Clearance of Mucus from Endotracheal Tubes during Intratracheal Pulmonary Ventilation

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Background: Intratracheal pulmonary ventilation (ITPV) is a form of tracheal gas insufflation in which all gas emerges in a cephalad direction from the tip of a reverse-thrust catheter positioned within an endotracheal tube. *In vitro* experiments have shown that this rapid gas flow, with 5 ml/h of normal saline added to the gas flow, continuously removes tracheal secretions from within the endotracheal tube. The authors evaluated its effectiveness to remove mucus in long-term studies in sheep.

Methods: Fourteen healthy sheep were tracheally intubated and ventilated for 3 days with ITPV or with volume-controlled ventilation. Measurements were made of the total amount of secretions within the endotracheal tubes (weight gain), the protein content within the endotracheal tubes, and the increase in resistance to constant air flow. The structure of the airways was examined grossly and histologically. Three additional sheep were ventilated for 24 h with ITPV, and Evans Blue dye was added to the saline to assess the distribution of the infused saline.

Results: There was significantly less mucus in endotracheal tubes of sheep ventilated with ITPV than with conventional ventilation, as shown by minimal weight gain (0.70 ± 0.14 g *vs.* 2.44 ± 0.81 g; $P < 0.001$), lower protein content (14.09 ± 10.79 mg *vs.* 294.99 ± 153.06 mg; $P < 0.001$), and lower resistance to constant air flow (6.15 ± 0.54 cm $H_2O \cdot l^{-1} \cdot s^{-1}$ *vs.* 15.34 ± 5.28 cm $H_2O \cdot l^{-1} \cdot s^{-1}$; $P < 0.001$). Results of gross and histological examinations of the tracheas of animals in both groups were similar, and the tracheas were well preserved. More than 95% of the instilled saline was recovered during ITPV. Only traces of Evans Blue dye were found near the tip of the endotracheal tubes.

Conclusion: Intratracheal pulmonary ventilation makes it possible to keep the endotracheal tubes of sheep ventilated for 3 days free of mucus without suctioning. (Key words: Anesthetic techniques: endotracheal tube; endotracheal suctioning; intratracheal pulmonary ventilation; tracheal mucus. Trachea: pathology; histology. Equipment, design: tracheal tubes.)

TRACHEAL intubation interrupts the transport of tracheobronchial secretions through the mucociliary escalator. Because mucus is not transported through the endotracheal tube (ETT), it becomes inspissated, accumulating in the trachea and in the ETT. To remove accumulated secretions, it is common practice to suction the artificial airways and the trachea.[1] However, suctioning can lead to adverse reactions, damage the mucosa of the airways,[2] and impair mucociliary transport,[3] which may render the respiratory tract more vulnerable to opportunistic infections.

In previous unrelated experiments in sheep managed with intratracheal pulmonary ventilation (ITPV), we found no need for suctioning during prolonged mechanical ventilation (MV) in healthy lungs and in sheep with induced severe acute respiratory failure, using a bubble humidifier, because the ETTs remained clean (unpublished observations). Briefly, ITPV is a form of tracheal gas insufflation in which all inspiratory gas emerges from the tip of a reverse-thrust catheter (RTC) placed within the distal end of the ETT (fig. 1). Gas exits from the RTC continuously through an annular narrow orifice, in the direction away from the carina, creating a venturi. Intratracheal pulmonary ventilation reduces the anatomic dead space fraction, enhances carbon dioxide elimination, and facilitates expiration. We hypothesized that, during the expiratory phase of ITPV, the rapid cephalad gas stream as it emerges from the tip of the RTC might also carry along mucus and debris deposited in the ETT and expel it from within the ETT.

