






ORIGINAL ARTICLE

Management of cardiorespiratory function of rabbits by a customized chest drain: An experimental study

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ABSTRACT

Objective: This study aimed to salvage the study population from the fatality that occurs due to iatrogenic injury to the thoracic cavity's pleural membrane.

Materials and Methods: An experimental study of temporomandibular joint arthroplasty with costochondral graft was carried out on 72 healthy '*Oryctolagus cuniculus*' species of male rabbits. The rabbits were distributed into two age groups: growing (3–4 months) and adult (12–18 months). All the procedures were carried out under general anesthesia with xylazine hydrochloride and ketamine hydrochloride after calculating the doses, maintained by halothane and O₂ inhalations. Out of 72 rabbits, 33 rabbits had accidental perforation of the pleural membrane observed that required a chest drain.

Results: In this study, 21 (63.64%) rabbits received chest drain and salvaged. The rest of the rabbits ($n = 12$; 36.36%) that did not receive any chest drain and died. Most of the rabbits ($n = 17$; 81%) were under the growing group, weighing less than 2 kg and four (19%) were adult rabbits.

Conclusion: This manual chest drain is life-saving for rabbits. It is a new addition to the advancement of thoracic surgery on animals. It is cost-effective and safe. The developed customized drainage system may make it easier to harvest the costochondral graft-related experiments.

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Introduction

Autogenous costochondral grafts (ACGs) have been used for many years to reconstruct diseased or lost temporomandibular joints (TMJs) [1,2]. To replace the growth potentiality of condylar cartilage, which has been regarded as the mandible's major growth center, is the basis for the use of costochondral grafts (CCGs) in growing children. [3]. It is thought that the growth potential of CCGs will restore normal symmetrical faces in growing children. Unfortunately, they still have some disadvantages related to growth patterns and relapse because of the graft's resorption or overgrowth [3–6]. Therefore, the lack of clear experimental evidence of predictability of growth patterns emphasizes further experiments that will provide the basis for applying this knowledge to humans with similar problems.

The CCG comprises a portion of the rib with a segment of costal cartilage and bone on the same strut [7]. The perichondrium covering the cartilaginous part of the CCG is essential for the survival of the graft. It protects the cartilage against fibroblastic invasion, which is responsible for chondrocyte destruction [8]. Again the perichondrium is a dense connective tissue and is the precursor of chondrogenic cells [9], especially the proliferating chondrocytes, which play an essential role in mandibular growth [3]. Experimental studies on animals also showed that costochondral regeneration comes from the perichondrium [10]. But harvesting the CCG from the donor site in a conventional way with an intact perichondrium increases the chances of pleural membrane perforation [11,12]. It is one of the rare donor site morbidities in humans, despite

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its favorable anatomical and adaptive characteristics as a graft. This distortion of the chest wall might reduce the chest wall compliance, pneumothorax, lung collapse, and ultimate death [13]. This type of catastrophe is usually managed either by thoracic surgeons, pulmonary physicians, or radiologists through chest drain tubes [14]. A chest drainage system is commonly used in humans when penetrating chest trauma for maintaining negative chest pressure in the post-surgical period to allow lung expansion [13].

It is anatomically evident that the rabbit's pleural membrane is distinctly fragile [15]. According to Du et al. [16], animal studies on the Japanese white male rabbits of 20 weeks of age discovered the reason for pleural injury during CCGs isolation; it was due to the intimate attachment of pleura to the inner surface of the rib around the costochondral junction [16]. Indeed, during an experimental study on rabbits, there is a chance of accidental exposure of the thoracic cavity and the penetrating injury to the pleural membrane. On the other hand, there is no reference to support the evidence on the management of rabbits in the case of iatrogenic pneumothorax; it is a rare unavoidable complication of the rib-harvesting procedure of rabbits.

In this study, a novel modified CDS has been developed distinctly for rabbits to salvage them from the fatality in the preoperative and the immediate postoperative period; the system has been developed based on the original 'one bottle' system used for humans [14,17].

Materials and Methods

To identify the potential growth center of ACG in the TMJ, a prospective experimental study was carried out on 72 clinically healthy '*Oryctolagus cuniculus*' species, outbred New Zealand white male rabbits during the period of 1 July 2018–30 June 2020. The rabbits were distributed into two age groups: growing (3–4 months) and adults (12–18 months). The body weight of the rabbits ranged from 1.5 to 2.5 kg. They were purposively collected from authentic sources; these were Animal Resources Branch, Laboratory Sciences and Services Division, icddr,b, Dhaka, and Dhaka University Market, Katabon, Dhaka, Bangladesh. They were kept in a standard environment with a nutritious diet. The procedures, including euthanasia, were carried out according to the guidelines of the Animal Experimentation Ethics Committee (AEEC) of Bangladesh Agricultural University (BAU) [ID No.- BAU (Surgery & Obstetrics Dept.)-17/9/2014-(01)], Mymensingh, and Institutional Review Board (IRB) of Bangabandhu Sheikh Mujib Medical University (BSMMU) (IRB; ID No. Bangabandhu Sheikh Mujib Medical University /2015/509), Dhaka, Bangladesh.

