



Three-Dimensional Imaging and Plastination in Veterinary Anatomy Education

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ABSTRACT

Veterinary anatomy plays a crucial role in veterinary education. However, cadaveric dissection encounters significant challenges such as high costs, ethical concerns, and limited accessibility, particularly in developing countries. The present study aimed to develop supplementary, cost-effective educational tools using smartphone-based three-dimensional (3D) imaging and a modified Elnady plastination technique applied to ox and camel organs to enhance gross anatomy teaching. Digital photogrammetry was employed to create 3D models of the axis, skull, femur, lumbar vertebrae, and soft-tissue specimens of the head, including the nasal and oral cavities of an adult bull, utilizing a smartphone camera. Subsequently, the 3D models were processed and refined using the KIRI Engine 3D Scanner application. Bull specimens were obtained from the Anatomy Laboratory at the College of Veterinary Medicine, King Faisal University, Al-Ahsa, Saudi Arabia, from the existing collection. Meanwhile, anatomical specimens from five adult male camels (5 hearts, 10 kidneys, 5 penises, and 5 testes) and six adult bulls (6 testes) were plastinated using a modified version of the Elnady protocol to create long-lasting teaching tools. The 3D imaging produced precise, high-resolution, and interactive models of the ox skeleton, which clearly exhibited anatomical structures and spatial relationships with adjacent bones. The ox skeletal and soft-tissue 3D models demonstrated excellent detail, allowing clear visualization of anatomical structures. Plastinated specimens of the adult male camel preserved their anatomical integrity, exhibited minimal shrinkage, had adequate structural clarity, remained dry, odorless, and suitable for repeated handling. Among the ox specimens, shrinkage was observed in the testicular parenchyma following plastination. Using 3D imaging alongside plastination enhanced visual and tactile learning. The current findings indicated that smartphone-based 3D photogrammetry and the modified Elnady plastination technique offered a practical, scalable approach to modernizing anatomy teaching, especially in limited-resource settings.

Keywords: Anatomy, Education, Ox, Plastination, Veterinary, 3D imaging

INTRODUCTION

Veterinary anatomy is essential in veterinary education, forming the fundamental knowledge needed for clinical and diagnostic practices (Gummery et al., 2024). Anatomical knowledge and practical skills provide students with a strong academic foundation essential for advanced studies and clinical practice, including radiology, pathology, and surgery (Older, 2004; Sugand et al., 2010). Anatomy is widely considered one of the most challenging subjects due to the complexity of anatomical structures and the vast volume of information that students are required to master, despite its significant role in veterinary education (Dickson et al., 2022; Inpanbutr et al., 2024). Traditional teaching methods are often uninspiring, which can reduce student motivation and hinder knowledge retention (Warren and Donnon, 2013; Armitage-Chan et al., 2018). Cadaver-based instruction poses challenges, including health risks from formalin exposure, high costs, and ethical concerns (Ye et al., 2020).

In recent years, the global effort to reduce animal use in educational settings, coupled with financial constraints, infrastructural limitations, and reduced instructional time, has substantially affected veterinary anatomy education (King, 2004; Dickson et al., 2022). Collectively, these factors may result in deficiencies in anatomical knowledge and diminished student confidence. Consequently, there is a growing necessity to revise and modernize veterinary anatomy education, particularly in developing countries and other low- and middle-income countries such as Sudan, where resource constraints require cost-effective and sustainable teaching strategies (Sugand et al., 2010; Ozkadif and Eken, 2012; Chang Chan et al., 2024).

Advances in educational technologies have reshaped anatomy teaching, with growing emphasis on methods that reduce reliance on cadavers while enhancing learning outcomes (Guevar, 2020). Although cadaver dissection is valuable, educators are increasingly integrating innovative tools such as artificial intelligence (AI; Martín-Alguacil et al., 2025),

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digital imaging (Alcobaça et al., 2025), virtual platforms, three-dimensional (3D) visualization and modeling, 3D printing (Choudhary and Sarkar, 2025; Doğan and Kaçmaz, 2025), and plastinated specimens (Bernal et al., 2022). These teaching methods promoted active learning, enhanced understanding of complex anatomical connections, and addressed challenges in veterinary anatomy education related to specimen availability, safety, and ethics (Choudhary, 2026).

