samples were deproteinized with 5% perchloric acid and centrifuged for 10 min at 1000 × g. A total of 2 mg (base) terbutaline sulfate (Astra) dissolved in distilled water was applied topically in 2 50-µl doses given unilaterally 2 min apart. IOP was measured and aqueous humor withdrawn 1 h later. Student t-tests discriminated levels of probability.

Results. Aqueous humor taurine levels were found to be significantly higher than plasma taurine levels in anesthetized rabbits ($p < 0.01$) (table 1). 2 mg (base) terbutaline significantly reduced IOP ($p < 0.001$) 1 h after topical instillation in treated eyes, but not fellow eyes (table 2). Terbutaline failed, however to alter aqueous humor taurine levels in treated or fellow eyes ($p > 0.05$) (table 2).

Discussion. Our results indicate that, like most of the other amino acids[7], taurine exists in significantly higher concentrations in the aqueous humor than in the plasma of anesthetized rabbits. In general, amino acid levels have been found to be lower in the aqueous humor than in plasma of non-human primates, cats and rats[6]. Amino acid ratios of dogs slightly favor the aqueous humor[11]. Eyes of sheep[12], bovines[13], rabbits[7] and diseased eyes of humans[14–17] all show generally greater amino acid concentrations in the aqueous humor. However, of the species studied, rabbits alone have significantly higher aqueous than plasma taurine levels, which may imply the involvement of an active transport mechanism. In this study no correlation between an adrenergically mediated decrease in IOP and aqueous taurine content was found. In another study[18] however, while topical taurine had no effect on normal IOP in rabbits, subconjunctival injections of taurine apparently reduced the hypertensive effect of a subsequent injection of prostaglandins (PGE_2 and $PGF_{2\alpha}$).

1 Supported by National Eye Institute grants Nos EY05430 (JMR) and EY02156 (DEP).
2 Acknowledgments. We thank Dr J. Gintautas, this institution, for translating reference 18 and Dr S.Y. Schmidt, Massachusetts Eye and Ear Infirmary, for valuable advice.
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Activity of bulbar respiratory modulated neurons and restart of respiration after hypocapnic apnea in rabbits

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Summary. The activity of respiratory modulated neurons at the end of the apneic pause and during restart of respiration and the diaphragmatic mass activity were examined and both were compared to quiet respiration. Thresholds of mutual inhibition of neurons are unevenly distributed within various phase types of neurons.

In states of hypocapnic apnea following artificial hyperventilation, part of the respiratory modulated neurons (RMN) become silent while others discharge tonically at a low rate. Both types of responses have been found in phase-bound inspiratory (I neurons) and expiratory units (E neurons), as well as in phase-spanning inspiratory-expiratory (IE neurons) and expiratory-inspiratory cells (EI neurons)[1–3]. As apnea regresses, the spike density (spd) of the tonically discharging RMN and the tonic diaphragmatic activity steadily increase.

The first phasic movement terminating apnea is expiratory[4]. In rabbits it was found that respiratory movements started again when Pa_{CO2} averaged 28.3 mm Hg (30.3 during normal respiration) and pH_{art} 7.34 (control value 7.31).

The present study describes the discharge of RMN at the time when respiration restarts. In rabbits anesthetized with 1.1–1.3 g urethane/kg b.wt, the medulla oblongata was sounded stereotaxically from 2 mm rostrally to 4 mm caudally of the promontorium gliosum and 3 mm laterally from the midline. Extracellular recordings were made from RMN located near the solitary tract and in the neighbourhood of the nucleus ambiguus with 'floating' 25-µm platinum wire electrodes (for location of RMN, see Fallert and Wassermeyer[5]). The tracheal pressure, the excursions of a bellows-type spirometer and the electrogram of the diaphragm were recorded. The strength of diaphragmatic mass activity (dma) was estimated from light extinction of the original recording measured within a time 'window' of 250 msec duration with a lux-meter (Gossen). Both cervical vagal nerves were intact. Following artificial hyperventilation with a positive-negative-pressure respirator during 5 min, apnea of 10–20 sec duration occurred. After release from inflation, in rabbits vagally mediated tonic inspiratory activity occurs, the duration of which, however, hardly exceeds 3 sec[4]. The apnea examined in the present study may thus well be attributed to hypocapnia. Bursting activity of 5 EI, 16 I, 6 IE and 6 E neurons during normal respiration is shown on the left side of figure 1, A and B. Units which fired during one respiratory half cycle and during less than 15% of the other half cycle were denoted as I or E neurons; the remainder cells were labelled as IE or EI units. α- and β-classification was based on unit response to lung distension and collapse. Decrease of burst duration