The surgical procedures were carried out under general anesthesia in the Central Veterinary Hospital (CVH), 48 Kazi Alauddin Road, Dhaka-1000, Bangladesh. The carcasses were disposed of as per rules of the Central Disease Investigation Laboratory (CDIL), 48, Kazi Alauddin Road, Dhaka-1000, Bangladesh, by instantaneous destruction of the body in the incinerator.

Anesthesia

All the rabbits received general anesthesia under the close supervision of an expert veterinary anesthetist. The anesthetic agent was xylazine hydrochloride (3 mg/kg body weight, intramuscular route), followed by the use of ketamine hydrochloride (30 mg/kg body weight intramuscular route) after 5 min [18]. To maintain the anesthesia, halothane inhalation (up to 3.5%) with a vaporizer and O₂ (4 l/min) incrementally used as a supplement through a face mask [19]. Adequate anesthesia was confirmed when the ear pinch response and the pedal withdrawal reflex were absent [20].

Surgical procedure

Before surgery, the rabbits were acclimatized for 10 days and confirmed as healthy based on the physical examination, complete blood count, and chest x-ray [21]. After proper shaving and all aseptic preparations under general anesthesia, TMJ arthroplasty with an ACG was carried out in collaboration with a skilled veterinary surgeon. A slightly curved incision of 3–4 cm was made below the folded elbow parallel to the fifth or sixth rib level. Dissection was continued by undermining the fascia and thin panniculus carnosus muscle; the latissimus dorsi muscle was retracted. Then, the pre-planned CCG was isolated after giving an incision along with the pectoralis major muscle. The perichondrium covering the cartilage and the periosteum covering the rib a few millimeters beyond the costochondral junction were kept intact with the graft. Special attention was given to prevent the exposure of the thoracic cavity and injury to the pleural membrane. In the thoracic cavity perforation, a customized chest drain was immediately inserted to maintain negative thoracic pressure and treat a suspected tension pneumothorax [13,17]. Then, closing was carried out in layers with tight closure around the chest drainage tube.

Aim for the chest drainage system

A customized CDS was aimed at in experimental rabbits with an iatrogenic pneumothorax to drain the entrapped air, blood, and fluid and restore the negative pleural pressure, thereby facilitating lung expansion and reinflating a collapsed lung.

Device design

This customized water seal CDS's basic principle was to drain air and fluid from the pleural cavity on an exhalation and prevent these from returning to the lung or mediastinum on inhalation. Thus, the system maintained a one-way valve and continuous drainage (Fig. 1) [17].

This system consists of a 20 ccs disposable syringe (DPS), half-filled with normal saline with two semi-rigid tubes (tube A and tube B) fixed at its upper end. Tube A (tube to the rabbit) connected with the thoracic drainage tube placed in the pleural cavity, which collects air and fluid. Tube B (tube to the suction apparatus) was fixed above the saline water level and connected with a suction control bottle (low-pressure suction apparatus). To maintain an effective water seal, tube A was dipped in saline water so that its tip was 3–5 mm beneath the upper surface of saline water (water seal system). This water seal system acts as a one-way valve that includes a place for drainage collection and prevents air or fluid from returning to the chest. To create a pressure gradient or to apply negative pressure, tube B was used for air suction and decompression. The suction control bottle allows the controlled application of suction (max. vacuum: 15 ± 1.5 kpa; max. capacity: 700 ml/min). The suction machine contained a manometer with a standard scale of readings to measure the pleural cavity's negative pressure. This negative pressure was maintained by the gradual rise of suction pressure (starting from -5 cm H_2O reaching -10 cm H_2O) [14,17]. The high negativity, evidenced by the water level rise in the water seal, was observed for constant or intermittent bubbling in the water seal chamber

indicated an air leak. Although not introduced in this manual system, the air-leak meter was made up of a numbered scale, labeled from 1 (low) to 10 (high), which was just above the saline water level of the 20 ccs DPS. The greater the height of air bubbles in the syringe, the more the air leaks. These air leaks either increased or decreased were monitored by documenting the number from the scale. Any drainage from the chest flowed into the water seal system allowed easy measurement, thereby recording the drainage amount. It was essential to keep the chest drainage unit upright at all times to maintain the saline water level.