Imaging techniques, which broadly refer to methods that generate visual representations of internal body structures, play a crucial role in modern anatomy education (Hussain et al., 2022). However, students frequently encounter difficulties when transforming 2D images into 3D anatomical models (Preece et al., 2013; Wainman et al., 2018). The incorporation of 3D models into anatomical education effectively enhances spatial cognition and promotes understanding of complex anatomical relationships, especially for students with limited spatial visualization skills (Höffler, 2010; Schirone et al., 2024).

Plastination is now widely used in veterinary education as either an alternative or supplementary resource to traditional cadaveric specimens (Riederer, 2014). Plastinated specimens are widely accepted and have demonstrated educational benefits, particularly at the College of Veterinary Medicine, University of Murcia, Spain (Latorre et al., 2007). The plastination technique comprises four main stages, including fixation, dehydration, forced impregnation, and curing (von Hagens et al., 1987). Throughout these procedures, water and lipids within tissues are replaced with polymers such as silicone, polyester, or epoxy (Henry et al., 2019). The plastinated specimens provide sufficient contrast and structural clarity for educational purposes (Bernal et al., 2022). The Elnady technique is a recent advancement in plastination, offering a simpler, more cost-effective method for producing long-lasting specimens with minimal shrinkage and superior morphological preservation (Elnady et al., 2015; Elnady, 2016; 2019). Despite the growing adoption of low-cost 3D imaging and simplified plastination methods in anatomy education, their application remains limited within veterinary anatomy, especially in resource-limited settings. Few studies have integrated 3D imaging and simplified plastination approaches as supplementary educational tools. Therefore, the present study aimed to develop smartphone-based 3D imaging to generate 3D models of ox skeletal specimens, and a modified Elnady plastination technique to produce plastinated camel and ox specimens as low-cost educational resources for veterinary gross anatomy.

MATERIALS AND METHODS

Ethical approval

All experimental procedures were carried out in strict accordance with institutional and national regulations concerning the ethical use of animals in research. The present procedures received approval from the Animal Ethics Committee of King Faisal University (Ref. No. KFU-2026-ETHICS4248), Saudi Arabia. Biological samples were sourced from animals processed for commercial slaughter at an authorized abattoir in Al-Ahsa, Saudi Arabia.

Sampling

The present study was conducted in the dissecting room of the Department of Anatomy at the College of Veterinary Medicine, King Faisal University, Al-Ahsa, Saudi Arabia, over 16 weeks. Four skeletal specimens from an adult ox, consisting of the axis, skull, femur, and a lumbar vertebra, were selected from the Department of Anatomy teaching collection at King Faisal University, Saudi Arabia, for 3D imaging. The sample size was consistent with previous specimen-based plastination studies using a limited number of animal specimens (Medina et al., 2025). An expert veterinary anatomist with a PhD in Veterinary Anatomy selected these specimens based on their completeness, structural integrity, and the absence of visible damage or deformity, thereby ensuring precise 3D reconstruction. Camel bones were not included because the study focused on demonstrating the feasibility of photogrammetry rather than on species comparison.

Soft-tissue specimens were collected from animals slaughtered at a local abattoir in Al-Ahsa, Eastern Province, Saudi Arabia. Samples included one heart, two kidneys, one penis, and one testis from each of five healthy adult male dromedary camels (*Camelus dromedarius*), totaling 25 camel organ specimens. Additionally, one testis was collected from each of six healthy adult bulls, resulting in six bull testicular specimens. Bull testes were included to demonstrate the preservation technique in another domestic species, not for interspecies comparison. Soft tissues intended for the plastination process were obtained from 11 animals, comprising five camels and six bulls, all healthy and free of gross pathological lesions.

Preparation of 3D anatomical images

Digital photogrammetry was employed to produce original 3D anatomical models of gross specimens. Each specimen was documented utilizing a smartphone, equipped with a 48-megapixel Fusion main camera (Apple Inc.,

Cupertino, CA, USA). Images were systematically captured under consistent diffuse lighting conditions as the specimen was rotated incrementally at approximately 10-15° intervals until a comprehensive 360° image set was acquired. This process yielded approximately 60 to 130 overlapping photographs per specimen, depending on the specimen's size and complexity.

The photographs were processed using the KIRI Engine 3D Scanner (Version V4.2.8, KIRI Innovations, Hong Kong, China), with high-quality reconstruction settings, automatic image alignment, and quad-mesh generation with Physically Based Rendering (PBR) materials. The resulting models were subsequently refined in Blender (Version 5.3.0, Blender Foundation, Amsterdam, The Netherlands) by removing background artifacts, smoothing the mesh as needed, and optimizing the geometry while preserving the original anatomical morphology. Anatomical labels were subsequently added using Sketchfab (Epic Games Inc., New York, NY, USA) and the Emb3D Viewer application (Theia Interactive, USA) to produce interactive educational 3D models. The finalized 3D models were incorporated into gross anatomy lectures and practical laboratory sessions following previously described digital anatomy teaching workflows (Silva *et al.*, 2026).