This study in sheep was designed to determine whether ITPV combined with the continuous infusion of saline prevents the accumulation of secretions within airways and within ETTs during 3 days of MV, without suctioning. We conducted a controlled randomized study in healthy, anesthetized, paralyzed sheep maintained on ITPV or on volume-controlled ventilation. We
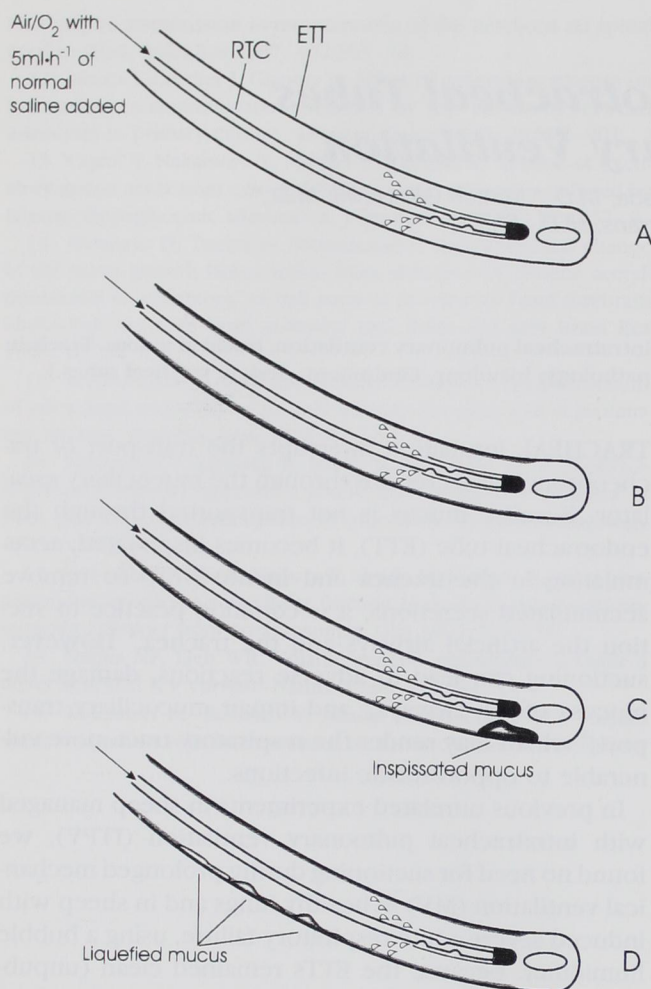

Fig. 1. Transport of mucus deposits from *in vitro* experiments. (A) Infused saline is dispersed into small droplets, exiting the reverse-thrust catheter. (B) Saline droplets form a thin layer of liquid on the inner surface of the endotracheal tube (ETT). (C) This layer of saline is transported cephalad by the intratracheal pulmonary ventilation gas flow. A small sample of mucus is introduced at the tip of the ETT. (D) Mucus is liquified and transported cephalad together with the saline. Mucus and saline are expelled from the ETT.

measured the quantity of accumulated secretions within the ETTs and the macroscopic and microscopic appearance of the trachea.

Materials and Methods

In Vitro *Studies*

We performed studies using a mechanical lung and trachea model placed into an enclosed box with trans-

parent windows at 38°C. We used small in-line water traps at the distal and proximal ends of the ETT to collect water. We performed ITPV with and without 5 ml/h normal saline added to the gas flow. Approximately 0.3 ml of mucus was introduced at the tip of the ETT, and we followed its movement by sight.

Animal Preparation

All animal studies were approved by the Animal Care and Use Committee of the National Heart, Lung, and Blood Institute of the National Institutes of Health. The study was conducted in 14 healthy female sheep of mixed breed, with a mean body weight of 27.79 ± 3.29 (SD) kg (range, 20–33 kg). All sheep were orotracheally intubated with a standard 8-mm ETT (Sheridan Catheter Corp., Argyle, NY). Each ETT was weighed immediately before intubation.

Anesthesia was induced with a loading dose of 25 mg/kg sodium pentobarbital, followed by continuous infusion of $10 \text{ mg} \cdot \text{kg}^{-1} \cdot \text{h}^{-1}$ ketamine chloride. Paralysis was maintained with pancuronium bromide, in a 0.15 mg/kg loading dose, followed by a maintenance dose of $0.06 \text{ mg/kg}^{-1} \cdot \text{h}^{-1}$. After surgical exposure, the right common carotid artery and the right jugular vein were cannulated with 16-gauge catheters for blood pressure monitoring and blood sampling. Normal saline, lactated Ringer's solution, and 5% dextrose were infused at a rate totaling $4–8 \text{ ml} \cdot \text{kg}^{-1} \cdot \text{h}^{-1}$. Five-hundred milligrams ceftriaxone sodium (Rocephin; Roche, Nutley, NJ) was given routinely intravenously every 12 h. A 10-French Foley catheter was inserted to monitor urinary output. Esophageal temperature was monitored continuously and maintained at 39°C with a heating/cooling blanket (Blanketrol; Cincinnati Subzero, Cincinnati, OH). Systemic blood pressure and central venous pressure were measured with calibrated pressure transducers (Statham P23D; Gould, Cleveland, OH) and continuously recorded on an eight-channel amplifier/recorder (3800 signal conditioner; Gould). The midaxillary line was considered the zero reference point. Arterial blood gases, electrolytes, and blood glucose were measured each hour (Stat Profile 9; Nova Biomedica, Waltham, MA). Blood cell counts were performed every 4 h (Baker System 9000 hematology counter; Seronco Baker Diagnostics, Allentown, PA). Urinary output was measured hourly. Sheep were positioned prone throughout the experiment.