Methods of drain fixation

A nasogastric tube (6F) was used as a chest tube keeping in mind the tube's length and internal diameter, which is responsible for drainage and risk of blockage. Then, the tube was held with an artery forceps and usually advanced through one of the accidentally created holes at the mid-axillary level into the pleural space. The tube was placed carefully not to enter into the potential space between the pleura and chest wall. The tube's tip was directed by direct visualization medially and upward, more likely to abut the mediastinum along with the upper border of the isolated graft. The tube remained inside the chest wall up to the chest tube's last hole (approx. 5 mm) in the pleural cavity. The tube was proceeded carefully not to go too far into the chest and injure the lung or the heart. Further attention was given not to direct the tube too far beyond the mediastinum limit because it would have resulted in contralateral pneumothorax. The diaphragm, liver, or spleen might be lacerated if the rabbit was not appropriately positioned, moved, or the chest tube was inserted too low. Usually, immediately after a pleural tear, the lung retracted from the

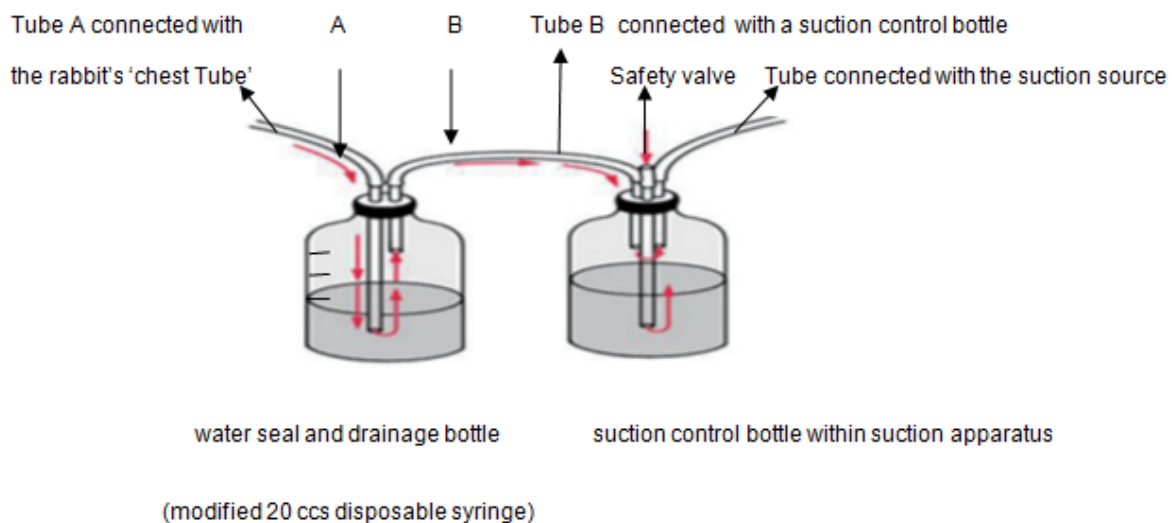


Figure 1. A modified 'one bottle' chest drainage system.

chest wall and helped the procedure. A peroperative chest X-ray (A-P view) was obtained to avoid complications and execute a successful chest drain insertion. Tube A was connected with the rabbit's chest tube, and tube B was associated with the previously built suction drainage system. The suction tube was adjusted as needed until a steady stream of bubbles was produced in the saline water column. If tube A was not appropriately inserted in the pleural space, no air would be suck out, and the level of the saline water column would not vary with respiration [14,17].

Mechanism of action

When the pleural pressure was positive, the pressure within tube A became positive as the pressure was more than the depth to which the tube was immersed in the saline water. Hence, the saline water did not enter into tube A. Only when this high positive air pressure in tube A was sucked out and released through tube B by the controlled suction system, the pressure became negative, and then the saline water moved from the 20 ccs DPS to tube A and sealed the tube. Thus, no air entered into the pleural cavity or tube A. This is the water seal drainage system with the saline water in the DPS blocked the pleural cavity from the drained air or fluid and the exterior.

Securing the tube

Before the surgical procedure, the chest tube was kept in place by suturing it with 1-0 or 2-0 silk or other non-absorbent sutures with the abdominal wall. After the graft isolation and chest tube insertion, a purse-string stitch was given around the wound edge to secure the tube in position and to prevent air and water leakage. Then, the closure was made in layers. The sutures' ends were tied tightly around that tube which provided an airtight seal. A second suture was placed four fingers apart from the incision site along the abdominal wall; thereby, it did not slip from the location.