Preparation of plastinated specimens

The main steps of the modified Elnady plastination technique used in the present study are summarized in Table 1. After collection of the specimens from the slaughterhouse, the soft-tissue specimens (heart, kidney, penis, and testes) were immediately washed under running tap water for 10-15 minutes to remove residual blood, dirt, and tissue debris before fixation (Bancroft and Gamble, 2008). The specimens were then blotted dry with absorbent paper and air-dried at room temperature (approximately 25°C) for 15-30 minutes to remove surface moisture before fixation, following a modified protocol adapted from Bancroft and Gamble (2008).

Table 1. Modified Elnady technique steps for preparation of adult ox and camel plastinated specimens

Stage	Description	Procedure	Duration	Conditions	Notes
1	Collection and washing	Specimens collected post-slaughter and washed under running tap water	10-15 minutes	RT	Removal of blood and debris
2	Initial drying	Air-dried on absorbent paper	15-30 minutes	RT	Surface moisture removal
3	Fixation	Immersion in 10% buffered formalin ($\geq 10\times$ specimen volume)	2-3 weeks	RT, sealed containers	Formalin was changed twice; specimens were kept submerged using a cotton cloth
4	Post-fixation drying	Air-dried on absorbent paper	Overnight	RT	Preparation for dehydration
5	Dehydration (80% acetone)	Immersion in 80% acetone (at least $10\times$ specimen volume)	1-2 weeks	RT	Acetone was tested every 2-3 days
6	Dehydration (90% acetone)	Immersion in 90% acetone	1-2 weeks	RT	Concentration maintained
7	Dehydration (100% acetone)	Immersion in 100% acetone	2-3 weeks	RT	Completed when water content was $<1\%$
8	Post-dehydration drying	Air-dried on absorbent paper	Overnight	RT	Before impregnation
9	Impregnation	Immersion in glycerin at a 5:1 glycerin-to-specimen volume ratio in a sealed container	5-8 weeks	RT	Duration depends on specimen size
10	Draining	Specimens drained	3-14 days	RT	Excess glycerin removal
11	Curing (cornstarch method)	Stored in cotton sacks inside buckets covered with cornstarch (no direct contact)	1-3 weeks	RT	Until glycerin exudation stops
12	Final cleaning and storage	Brushing/air compressor cleaning	—	RT	Stored at room temperature

The reference is adopted from Bancroft and Gamble (2008) and Elnady (2016). RT: Room temperature

Fixation

The specimens were immersed in 10% neutral buffered formalin in sealed plastic containers, with a fixative volume at least 10 times the specimen volume to ensure adequate fixation (Bancroft and Gamble, 2008). Cotton cloths kept the specimens fully submerged. Fixation time was adjusted based on specimen size, with smaller specimens fixed for 2 weeks and larger specimens for 3 weeks to ensure adequate penetration of fixative into deeper tissues. The formalin

solution was replaced on days 7 and 14 to maintain fixation quality. After fixation, the specimens were removed from formalin and air-dried overnight on absorbent paper at room temperature before dehydration ([Bancroft and Gamble, 2008](#)).

Dehydration

Dehydration was performed at room temperature (25°C) following a protocol adapted from [Elnady \(2016\)](#), with modifications to acetone concentration and exposure duration based on specimen type. Gradual acetone exchanges were employed to remove water and lipids, with specimens sequentially immersed in baths of 80%, 90%, and 100% acetone (Honeywell, Charlotte, NC, USA). Smaller specimens remained at each concentration for one week, whereas larger specimens required two weeks per step. The final dehydration step in 100% acetone lasted 2-3 weeks, depending on the specimen size, using about ten times the specimen volume of acetone. The purity of acetone was monitored every 2-3 days using a dual-scale hydrometer (DURAC® 50325 Specific Gravity/Baumé, SP Bel-Art, Wayne, NJ, USA), with the acetone solution replaced whenever its concentration fell below the target level. Dehydration was considered complete once the acetone concentration exceeded 99%, indicating that residual water was below 1%, as confirmed by hydrometer measurements. After dehydration, the specimens were extracted from the acetone and air-dried overnight at room temperature (25°C).