Ventilation

After operation, the sheep were allowed to stabilize for 20 min on continuous positive-pressure ventilation

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on a Siemens Servo 900C ventilator (Siemens, Elema, Sweden) connected to the ETT with 22-mm low-compliance silicone rubber tubing. Tidal volume was 10 ml/kg; respiratory rate (RR) was 12 breaths per minute; inspiratory-to-expiratory ratio was 1:2; positive end-expiratory pressure was 5 cm H_2O, and the inspired oxygen fraction (F_{IO2}) was 0.4. Throughout the three-day duration of the study the trachea was not suctioned and maneuvers to facilitate removal of mucus were not performed.

Sheep were randomly assigned to two groups. Sheep in the control group (n = 7) were ventilated with volume-controlled ventilation on a Siemens Servo 900C ventilator. Tidal volume was 10 ml/kg; RR was 10–18 breaths per minute; F_{IO2} was 0.3; inspiratory-to-expiratory ratio was 1:2, and positive end-expiratory pressure was 5 cm H_2O. A standard Conchatherm III (Hudson Respiratory Care., Tennecula, CA) was used to humidify inspiratory gases. The inspiratory gas lines were insulated with plastic wrap to prevent heat loss and precipitation in the tubing. The temperature of the inspiratory gas was maintained at 37°C; all liquid exiting was collected in a water trap in the expiratory limb of the silicone rubber tubing. We measured the quantity of liquid collected in the water trap and the protein concentration (Pierce BCA Protein Assay; Pierce Co., Rockford, IL).

Animals in the ITPV group (n = 7) were ventilated as previously described.[4] A 7-French RTC (Cook, Bloomington, IN) was advanced through the ETT to within 0.5 to 1 cm of the tip of the ETT. The ITPV gas flowed through a Conchatherm III humidifier. The lid of the standard humidifier cartridge was fortified with epoxy resin to withstand a pressure of up to 1 atmosphere. Gas entered the canister through vinyl tubing with multiple side holes positioned approximately 5 mm above the surface of the water, providing good gas mixing. Water in the cartridge was heated to 39–41°C. In this manner, humidified gas entered the RTC through insulated PVC tubing at a temperature of 36–38°C. Normal saline was infused at a rate of 5 ml/h directly into the inlet of the RTC using a syringe pump (Baxter model AS 20A; Baxter Healthcare Corp., Hooksett, NH). The RTC was weighed before each experiment after flushing it with normal saline and 10 l/min of air (wet weight). A Siemens Servo 900C ventilator was used to control RR, inspiratory-to-expiratory ratio, and external positive end-expiratory pressure. The ventilator was placed in the pressure control mode, with the inspiratory pressure set at zero. At those settings, the ventilator

functioned only as a shutter because all gas was delivered through the RTC.[4] Intratracheal pulmonary ventilation gas flow was 9.5 to 14 l/min, RR was 10–18 breaths per minute, and tidal volume was 10 ml/kg. Positive end-expiratory pressure was set at 6–8 cm H_2O; because of the features of the RTC, this resulted in a carinal positive end-expiratory pressure of 5 cm H_2O. Both groups of sheep were ventilated at comparable tidal volume and RR. Because ITPV enhances carbon dioxide removal by flushing the anatomical dead space, to achieve normal partial pressure of carbon dioxide (between 35 and 45 mmHg) in both groups of sheep, we added carbon dioxide gas to the ITPV gas flow to increase the inspired fraction of carbon dioxide to between 0.02 and 0.04. All liquid exiting from the ETT was collected in a water trap and analyzed as in the control group (fig. 2).