An antibiotic ointment was applied along with the incision site. A petroleum gauze dressing was given and fixed with an adhesive tape. After the operation, a radiograph, either an AP or LP view, was taken to check the chest tube's position and effectiveness on the iatrogenic pneumothorax (Fig. 2A-J).

Monitoring

The rabbit's cardiorespiratory status and O₂ saturation were monitored throughout the procedure. The recovery time was when ear pinch reflex and pedal withdrawal reflex parameters returned to the normal preoperative state and when the rabbit could walk [20].

Chest tube removal

Postoperatively, the chest tube was removed when there was no air or fluid leak or drainage for over 6 h; rabbits

were fully orientated and stable and recovered from anesthetic effects.

Complications

Based on the chest drain associated complications in the postoperative period, to get an idea about the efficacy of the chest drain, the rabbits were divided into four groups: cardiorespiratory deficiency (Group I), systemic infection (Group II), others (Group III), and technical (Group IV). In Group I, the most remarkable complication was cardiorespiratory deficiency (Table 1), which aggravated without delay over time due to the pleural tear. In this study, this particular condition was managed only by a customized chest drain. This observed complication was supported by Richter and Ragaller [13]. In Group II, two rabbits developed signs and symptoms of pneumonia, and another one had symptoms of septicemia. However, the infected group of rabbits was managed correctly by administrating broad-spectrum antibiotics.

Additionally, the infection-related respiratory distress was managed by frequent mechanical ventilation along with an O₂ supplement. In Group III, one rabbit developed surgical emphysema, and another one set drain insertion site abscess. The surgical emphysema was managed by medication and the natural way. The insertion site infection was managed by drainage of pus. Also, a specific antibiotic was administrated after the culture-sensitivity test. Finally, in Group IV, only one adult rabbit's chest tube was blocked by a blood clot and dislodged. The complications mentioned above are also reported by other authors [22,23] (Table 2) (Fig. 3).

Results

In this experimental study, pleural membrane perforation occurred in 33 (45.83%) out of 72 rabbits. The distorted chest wall configuration with pleural tear resulted in cardiorespiratory complications and peroperative death. Initially, no life-saving maneuver was thought to restore the rabbits from the casualty, resulting in a pleural tear. This led to the death of 12 (36%) rabbits peroperatively. The death rate of growing rabbits was 24% ($n = 8$), and of the adult rabbits was 12% ($n = 4$). In this study, the most significant complication was a cardiorespiratory deficiency. But there was no statistically significant difference between the growing and adult groups of rabbits found in all follow-up time (Table 1). After the insertion of a customized chest drain, 21 (63.64%) rabbits were salvaged. Most of them (17) were growing rabbits (81%) (body weight < 2 kg) and four were adult rabbits (19%). There was no peroperative death that occurred after the insertion of the chest drain. All the rabbits survived, except for a few minor complications; those were managed accordingly. A statistically significant difference of complications was

