, D. L., Salman, M. D. et al. (1991). A comparison of surgical training with live anesthetized dogs and cadavers. *Vet Surg 20*, 373-378. http://dx.doi.org/10.1111/ j.1532-950X.1991.tb00342.x

Garret, E. H. (2001). A human cadaveric circulation model. *J Vasc Surg 33*, 1128-1130. http://dx.doi.org/10.1067/ mva.2001.114214

Griffon, D. J., Cronin, P., Kirby, B. and Cottrell, D. F. (2000). Evaluation of a hemostasis model for teaching ovariohysterectomy in Veterinary Surgery. *Vet Surg 29*, 309-316. http:// dx.doi.org/10.1053/jvet.2000.7541

Jaung, R., Cook, P. and Blyth, P. (2011). A comparison of embalming fluids for use in surgical workshops. *Clin Anat 24*, 155-161. http://dx.doi.org/10.1002/ca.21118

Jukes, N. and Chiuia, M. (2nd ed.) (2003). *From Guinea Pig to Computer Mouse*. Leicester, UK: InterNICHE

Larsen, C. R., Oestergaard, J., Ottesen, B. S. and Soerensen, J. L. (2012). The efficacy of virtual reality simulation training in laparoscopy: A systematic review of randomized trials. *Acta Obstet Gynecol Scand 91*, 1015-1028. http://dx.doi. org/10.1111/j.1600-0412.2012.01482.x

Mathews, K. G., Riley, K., Lascelles, B. D. X. and Dernell, W. S. (2010). Preparation of canine and feline cadavers for surgical laboratories. *Vet Surg 39*, 224-225. http://dx.doi. org/10.1111/j.1532-950X.2009.00586.x

Mitchell, E. L., Sevdalis, N., Arora, S. et al. (2012). A fresh cadaver laboratory to conceptualize troublesome anatomic relationships in vascular surgery. *J Vasc Surg 55*, 1187-1194. http://dx.doi.org/10.1016/j.jvs.2011.09.098

Olsen, D., Bauer, M. S., Seim, H. B. and Salman, M. D. (1996). Evaluation of a hemostasis model for teaching basic surgical skills. *Vet Surg 25*, 49-58. http://dx.doi.org/10.1111/j.1532-950X.1996.tb01376.x

Pavletic, M. M. (1991). Anatomy and circulation of the canine skin. *Microsurgery 12*, 103-112. http://dx.doi.org/10.1002/ micr.1920120210

Reed, A. B., Crafton, C., Giglia, J. S. and Hutto, J. D. (2009). Back to basics: Use of fresh cadavers in vascular surgery training. *Surgery 146*, 757-762. http://dx.doi.org/10.1016/j. surg.2009.06.048

Silva, R. M. G., Matera, J. M. and Ribeiro, A. A. C. M. (2004). Preservation of cadavers for surgical technique training. *Vet Surg 33*, 606-608. http://onlinelibrary.wiley.com/doi/10.1111/ j.1532-950x.2004.04083.x/abstract

Silva, R. M. G., Matera, J. M. and Ribeiro, A. A. C. M. (2007). New alternative methods to teach surgical techniques for veterinary medicine students despite the absence of living animals. Is that an academic paradox? *Anat Histol Embryol 36*, 220-224. http://dx.doi.org/10.1111/j.1439-0264.2007.00759.x

Smeak, D. D., Hill, L. N., Beck, M. L. et al. (1994). Evaluation of an autotutorial-simulator program for instruction of hollow organ Closure. *Vet Surg 23*, 519-528. http://dx.doi. org/10.1111/j.1532-950X.1994.tb00513.x

Sroka, R., Weick, K., Steckmaier, S. et al. (2012). The ox-foot-model for investigating endoluminal thermal treatment modalities of varicosis vein diseases. *ALTEX 4*, 403-410. http:// dx.doi.org/10.14573/altex.2012.4.403

Szinicz, G., Beller, S., Zerz, A. and Bodner, W. (1994). The pulsatile organ perfusion – a chance to reduce animal experiments in minimally invasive surgery training. *ALTEX 11*, 40-43. http://www.altex.ch/All-issues/Issue.50.html?iid=144&aid=7

White, K. K., Wheaton, L. G. and Greene, S. A. (1992). Curriculum change related to live animal use: A four-year surgical curriculum. *J Vet Med Educ 19*, 6-10.

Conflict of interest statement

We declare that no author has potential conflicts of interest.

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