Three additional female sheep (24 ± 1.5 kg) were ventilated with ITPV at the same setting as in the ITPV group, except that Evans Blue dye (43.2 µg/ml) was added to the saline infused into the RTC. All liquid exiting from the ETT was collected in a water trap and measured hourly. The concentration of Evans Blue dye infused into the RTC, and in the liquid collected in the water trap, was measured photometrically at a wavelength of 270 nm. After 24 h of MV, sheep were killed, and the tracheas were opened. The airways and the lungs were carefully dissected and inspected for the presence of Evans Blue dye; areas with blue staining were photographed.

Study of Tracheal Secretions

After 72 h of MV, the sheep were killed with an intravenous injection of sodium pentobarbital and potassium chloride. At autopsy, the trachea and the lungs were exposed, the position of the ETT was noted, and the trachea was opened through a right lateral longitudinal incision. Any airway secretions or tracheal injury were noted. The ETT was removed and inspected. All adherent secretions were wiped from the outer surface of the ETT. The ETT and the RTC were weighed, and the change in weight from before intubation was calculated and recorded. Any secretions adherent to the RTC were noted. The airway resistance[5] of all ETTs was calculated *in vitro* from the pressure decrease across the ETT at a constant air flow of 1 l/s. Mucus deposits within the ETTs were first dissolved in 1% sodium dodecyl sulfate (Quality Biological Inc., Gaithersburg, MD), after which we measured the total protein concentration. The total protein content within the ETT was computed from

A. Water Trap
B. Humidifier
C. Ventilator
D. Saline Injection
E. Endotracheal Tube
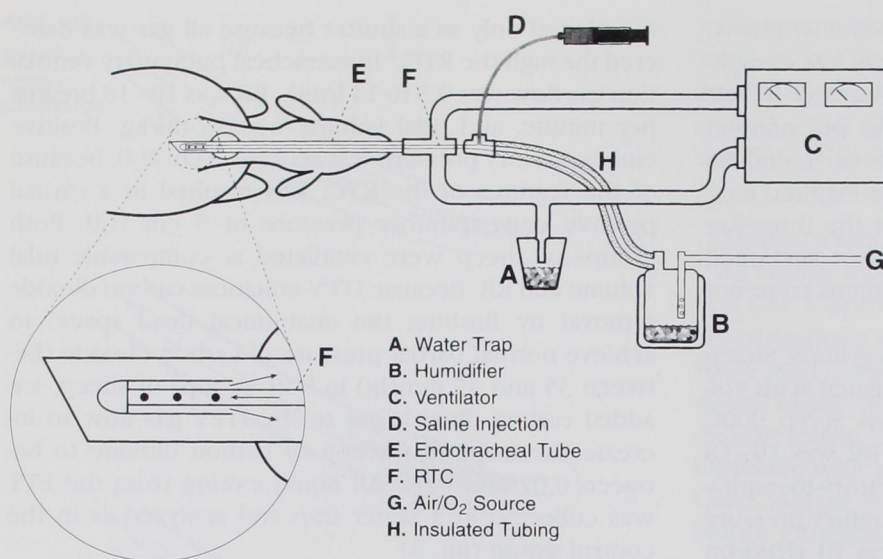F. RTC
G. Air/O₂ Source
H. Insulated Tubing

Fig. 2. The intratracheal pulmonary ventilation system. See the text for details.

the product of the volume of solution and the protein concentration.

Gross secretions within the ETT and the trachea were classified based on color and lucence (as shown by transillumination) as clear, opaque, or purulent.[6] The trachea and the ETT were photographed and graded using a modification of the scoring system of Reali-Forster *et al.*[7]

Histopathologic Studies

For histologic examination, samples of the trachea were taken from the following locations: 1) above the cuff of the ETT, 2) immediately distal to the tip of the ETT, 3) in the middle of the trachea, 4) at the level of the right upper lobe bronchus, and 5) at the level of the carina. Because of its length, the sample from the second location was subdivided into two sections, which were designated B_1 and B_2. Each sample included at least four tracheal rings and was fixed in 10% formalin. Samples for microscopic analysis were taken from the left side of the trachea, unless the lesions appeared more severe on the right. Tissue blocks were dehydrated and embedded in paraffin. Sections (5-μm thick) were stained with the hematoxylin and eosin, with the Movat pentachrome, and periodic acid-Schiff methods. Slides were evaluated by a pathologist unaware of the mode of ventilation, using a modification of the scoring system of Reali-Forster et al.[7] For this purpose, the following microscopic changes were specifically evaluated: tracheal epithelial injury, inflammatory reaction,