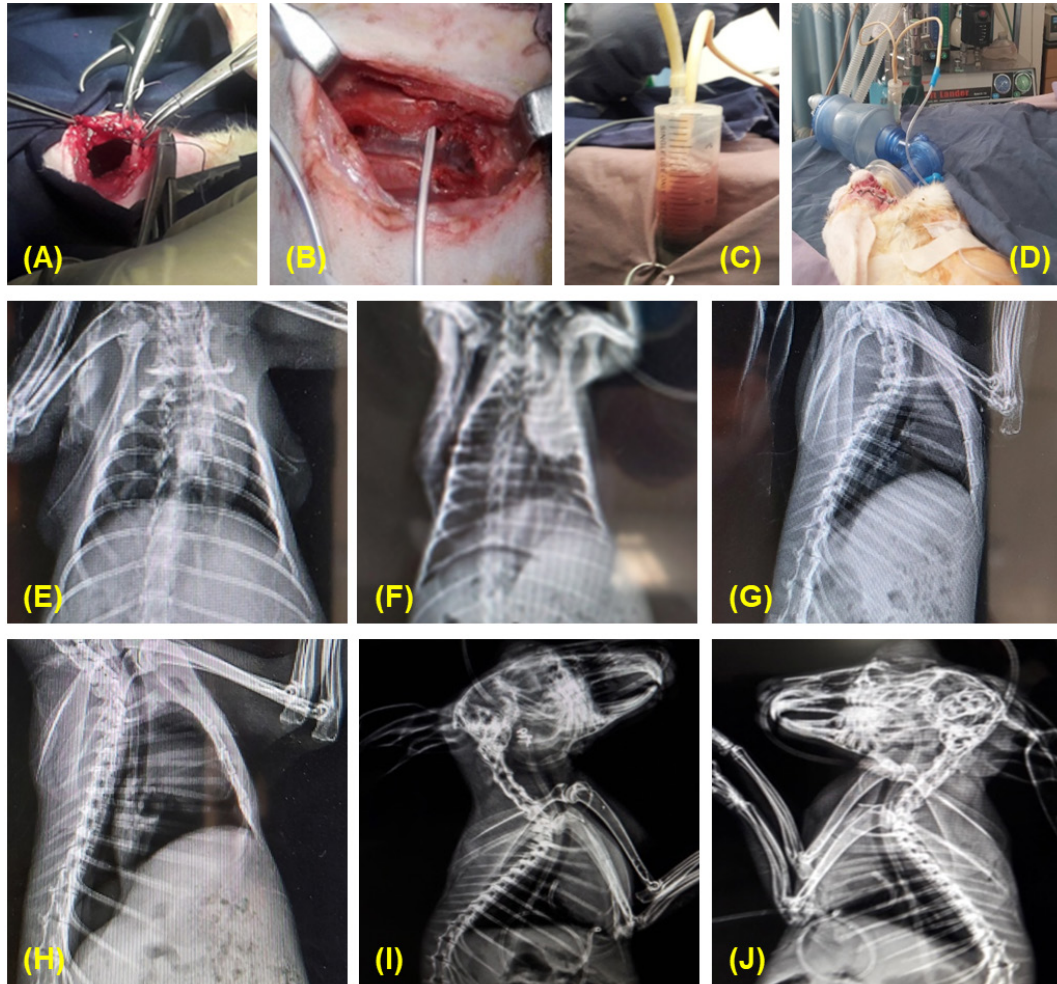


Figure 2. A customized chest drain for the management of cardiorespiratory function of the rabbit and the perioperative radiographic changes. (A) Enlarged thoracic cavity opening due to entrance of air in the pleural cavity during breathing cycle and atmospheric pressure. (B) Perforated pleural membrane immediately after the chest drain fixation. (C) 20 ccs DPS with two attached semi-rigid tubes. (D) Chest drainage system set-up in the operation theatre. (E) Preoperative X-ray chest (A-P) view showing normal chest. (F) Peroperative X-ray chest (A-P) view showing features of right-sided pneumothorax; air pushed the lungs and heart toward the left side. (G) Preoperative X-ray chest right lateral view. (H) Peroperative X-ray chest right lateral view after pleural membrane perforation. (I) Chest x-ray, right lateral view showing an *in-situ* chest drainage system. (J) Chest x-ray, left lateral view showing an *in-situ* chest drainage system.

Table 1. Mean distribution of SPO₂, respiratory rate, and heart rate of a growing adult rabbit.

Subject	SPO ₂ (mean ± SE) in minute			Respiratory rate (mean ± SE) in minute			Heart rate (mean ± SE) in minute		
	Immediate without drain	After 15 with drain	After 30 with drain	Immediate without drain	After 15 with drain	After 30 with drain	Immediate without drain	After 15 with drain	After 30 with drain
Growing n = 12 (Min, Max)	63.8 ± 3.1(50, 83)	93.2 ± 0.6(91,99)	96.9 ± 0.4(94,99)	66.6 ± 5.0(44,100)	48.6 ± 2.6(38,66)	45.5 ± 2.0(35,58)	54.5 ± 4.6(24,85)	108.8 ± 6.0(94,168)	155.8 ± 8.5(98,210)
Adult n = 3 (Min, Max)	70.0 ± 8.7(58, 87)	94.7 ± 1.2(93,97)	97.3 ± 0.9(96,99)	74.3 ± 13.4(55,100)	45.0 ± 5.0(40,55)	38.3 ± 2.0(35,42)	67.0 ± 8.4(52,81)	132.0 ± 13.3(110,156)	144.7 ± 7.5(132,158)
Total n = 15 (Min, Max)	65.1 ± 3.0(50, 87)	93.5 ± 0.5(91,99)	97.0 ± 0.4(94,99)	68.1 ± 4.6(44,100)	47.9 ± 2.2(38,66)	44.1 ± 1.8(35,58)	57.0 ± 4.1(24,85)	113.4 ± 5.8(94,168)	153.6 ± 7.0(98,210)
Test	$F(df) = 61.47 (1.05.26); p < 0.001; \eta^2 = 0.825$			$F(df) = 20.57 (1.32.17.23); p < 0.001; \eta^2 = 0.613$			$F(df) = 45.19 (2.26); p < 0.001; \eta^2 = 0.776$		

Test: one-way repeated measured analysis of variance (ANOVA)
df: degree of freedom

Table 2. Chest drain-related complications in the rabbit model.