Impregnation

Dehydrated specimens were immersed in glycerin (Sigma-Aldrich, St. Louis, MO, USA) in sealed containers at a volume ratio of approximately 5:1 (glycerin to specimen). The impregnation period ranged from 5 to 8 weeks, depending on specimen size and tissue density, following the modified Elnady plastination technique ([Elnady, 2016](#)). Glycerin impregnation was considered complete when the specimens exhibited uniform pliability throughout the tissue, and no further visible changes in tissue consistency were observed during routine inspection. Vacuum impregnation was not performed because the modified Elnady plastination technique relied on prolonged passive glycerin infiltration at atmospheric pressure instead of forced polymer impregnation under vacuum ([Elnady et al., 2016](#)).

Curing

After impregnation, specimens were removed and allowed to drain at room temperature (25°C) for 3-14 days. Excess glycerin was gently removed using absorbent paper. Specimens were then placed in cotton bags and suspended within sealed plastic containers filled with cornstarch powder (Jeddah, Saudi Arabia), ensuring no direct contact between the specimens and the powder. Following the revised Elnady plastination protocol, cornstarch was used as a desiccant to absorb excess glycerin migrating from the specimens, as documented by [Elnady \(2016\)](#). Subsequent to the curing process, any remaining cornstarch particles were thoroughly eliminated using soft brushes prior to storage at ambient temperature (25°C). The plastinated specimens have preserved their integrity and have been used for teaching without any visible deterioration for over 12 months after preparation.

Data analysis

The 3D models and plastinated specimens were descriptively evaluated for anatomical accuracy, structural integrity, and preservation quality. The current findings were recorded through photographs, so no statistical analysis was conducted.

RESULTS

Smartphone-based 3D imaging

The smartphone-based 3D imaging technique yielded precise, detailed 3D models of bone specimens from an adult ox (Figures 1 and 2). The 3D model of an axial vertebra of the ox accurately depicted the vertebral body, dorsal spinous process, ventral crest, cranial and caudal articular processes, transverse processes, vertebral foramen, and alar foramina. Surface morphology and bony landmarks were clearly visualized from multiple viewing angles, enabling a detailed examination of the anatomical features (Figures 1A and 1B). The color-enhanced 3D skull models from an adult ox enabled distinct differentiation of the individual cranial and facial bones. The boundaries between adjacent bones, including the frontal, parietal, temporal, maxillary, nasal, lacrimal, zygomatic, and occipital bones, as well as major foramina and external bony landmarks, were readily distinguished, facilitating understanding of their anatomical layout and spatial relationships (Figures 1C and 1D). The femur models derived from an adult ox demonstrated the proximal and distal extremities, incorporating the head, trochanters, trochanteric fossa, shaft, condyles, and trochlea, all of which were distinctly preserved in the reconstructed model (Figures 1E and 1F). The lumbar vertebrae of an adult ox were meticulously replicated, demonstrating the sequential arrangement of the vertebrae while maintaining the normal orientation, intervertebral spacing, vertebral bodies, transverse processes, spinous processes, and articular processes. This

precise reproduction facilitated a clear visualization of their 3D anatomical relationships (Figures 1G and 1H). The 3D imaging method reproduced soft-tissue specimens from the head of the adult ox (Figure 2). The 3D models preserved the morphology and major anatomical features of the nasolabial region and hard palate, including the muzzle and nostrils, and surrounding soft tissues, whereas the nasal cavity model displayed the nasal septum, nasal conchae, and meatuses. The hard palate model exhibited the palatine raphe and transverse palatine ridges with adequate anatomical detail suitable for interactive visualization and educational demonstration.