edema, vascular congestion, hemorrhage, glandular epithelial injury, and extent of accumulation of mucus within the ducts of the submucosal glands. Those changes were graded as 0 = absent, 1 = mild, 2 = moderate, and 3 = severe. A mean score was assigned to each field of view, and the total number of scores was averaged to determine the final mean score (rounded to the nearest whole number) for the entire section. In addition, measurements of the Reid index were made. The microscopic finding that most consistently correlates with hypersecretion of the mucus is enlargement of the mucous glands, and this can be appraised conveniently using the gland-to-wall ratio (Reid index),[8] the ratio of the thickness of the lobules of mucus glands to the distance between the perichondrium and the basement membrane of the bronchial epithelium. This ratio provides a measure of the volume of the mucus glands. In the present study, the Reid index was calculated at three different sites in each of the tracheal regions (1–5) listed previously.

Statistical Analysis

We used the two-tailed Student's *t* test to compare weight gain, total protein content, resistance of the ETTs, and the Reid index. The analysis of variance for repeated measurements was used to compare the mean oxygen tension throughout the 72-h study. We used the Mann-Whitney test to evaluate differences in the scores of the microscopic changes. All values are given as mean
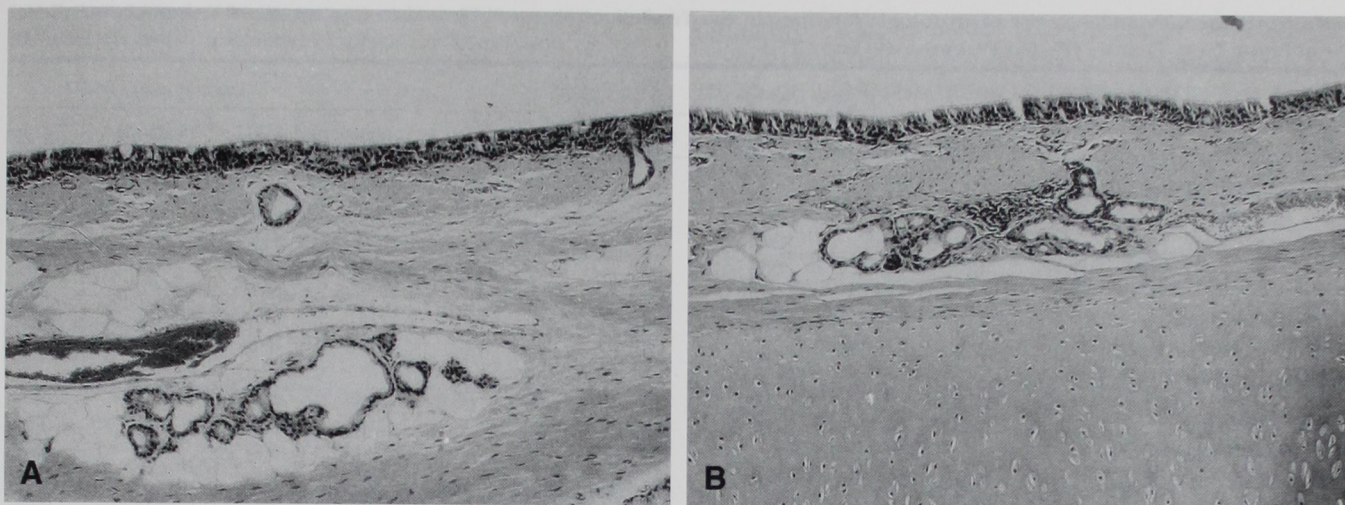
Fig. 3. Representative sections from the B_2 region of the trachea of sheep intubated and mechanically ventilated with volume-controlled ventilation (A) or ventilated with intratracheal pulmonary ventilation (B). Hematoxylin and eosin stain. Original magnification, x200. There is no damage to the tracheal epithelium. The submucosal glands show mild dilation.

± SD. A probability value of 0.05 or less was considered significant for all statistical tests.