Characteristics	Chest drain used and related complications				Chest drain not used	Chest drain required	Total rabbits
	Cardio-pulmonary Group-I	Systemic infection Group-II	Others- Group-III	Technical Group-IV			
Growing rabbit	12	3	2	-	8	25	36
Adult rabbit	3	-	-	1	4	8	36

Chi-square (df) = 11.36(1); $p < 0.001$.
df = degree of freedom.

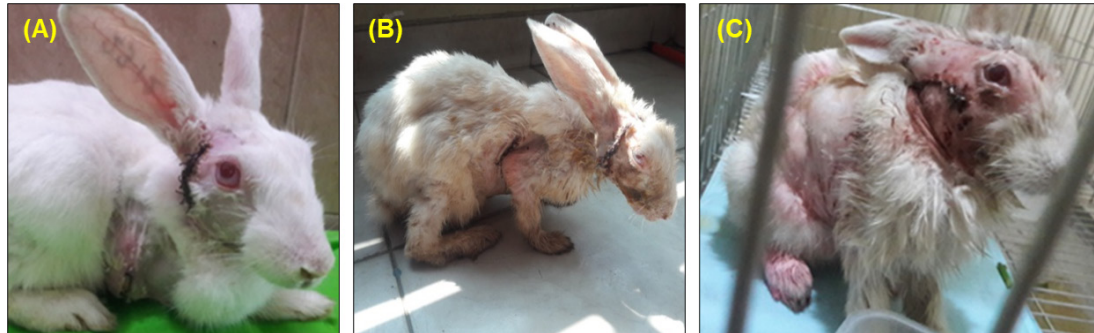


Figure 3. Complications related to the chest drain. (A) Wound infection at the site of an inserted chest drain. (B) Post-septicemic health deterioration. (C) Swelling at the chest region due to surgical emphysema.

also found between growing and adult rabbits wherein the proportion of complications was found to be high among the growing rabbits (Table 2).

Discussion

Exploring a human TMJ seems unethical after reconstruction with an ACG for study purposes. Therefore, considering ethical issues, the New Zealand white rabbit model was chosen to observe the effect of ACG by TMJ arthroplasty. The ACG was harvested from the donor site – thoracic cage. All rabbits were operated on under general anesthesia with the same drug, by the same veterinary anesthetist, and the same team of surgeons. During surgical procedures, no significant blood loss was observed. Consequently, no blood transfusion was required. Special attention was conferred on preventing laceration to the thoracic wall and exposure of the thoracic cavity, thereby injury to the pleural membrane.

Nevertheless, the pleural tear occurred accidentally due to the fragile pleural membrane [15]. Moreover, the pleura's intimate attachment to the costochondral junction's inner surface was observed [16]. However, the less traumatized costal cartilage harvesting technique was used to spare the perichondrial layer that reduced the donor site morbidity than the conventional method [11]. It is essential to preserve the perichondrium for the survival of the graft's chondrocytes [8]. Again, retaining both

the perichondrium and periosteum at the bone–cartilage junction's potentially fragile area is essential to maintain adequate graft strength [12].

On the other hand, the conventional costochondral graft harvesting technique usually increases the chances of a pleural tear [11,12]; it turns to cardiorespiratory complications like pneumothorax, air embolism, cardiac dysrhythmia, and peroperative death [22]. For an ethical issue, life has intrinsic value, and no animal should be killed unnecessarily. Consequently, a modified chest drainage system was aimed to develop for preventing iatrogenic pneumothorax.

Iatrogenic pneumothorax is usually associated with severe respiratory distress, increased respiratory rate, decreased heart rate, dropped O_2 saturation, and a sign of gasping. Our study observed that the above-mentioned conditions always improved dramatically after the immediate insertion of the customized chest drain. Indeed, rabbits ($n = 15$; 71.43%) having respiratory distress signs due to pleural tear became normal after the chest drain insertion, which was observed in the immediate postoperative period. However, three (25%) growing rabbits required bronchodilators within 24 h. But, no rabbit bronchodilators were needed after 24 h. Also, the signs and symptoms of pneumonia were noticed in two rabbits and septicemia in one rabbit (17.65%). Although the infection source was not understandable, it could be susceptible as a subclinical carrier [23]. Another complication was drain insertion site

infection. It might be due to oro-fecal contamination by the rabbit itself during grooming or due to insufficient immunity in the postoperative period. Apart from the mentioned complications, one technical difficulty was also identified in an adult rabbit where the drain tube was blocked by a blood clot, which might have resulted from a kink. Furthermore, re-expansion pulmonary edema is another chest drain-related complication of pneumothorax seen in humans [22]. But, we did not find similar complications in rabbits.