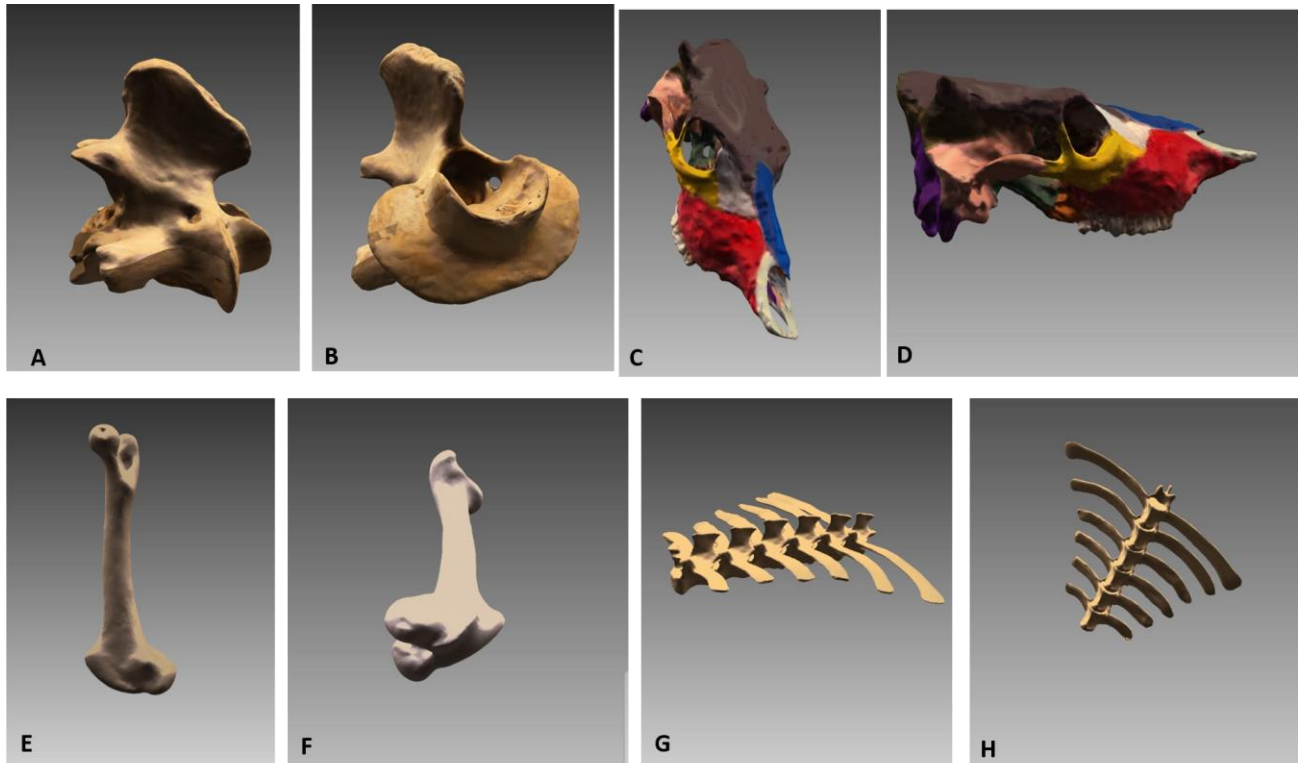


Figure 1. Smartphone-based 3D reconstructed skeletal models of an adult ox. **A and B:** Axis vertebra showing preserved morphology from different views, **C and D:** Skull models, including color-enhanced segmentation illustrating cranial bone differentiation, **E and F:** The femur demonstrates clear proximal and distal anatomical landmarks, **G:** Lumbar vertebrae showing sequential alignment, **H:** Associated rib structures illustrating spatial relationships