Results

In Vitro *Studies*

When air and oxygen exits the RTC, together with infused saline, the saline droplets form a continuous layer of liquid within the distal ETT, and all saline is gradually expelled (figs. 1A and B). A small sample of mucus introduced within the tip of the ETT (fig. 1C) becomes gradually liquefied and is expelled (fig. 1D).

In Vivo *Studies*

Blood pressure, urinary output, electrolytes, and blood cell counts remained within the normal range throughout the experiment. At the end of the study, the mean oxygen tension (± SD) of the control group was 177 ± 16.7 mmHg, and the mean oxygen tension of the ITPV group was 164 ± 12 mmHg ($P = 0.081$). The ETTs of all control sheep had significant accumulations of opaque secretions. There were minimal clear secretions in the ETTs of sheep ventilated with ITPV. The weight of the ETTs increased by 2.44 ± 0.81 g in the control group and by 0.70 ± 0.14 g in the ITPV group ($P < 0.001$). The weight of a new 8-mm ETT immersed in saline for 3 days increased by 0.31 g. The weight of the RTCs did not increase, and there were no adherent secretions. The mucus deposits within the

ETTs of the ITPV group contained 14.09 ± 10.79 mg protein, whereas those in the control group contained 294.99 ± 153.06 mg protein ($P > 0.001$).

The resistance of the ETTs at an air flow of 1 l/s was 15.34 ± 5.28 cm $H_2O \cdot 1^{-1} \cdot s^{-1}$ in the control group and 6.15 ± 0.54 cm $H_2O \cdot l^{-} \cdot s^{-1}$ in the ITPV group ($P < 0.001$). The resistance of a new, dry 8-mm ETT of the same series was 5.20 cm $H_2O \cdot l^{-1} \cdot s^{-1}$.

The gross appearance of the trachea was normal in both groups. In one sheep in the ITPV group, and in two sheep in the control group, there was moderate amounts of mucus in the trachea; in one control sheep, the mucus in the trachea was opaque. The lungs appeared normal in all sheep, except for some atelectasis of the right upper lobe in one sheep of the ITPV group and in two control sheep. On histologic examination, the tracheal epithelium below the tip of the ETT was well preserved in both groups (figs. 3A and B).

In the sheep of the ITPV group, we infused saline into the RTC at a rate of 5 ml/h, yet we collected 10.44 ± 1.68 ml/h of liquid in the water trap containing 33.38 ± 16.58 mg protein per 100 ml; or 751 ± 121 ml of liquid in 72 h, containing a total of 260 ± 9 mg protein. In the control sheep, we collected 0.93 ± 0.22 ml/h, or a total of 67 ± 16 ml/72 h of liquid within the water trap, containing 0.18 ± 0.21 mg of protein.

In the three sheep ventilated with ITPV for 24 h with Evans Blue dye added to the saline, we infused a total of 5.08 ± 0.11 mg Evans Blue dye into the RTC, and

Table 1. Frequency and Severity of Histologic Findings in the Trachea of Sheep Subjected to Conventional Volume-controlled Ventilation (n = 7) and ITPV (in Parentheses) (n = 7)

	Epithelial Damage					Submucosa			
	Absent	Mild	Moderate	Severe		Absent	Mild	Moderate	Severe
Tracheal epithelium					Inflammation				
A	0(0)	4(5)	2(2)	1(0)	A	0(0)	3(3)	4(4)	0(0)
B1	1(2)	6(5)	0(0)	0(0)	B1	0(3)	4(1)	3(3)	0(0)
B2	4(5)	3(2)	0(0)	0(0)	B2	0(2)	5(1)	2(4)	0(0)
C	7(5)	0(2)	0(0)	0(0)	C	1(2)	5(5)	1(0)	0(0)
D	7(7)	0(0)	0(0)	0(0)	D	1(3)	6(2)	0(2)	0(0)
E	7(7)	0(0)	0(0)	0(0)	E	1(3)	6(4)	0(0)	0(0)
Glandular epithelium					Edema				
A	0(0)	5(4)	2(3)	0(0)	A	0(0)	5(6)	2(1)	0(0)
B1	0(0)	4(3)	2(2)	1(2)	B1	1(1)	5(5)	1(1)	0(0)
B2	0(0)	2(4)	5(2)	1(2)	B2	0(0)	6(6)	1(1)	0(0)
C	0(0)	2(3)	5(4)	0(0)	C	0(0)	7(5)	0(2)	0(0)
D	0(0)	2(1)	4(5)	1(1)	D	0(0)	7(6)	0(1)	0(0)
E	0(0)	4(3)	3(3)	0(1)	E	0(0)	7(6)	0(1)	0(0)
Mucous accumulated in ducts					Congestion				
A	1(2)	5(4)	1(1)	0(0)	A	0(2)	6(5)	1(0)	0(0)
B1	1(2)	3(3)	3(2)	0(0)	B1	0(3)	6(4)	1(0)	0(0)
B2	0(1)	4(6)	3(0)	0(0)	B2	1(0)	5(7)	1(0)	0(0)
C	0(0)	3(2)	3(5)	1(0)	C	1(3)	6(4)	0(0)	0(0)
D	0(0)	3(2)	3(5)	1(0)	D	1(2)	6(5)	0(0)	0(0)
E	2(1)	4(3)	1(3)	0(0)	E	1(3)	6(3)	0(1)	0(0)
					Hemorrhage				
					A	7(7)	0(0)	0(0)	0(0)
					B1	7(7)	0(0)	0(0)	0(0)
					B2	7(7)	0(0)	0(0)	0(0)
					C	7(7)	0(0)	0(0)	0(0)
					D	7(6)	0(1)	0(0)	0(0)
					E	7(7)	0(0)	0(0)	0(0)