Initially, we tried to manage the perforation sites by suturing. Still, the rapidity of enlargement of the perforation size and the pleural membrane's fragility did not allow us to close the perforations. The rabbit's condition deteriorated quickly, which led to death. It seems that the perforations rapidly enlarged due to the pressure gradient and entrance of air in the pleural cavity during the various phases of the respiratory cycle [14]. The chest drain tube insertion method was carried out by a direct visualization technique that differed from the human where generally dissection of the chest wall is required [22]. So, there was no chance of injury to the vital structures that typically reside in the tube's close approximate.

The experimental study has shown that using the customized chest drain might salvage rabbit's life. However, the fabricated digital chest drainage system is available for humans [17], but no such equipment is available for the rabbit model. Therefore, digital devices for rabbits with required facilities will help secure the future's quality of life. This type of CDS is essential to salvage the rabbit's life, which is yet to be developed. Thereby, this field's advancement will explore safe and inexpensive surgical research on rabbits, especially in the thoracic region.

It can be mentioned that a plain radiograph was taken to rule out the drain tube insertion complications and identify the drain tube's exact position. Although a thoracic CT scan might be confirmatory [24], it was out of our facilities. Second, finding out the sources of chest infection was not possible due to the lack of advanced investigation facilities like cellular analysis; cytology and culture of bronchioalveolar fluid or polymerase chain reaction (PCR), especially for *Pasteurella multocida* and *Bordetella bronchiseptica* [23].

Conclusion

ACGs have been used for many decades to reconstruct non-functional TMJs in pediatric patients. But the cardiorespiratory complication is almost unavoidable due to a pleural tear in the conventional graft harvesting technique. It insisted on developing a customized chest drainage system to salvage the rabbit from the pneumothorax. Firstly, the developed customized chest drainage system would have

been utilized to plan thoracic surgery on rabbits. Secondly, it will facilitate a new addition to managing accidental exposure of the rabbit's thoracic cavity and pleural tear. In addition to that, it is not only cost-effective but also safe. Finally, the developed customized CDS will make it easier to harvest costochondral graft-related experiments.

List of abbreviations

TMJ: Temporomandibular joint, ACG: Autogenous costochondral graft, CCG: Costochondral graft, CDS: Chest drainage system, DPS: Disposable syringe, icddr,b: International Centre for Diarrhoeal Disease Research, Bangladesh.

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Conflict of interests

The authors have no conflict of interest to declare.

Authors' contribution

JS was the principal investigator who designed the study, and wrote the manuscript. QB contributed to supervision and manuscript draft preparation. EH helped in the design implementation. NS and JS carried out all the surgical procedures. AB helped design implementation, critical revisions that were important for the intellectual content, editing, etc. All authors read and approved the final manuscript.

References

- [1] Shaker A. The use of costochondral graft in the management of temporomandibular joint ankylosis. *Egypt J Plast Reconstr Surg* 2003; 27(1):73–83.
- [2] Naujoks C, Meyer U, Wiesmann HP, Jasche-Meyer J, Hohoff A, Depprich R, et al. Principles of cartilage tissue engineering in TMG reconstruction. *Head Face Med* 2008; 4:3; <https://doi.org/10.1186/1746-160X-4-3>
- [3] Raustia A, Pernu H, Pyhtinen J, Oikarinen K. Clinical and computed tomographic findings in costochondral grafts replacing the mandibular condyle. *J Oral Maxillofac Surg* 1996; 54(12):1393–400; [https://doi.org/10.1016/S0278-2391\(96\)90251-7](https://doi.org/10.1016/S0278-2391(96)90251-7)
- [4] Balaji SM, Balaji P. Overgrowth of costochondral graft in temporomandibular joint ankylosis reconstruction: a retrospective study.