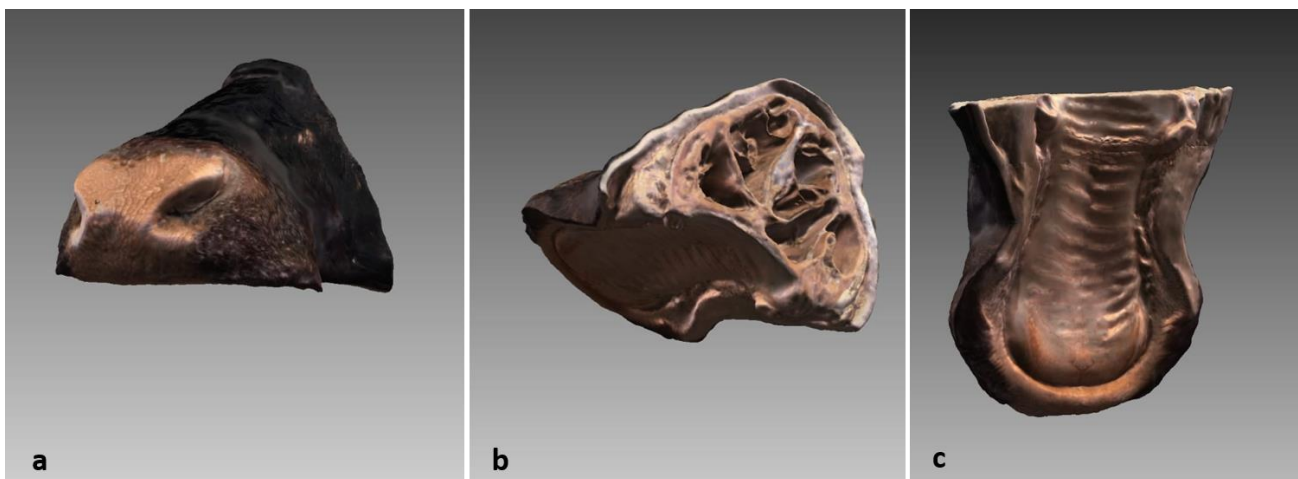


Figure 2. Smartphone-based 3D reconstructed soft tissue models of the head of an adult ox. **a:** External view showing the muzzle and nasolabial region, **b:** Sagittal section of the head showing the nasal cavity and associated internal structures, **c:** Internal view of the oral cavity showing the hard palate and palatine ridges

Plastinated specimens

The modified Elnady plastination method produced durable anatomical specimens with well-preserved morphology (Figure 3). The plastinated heart from an adult male dromedary camel maintained its shape and key features, including a well-defined apex and base, ventricular and coronary grooves, and visible openings of major blood vessels (Figure 3A).

The left kidney from an adult male camel demonstrated preserved structure with a typical bean shape, smooth external surface, distinct poles, a well-preserved hilum, and an outer capsule (Figure 3B). The testicular specimen from an adult male camel retained surface features such as the tunica albuginea, a smooth texture, epididymal attachment, and spermatic cord (Figure 3C). The penis of an adult male camel retained its natural curvature and structure, with the shaft, glans, and connective tissue clearly visible after plastination (Figure 3D).

Shrinkage artifact

Visual examination demonstrated that some tissue shrinkage was observed in plastinated testicular samples from an adult ox (Figure 4). The reduction in size was most noticeable in the testicular parenchyma, whereas the tunica albuginea remained intact, and the overall external morphology of the testes was preserved in ox plastinated models. Conversely, no remarkable shrinkage was observed in the plastinated heart, kidney, or penis of adult male dromedary camels, which retained their gross anatomical morphology following plastination (Figure 3).

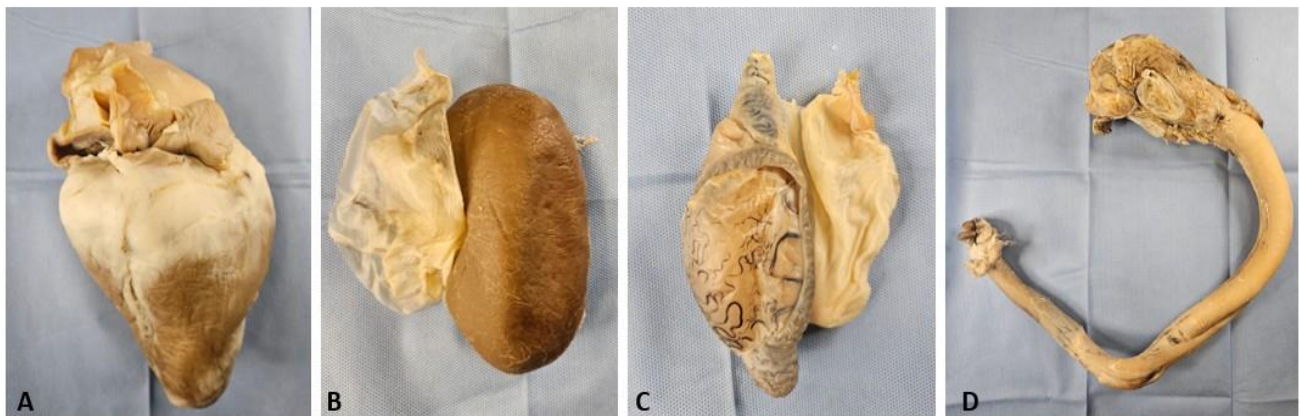


Figure 3. Plastinated anatomical specimens from an adult male dromedary camel prepared with the modified Elnady technique. **A:** Heart with preserved external morphology and anatomical contours, **B:** Left kidney with maintained shape and smooth surface texture, **C:** Testis showing the preserved tunica albuginea, smooth external surface, epididymal attachment, and spermatic cord, **D:** Penis demonstrating the retained natural curvature and preserved shaft, glans, and surrounding connective tissue.