Numbers represent the total number of sheep within categories. The location within the trachea is indicated by row, while the extent of damage is listed by column (Absent, Mild, Moderate). The Mann–Whitney test was used to make comparisons between the results obtained for each type of alteration in each area of the trachea in control group, and ITPV group animals. There were no significant differences for any of the values tested.

ITPV = intratracheal pulmonary ventilation.

we collected 4.90 ± 0.10 mg Evans Blue dye within the water trap. This recovery equals 96–98% of the infused saline with added dye. The tracheal mucosa in these three sheep was lightly stained blue for a distance of 1 or 2 cm just distal to the tip of the ETT. No areas stained with Evans Blue dye were found in the lower parts of the trachea or in the lungs.

The frequency and severity of the changes observed on microscopic examination of the trachea are indicated in detail in table 1. Because some variations were found in the severity of the lesions listed in table 1, these changes were graded semiquantitatively and analyzed statistically. The Mann-Whitney test failed to disclose any statistically significant differences between the two groups of animals with respect to any of these

changes. Comparison of the values of the Reid indices showed a significant difference ($P < 0.05$) between the two groups only in area B_1, in which the submucosal glands were slightly larger in the ITPV group than in the control group (table 2).

Discussion

In this controlled study, we show a significant decrease in mucus accumulation within the ETTs in sheep ventilated for 3 days with the ITPV system, without suctioning. Visual inspection showed that the ETTs of the ITPV group were free of gross mucus deposits, whereas those of the control group contained variable

Table 2. Reid Index in Sheep Subjected to Volume-controlled Ventilation and Intratracheal Pulmonary Ventilation

Gland/Wall Ratio	VSV (n = 7)	ITPV (n = 7)
A	0.356 ± 0.094	0.351 ± 0.094
B_1*	0.395 ± 0.066	0.464 ± 0.045
B_2	0.443 ± 0.078	0.374 ± 0.094
C	0.353 ± 0.059	0.343 ± 0.126
D	0.340 ± 0.123	0.318 ± 0.077
E	0.232 ± 0.077	0.252 ± 0.247

Values are mean ± SD.

VCV = ITPV =

* Significant difference between the two groups ($P < 0.05$; Student's t test).

and often copious mucus deposits. The weight gain of the ETTs and the resistance to air flow through the ETTs were significantly lower in the ITPV group compared with the control group. The ETTs of the ITPV group sheep contained significantly less protein compared with the control sheep.

The weight of protein collected in the water trap of the expiratory limb of the ITPV group almost equaled the weight of protein found within the ETTs in the control group. The water trap in the control group contained only traces of protein. Thus we conclude that the mucus that might have accumulated in the ETTs of the ITPV group was actively removed and collected in the water trap. It seems likely that the mechanism for this is the same as seen in our previous *in vitro* studies‡; that is, the rapid gas flow and the saline droplets, as they emerged from the RTC, liquefied the secretions, which were then transported from the tip of the ETT in a cephalad direction and finally expelled from the ETT. This suggests the importance of continuous saline infusion.