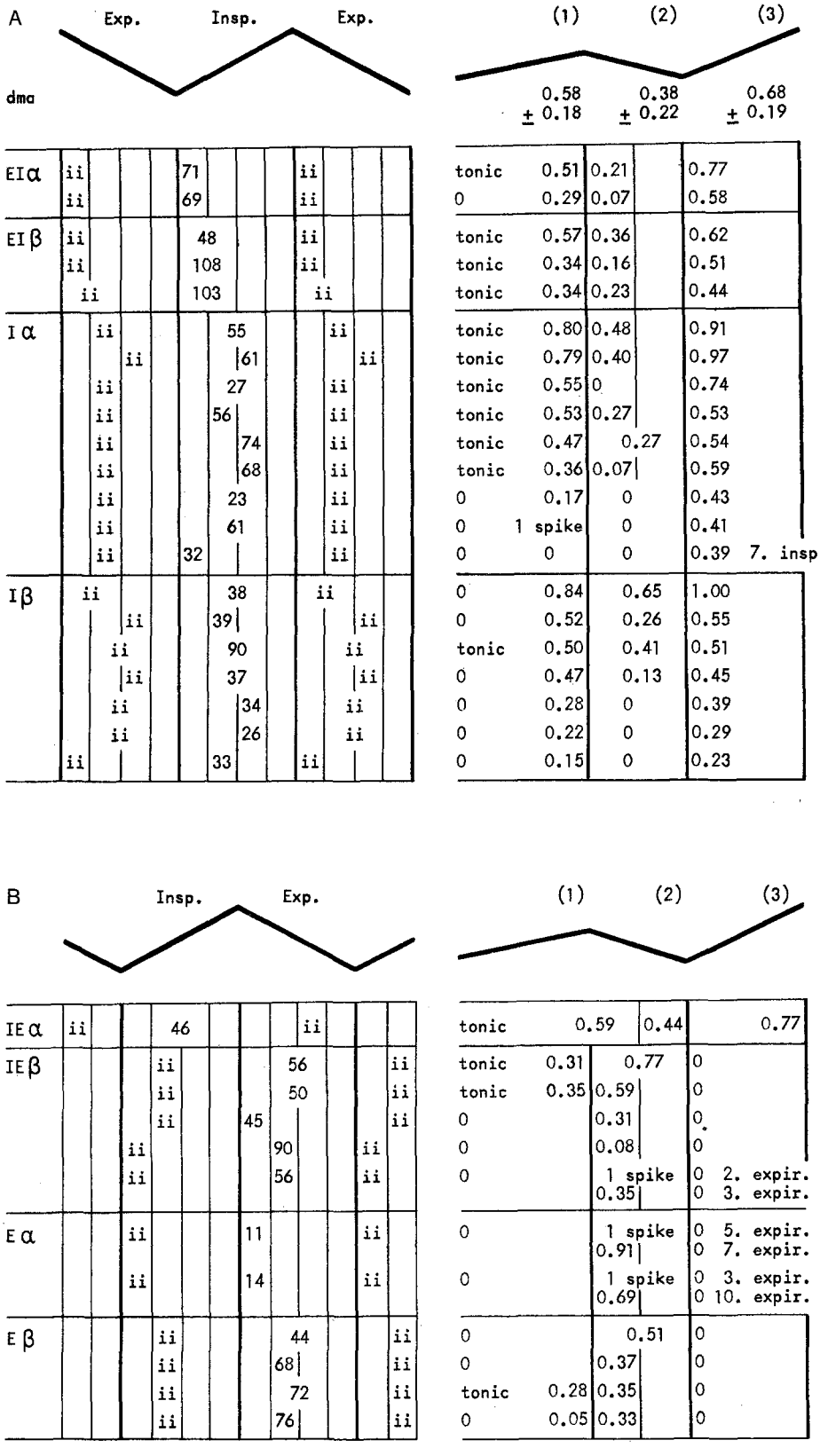

Fig. 1, A and B. On the left side bursting activity of 33 RMN during quiet respiration is shown. On top the lung volume is drawn schematically. Inspiration and expiration are divided into 4 equal time intervals. Values of spd are given at their actual time incidence within the cycle; ii indicates the middle of the interburst intervals. On the right side the end of the apneic pause (1), the 1st expiration (2) and the 1st inspiration (3) are shown. Values of dma are related to the strength of activity during quiet inspirations (mean ± SD; n = 31). 'Tonic' means continuous low-rate firing, 'O' stands for arrest of spike generation during phase (1). Values of spd related to values during normal respiration are given. EI and I units are ordered from high to low relative spd at the end of the apneic pause. In almost the same order of neurons during the first inspiration relative spd decreased from top to bottom of the list.