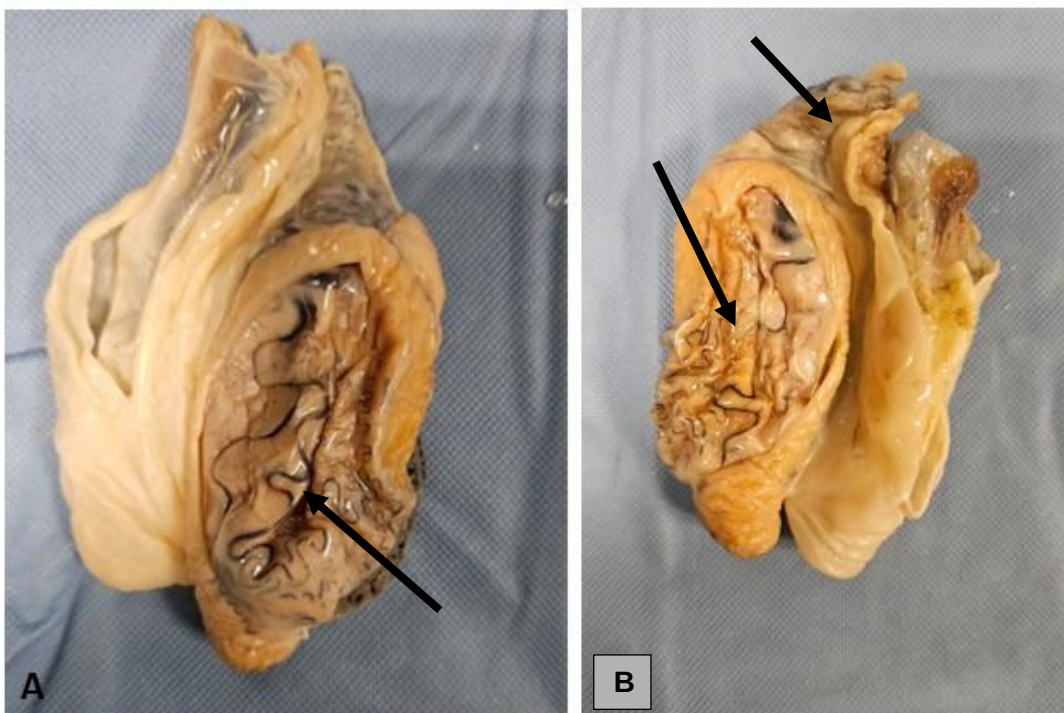


Figure 4. Plastinated testicular specimens from an adult ox showing different degrees of tissue shrinkage and surface wrinkling following the modified Elnady plastination technique. **A** and **B** represent two different plastinated testicular specimens illustrating variation in the extent of shrinkage. The arrows indicate areas of tissue shrinkage and surface wrinkling.

DISCUSSION

The present study demonstrated that smartphone-based photogrammetry successfully generated high-resolution interactive 3D models of both skeletal and soft-tissue specimens of an adult ox. Similar findings were reported by [Silva et al. \(2026\)](#), who used the KIRI Engine to reconstruct equine bones for veterinary anatomy education and found that the resulting models accurately preserved major anatomical landmarks and served as an accessible educational resource. Similarly, [Morichon et al. \(2024\)](#) demonstrated that smartphone photogrammetry can produce anatomically realistic 3D models suitable for anatomy education without requiring specialized imaging equipment. Consequently, these findings indicated that smartphone-based photogrammetry is a reliable and cost-effective method for generating interactive anatomical models for veterinary education.

In addition to ensuring anatomical accuracy, the reconstructed 3D models enabled rotation, magnification, and examination from multiple perspectives, thereby enhancing the visualization of complex spatial relationships among anatomical structures. [Preece et al. \(2013\)](#) and [Wainman et al. \(2018\)](#) documented that physical 3D models improved anatomical learning compared with computer-based counterparts. These findings partially supported the current results, affirming the educational importance of 3D visualization and emphasizing the benefits of interactive 3D anatomical models. Moreover, smartphone-based photogrammetry has demonstrated that detailed anatomical models can be produced without expensive imaging systems or specialized infrastructure, thereby making the approach especially appropriate for veterinary institutions with limited financial and technical resources ([Chang Chan et al., 2024](#)). The findings of [Chang Chan et al. \(2024\)](#) are consistent with the present results, demonstrating clear and detailed 3D anatomical models using smartphone-based photogrammetry. As a result, these accessible digital models have the potential to enhance spatial visualization and supplement traditional anatomy teaching, especially for students with lower spatial visualization abilities as reported by [Höffler \(2010\)](#). However, [Schirone et al. \(2024\)](#) found no notable advantage despite students' preference for 3D models.