The gross appearance of the trachea and the lungs were similar in both groups, with no sign of injury in either organ. This suggests that injury to the airways below the tip of the ETT during MV results primarily from insufficient humidification[9] and direct trauma from suctioning.[2] Humidification is a problem in all forms of ventilation that use high gas flows delivered from a source of high pressure, because relative humidity decreases when gas expands as it exits through a catheter. When gas flow is delivered at the distal end of the

‡Trawöger R, Kolobow T, Giacomini M, Cereda M: Mechanism of mucous clearance from endotracheal tubes during intratracheal pulmonary ventilation (abstract). Am J Respir Crit Care Med 1996; 153:A374

ETT, as in high-frequency jet ventilation,[10] tracheal gas insufflation,[11] or ITPV, possible injury to the airway epithelium is an important concern. In the first region of the trachea (above the cuff), both groups showed epithelial damage (mild in four sheep in the control group and in five sheep in the ITPV group, and severe in one sheep in the control group). In the second area (just distal to the tip of the ETT), damage was mild, and it was practically absent in areas three to five in both groups. This suggests that ITPV with saline infused into the RTC is safe, because the gas remains well humidified, and there is no need for suctioning. Sheep in both groups were managed at comparable RR, tidal volume, F_{IO_2}, airway pressures, and medications. No sheep showed evidence of systemic or respiratory infection. We assume that mucus production was similar in both groups of sheep. This is also suggested by the similar Reid indices in the two groups of sheep. No abnormal accumulations of mucus developed within the tracheas of any of the animals. These results suggest that mucociliary transport remained reasonably intact in all animals.

Cuffed ETTs, as in this study, have been shown to impair mucociliary transport for several (24) hours. We used low cuff inflation pressures, and thus mucociliary transport may have recovered during our studies. By using the RTC with continuous infusion of saline, we maintained clean airways.

Suctioning can severely traumatize airway epithelium.[2] Improved catheter design and intermittent application of vacuum have been suggested as a way to minimize mucosal damage.[12,13] Catheters with multiple radial side holes reportedly cause less tissue injury compared with those with one side hole.[2] Mucosal damage appears to be cumulative and is more pronounced at higher applied vacuum pressure.[2] Catheter contact alone can also lead to loss of cilia, and to exfoliation of airway epithelium.[14,15] Many investigators now recommend that suction catheters not be passed beyond the tip of the ETT to avoid mucosal injury.[16] We believe the ITPV system with the RTC catheter can help prevent tracheal injury and eliminate adverse effects from endotracheal suctioning.

It is common practice to instill 5–10 ml saline into the ETT before suctioning to liquefy inspissated secretions and to facilitate removal of mucous during suctioning. However, enhanced mucus removal after saline instillation may result from coughing provoked by the saline.[17] Oxygenation commonly decreases after saline instillation.[18] Bacteria can be dislodged from the ETT

and spread to the lower airways during tracheal suctioning.[5] In one study, only 10.7–18.7% of the instilled saline was retrieved on suctioning.§ This contrasts with total recovery of all saline infused during ITPV without suctioning.

A suction system consisting of simultaneous instillation of 40 ml normal saline and suctioning with a vacuum of 180 cm H_2O through a double-lumen catheter was shown to be more effective than conventional suctioning for removing secretions and retrieving instilled saline.[19] In some respects, that system is similar to the ITPV system in which saline is simultaneously instilled and then removed. However, the former system provides neither continuous removal nor continuous dilution of secretions. This may result in accumulation of secretions between suctioning.

Our study has certain limitations. We used healthy sheep, and there was relatively little accumulation of airway secretions, even in the control sheep. We found no major complications, such as blockage of ETTs or severe atelectasis. We believe this reflects adequate humidification in both groups of sheep. It remains to be shown whether ITPV can remove the copious and often purulent secretions frequently encountered in patients, although observations from our earlier studies in a model of severe acute respiratory failure shows it to be equally effective (unpublished observations). Finally, we cannot extrapolate our results beyond the 3-day duration of the study.

We conclude that without sectioning, the ETTs in sheep were kept free of airway secretions during 3 days of MV with ITPV using the RTC catheter, when 5 ml/h normal saline is continuously infused into the RTC. This method could become an attractive alternative to airway suctioning in the general patient population, particularly in those with head injuries, and in premature neonates.

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