and/or spd during artificial lung distension[6] and increase during lung collapse was denoted as an α-type response and vice versa for a β-type response. The spd was measured within a time 'window' of 250 msec duration at the time of shortest interspike intervals. On the right side of figure 1, A and B, the end of the apneic pause (1), the 1st expiration (2) and the 1st inspiration (3) are shown. Many EI and about half of the I neurons discharged tonically during apnea. At the end of the apneic pause their spd ranged from about $^4/_5$ of spd during quiet respiration to almost zero; dma had reached about $^3/_5$ of that of normal inspirations. In many EI and I units which normally discharged at a high spd, inhibition during the 1st expiration was only partial (figure 2). During the 1st inspiration spd was still below control values and gradually increased to normal values from breath to breath. In some tonically discharging RMN and in some units which were silent after respirator arrest and which started to generate spikes towards the end of the apneic pause, during phase (1) minute oscillations of spd occurred with a period which was somewhat inferior to that of normal respiration. The phenomenon was observed in 2 EIα neurons, 1 EIβ unit (figure 2), 3 Iα cells, 2 Iβ neurons, 2 IEβ units and 1 Eβ cell (figure 2). Rhythmic activity of I and E neurons before the onset of respiration has been described[7]. Many E neurons were silent throughout the apneic pause (figure 1, B); onset of spike discharge coincided with the 1st expiration or, in 1 IEβ and 2 Eα neurons, with the 2–10th expiration only, and initially relative spd was low. Onset of discharge of some E neurons several cycles after resumption of spontaneous breathing confirms the findings of previous investigations[7]. In IE and E units which were active towards the end of the apneic pause spd sharply rose during the 1st expiration up to about $^3/_{10}$–$^6/_{10}$ of values during quiet respiration; spd and discharge patterns were gradually normalized from breath to breath (figure 2). If inhibition of I neurons by E units takes place[6,8], inhibition of I (and EI) neuron discharge terminating apnea comes into operation at a relatively low level of Eβ (and IEβ) neuron excitation. Respiration then restarts and Eα units become active only after a couple of cycles have been