Indian J Dent Res 2017; 28:169–243; https://doi.org/10.4103/ijdr.IJDR_141_17

- [5] Lopez EN, Dogliotti PL. Treatment of temporomandibular joint ankylosis in children: is it necessary to perform mandibular distraction simultaneously? J Craniofac Surg 2004; 15(5):879–885; <https://doi.org/10.1097/00001665-200409000-00037>
- [6] Kumar P, Rattan V, Rai S. Do costochondral grafts have any growth potential in temporomandibular joint surgery? A systematic review. J Oral Biol Craniofac Res 2015; 5(3):198–202; <https://doi.org/10.1016/j.jobcr.2015.06.007>
- [7] Ng CY, Watts AC. The use of non-vascularised osteochondral autograft for reconstruction of articular surfaces in the hand and wrist. J Bone Joint Surg Br 2012; 94(11):1448–54; <https://doi.org/10.1302/0301-620X.94B11.30082>
- [8] Obert L, Loisel F, Gindraux F, Tropet Y, Lepage D. Rib cartilage grafting in upper limb surgery: an overview. SICOT J. 2015; 1(13):1–6; <https://doi.org/10.1051/sicotj/2015003>
- [9] Gao Y, Gao J, Li H, Du D, Jin D, Zheng M, et al. Autologous costal chondral transplantation and costa-derived chondrocyte implantation: emerging surgical techniques. Ther Adv Musculoskelet Dis 2019; 11:1–16; <https://doi.org/10.1177/1759720X19877131>
- [10] Matsuura H, Miyamoto H, Kurita K, Goss AN. The effect of autogenous costochondral grafts on temporomandibular joint fibrous and bony ankylosis: a preliminary experimental study. J Oral Maxillofac Surg 2006; 64:1517–42; <https://doi.org/10.1016/j.joms.2006.06.256>
- [11] Yang HC, Cho HH, Jo SY, Jang CH, Cho YB. Donor-site morbidity following minimally invasive costal cartilage harvest technique. Clin Exp Otorhinolaryngol 2015; 8(1):13–9; <https://doi.org/10.3342/ceo.2015.8.1.13>
- [12] Sharma H, Chowdhury S, Navaneetham A, Upadhyay S, Alam S. Costochondral graft as interpositional material for TMJ ankylosis in children: a clinical study. J Maxillofac Oral Surg 2015; 14(3):565–72; <https://doi.org/10.1007/s12663-014-0686-9>
- [13] Richter T, Ragaller M. Ventilation in chest trauma. J Emerg Trauma Shock 2011; 4(2):251–9; <https://doi.org/10.4103/0974-2700.82215>
- [14] Zisis C, Tsirgogianni K, Lazaridis G, Lampaki S, Baka S, Mpoukovinas I, et al. Chest drainage systems in use. Ann Transl Med 2015; 3(3):4.
- [15] Zocchi L. Physiology and pathophysiology of pleural fluid turnover. Eur Respir J 2002; 20:1545–58; <https://doi.org/10.1183/09031936.02.00062102>
- [16] Du D, Sugita N, Liu Z, Moriguchi Y, Nakata K, Myoui A, et al. Repairing osteochondral defects of critical size using multiple costal grafts: an experimental study. Cartilage 2015; 6(4):241–51; <https://doi.org/10.1177/1947603515591628>
- [17] George RS, Papagiannopoulos K. Advances in chest drain management in thoracic disease. J Thorac Dis 2016; 8(Suppl 1):S55–64.
- [18] Alves JR, Lopes LR, Sasasaki T. Perioperative care in an animal model for training in abdominal surgery. Is it necessary a preoperative fasting? Acta Cir Bras 2011; 26(6):5011–547; <https://doi.org/10.1590/S0102-86502011000600022>
- [19] Gil AG, Silván G, Villa A, Millán P, Martínez-Fernández L, Illera JC. Serum biochemical response to inhalant anesthetics in New Zealand white rabbits. J Am Assoc Lab Anim Sci. 2010; 49(1):52–6.
- [20] Peeters ME, Gil D, Teske E, Eyzenbach V, vd Brom WE, Lumeij JT, et al. Four methods for general anaesthesia in the rabbit: a comparative study. Lab Anim 1988; 22:355–6; <https://doi.org/10.1258/002367788780746197>
- [21] Praag EV. Anesthesia of the rabbit Part 1: pre-anesthetic preparations. Copyright (C) 2003–2009. MediRabbit.com. Available via http://www.medirabbit.com/EN/Surgery/Anesthesia/Pre/anes_pre_en.htm (Accessed 15 December, 2020)
- [22] Kesieme EB, Dongo A, Ezemba N, Irekpita E, Jebbin N, Kesieme C. Tube thoracostomy: complications and its management. Pulm Med 2012; 2012:256878; <https://doi.org/10.1155/2012/256878>
- [23] Meredith A. Managing respiratory disease in rabbits. PROCEEDINGS: NAVC Conference 2013 Small Animal. Vet Folio 2019. Available via <https://www.vetfolio.com/learn/article/managing-respiratory-disease-in-rabbits> (Accessed 15 December 2020)
- [24] Ball CG, Lord J, Laupland KB, Gmora S, Mulloy RH, Ng AK, et al. Chest tube complications: how well are we training our residents? Can J Surg 2007; 50(6):450–8.