The modified Elnady plastination technique resulted in durable, dry, and well-preserved anatomical specimens. The current results aligned with the findings of [Elnady et al. \(2015\)](#), [Elnady \(2016\)](#), and [Bernal et al. \(2022\)](#), who demonstrated that glycerin-based plastination produced durable teaching specimens without requiring vacuum impregnation. Additionally, [Latorre et al. \(2007\)](#) observed excellent preservation of gross morphology in veterinary specimens prepared using conventional plastination techniques, which is consistent with the current findings. In the present study, the modified Elnady technique achieved similar anatomical preservation through a simpler, more cost-effective process, highlighting its suitability for resource-limited veterinary institutions, as previously demonstrated in Egypt ([Elnady, 2016](#)). While the gross morphology of the plastinated specimens was largely maintained, shrinkage was observed in the testicular parenchyma, whereas the heart, kidney, and penis exhibited no notable deformation. [Elnady \(2016\)](#) similarly reported that tissue shrinkage during plastination depends on tissue composition, specimen size, and dehydration conditions, with soft parenchymatous tissues being more susceptible than dense fibrous tissues. The shrinkage observed in the bull testicular parenchyma in the present study was likely attributable to both acetone-induced dehydration and lipid extraction, consistent with the mechanism described by von [Hagens et al. \(1987\)](#). During dehydration, acetone removes water from tissues and dissolves lipids, reducing tissue volume and causing shrinkage. Despite the reduction in testicular parenchyma, the tunica albuginea and external anatomical landmarks remained intact, preserving the overall morphology of the specimens. These findings suggested that the modified Elnady plastination technique offered a practical balance among anatomical preservation, specimen durability, affordability, and procedural simplicity, supporting its applications in routine veterinary anatomy teaching.

CONCLUSION

The present study demonstrated that smartphone-based photogrammetry and the modified Elnady plastination technique were complementary, low-cost tools for veterinary gross anatomy education. These techniques provided interactive digital models and durable anatomical specimens that support practical instruction while reducing reliance on costly equipment and traditional preservation methods, especially for veterinary institutions with limited financial and technical resources. Using digital models for visual learning alongside plastinated specimens for tactile learning provides a multimodal educational approach that addresses different learning preferences. The present study was limited by the small sample size and the absence of student-based evaluation of the educational effectiveness of 3D imaging and plastination techniques. Future studies should investigate the educational impact of these techniques through comprehensive student-based assessments, including anatomical knowledge, understanding of the 3D relationships among anatomical structures, student engagement, and satisfaction.

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Authors' contributions

Ayoub Ben Hadj contributed to the study's conceptualization, methodology, and investigation. Abdelrahman Mohamed Ali Elseory contributed to the investigation, provided funding, and helped review and edit the manuscript. Mohammed Sami Aleid contributed to the conceptualization, methodology, and investigation. Abdulla Al-Ramadan contributed to the conceptualization, methodology, and investigation. Mortada Mahgoub Osman Elhassan contributed to the review and editing of the manuscript. Lemiaa Eissa contributed to the conceptualization and editing of the manuscript. Abdelhay Mohamed Ali contributed to the review and editing of the manuscript. Thnaian Ali Al-Thnaian contributed to the review and editing of the manuscript and supervised the study. All authors read and approved the final edition of the manuscript.

Availability of data and materials

All data generated and analyzed during this study are included in this manuscript. No supplementary data files were generated. Requests for additional information related to this study should be directed to the corresponding author upon reasonable request.

Competing interests

The authors declare no conflicts of interest.

Ethical considerations

The authors confirmed that this manuscript is original, has not been published previously, is not under consideration for publication elsewhere, and contains no redundant publication, plagiarism, data fabrication, data falsification, or other forms of scientific misconduct. The authors employed the Superhuman AI Writing Assistant (Superhuman Platform Inc., USA, Version 2024.6) to enhance the manuscript's grammar and linguistic quality. The authors meticulously reviewed, edited, and authenticated all AI-generated suggestions and take full responsibility for the manuscript's accuracy, integrity, and final content. All authors have approved the originality of the manuscript before submission and during the peer review process, and accept full responsibility for its content.

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