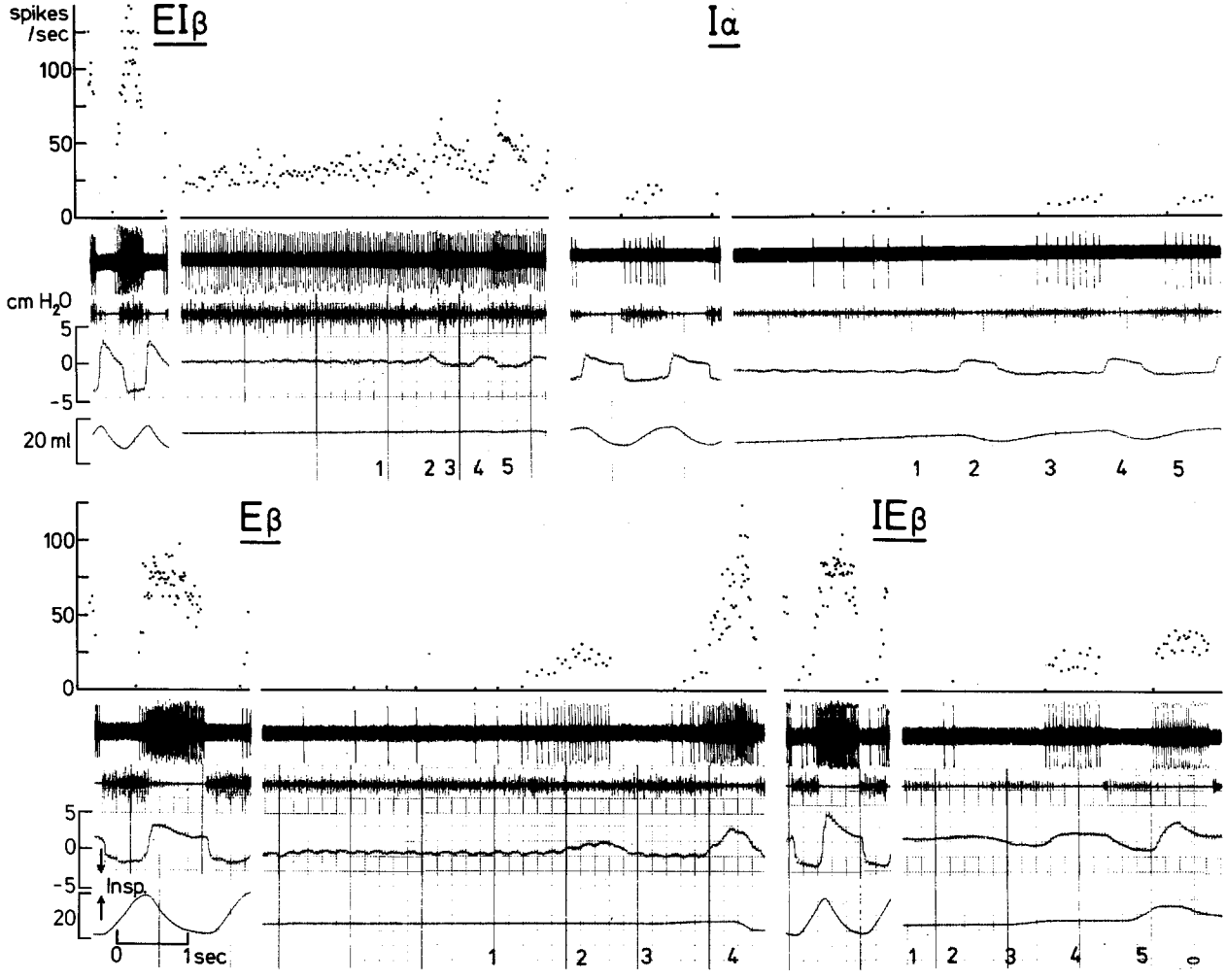

Fig. 2. Paired recordings illustrating end of the apneic pause (1) and restart of breathing in four RMN on right side; (2) and (4) are 1st and 2nd expiratory, (3) and (5) 1st and 2nd inspiratory movement. Normal respiration is shown on the left side. From top to bottom, the traces are: unretouched unit recording with single interspike intervals plotted as instantaneous spd; electrogram of the diaphragm; tracheal pressure; spirogram. The EIβ neuron exhibited steadily increasing tonic firing throughout phase (1); from breath to breath spd became gradually lower during expirations but higher during inspirations. The Iα neuron was silent throughout phase (1); some spikes were generated towards the end of the apneic pause. Bursting inspiratory activity was gradually restored from breath to breath. The Eβ neuron generated some spikes towards the end of phase (1). A fairly sharp increase of spd coincided with the 1st expiration. Discharge was still inspiratory-expiratory during the 2nd respiratory movement. The IEβ neuron was silent throughout the apneic pause. The initially expiratory discharge gradually turned to a normal phase-spanning pattern from breath to breath.

generated. When considering that Eα discharge occurs in 2% of all RMN only, it is doubtful if these neurons are indispensable for bulbar rhythmogenesis. EI neurons are supposed to activate I units[10], I cells to excite IE neurons[9,11] and IE units to facilitate E cells[9]; if this mechanism is true, a relatively low level of EI and I neuron excitation is sufficient to initiate oscillation. The excitation level of EIα and β and Iα and β neurons at the end of the apneic pause covered a relatively wide range; the range of dma was in a comparable order of magnitude. Most Iβ units, however, were silent immediately after respirator arrest. In some RMN, rhythmic modulation of spd starts well before oscillation of the entire neuronal network, i.e. before the 1st expiration occurs; this is probably due to unequal thresholds for mutual inhibition within each phase type of RMN.

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Cardiac output and regional blood flow studies in golden hamsters

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Summary. A method is described for the acute catheterization of the left ventricle of the heart and descending aorta combined with the use of the radioactive microsphere technique to study hemodynamic parameters in anaesthetized hamsters. The hemodynamic parameters studied include mean blood pressure, cardiac output, percentage distribution of cardiac output and regional blood flows in different organs.

Recently the hamster has been used as a model animal for the study of renal hypertension[1], microcirculation[2] and thrombosis[3]. Scanty data however, are available on hemodynamic aspects of this species. We attempted to study cardiac output, its distribution and regional blood flows in different organs using the modified radioactive microsphere technique of Malik et al.[4] in anaesthetized hamsters.

Materials and methods. 10 golden hamsters weighing between 90 and 110 (99 ± 3) g were used. They were anaesthetized with pentobarbitone 80 mg/kg i.p. A median incision was made on the ventral surface of the neck, tracheotomy was performed for free ventilation, and the right carotid artery was isolated from its position beside the trachea. The vessel was catheterized with PE-50 polythene tubing (inner diameter 0.58 mm and outer diameter 0.97 mm) connected to PE-10 polythene tubing (inner diameter 0.28 mm and outer diameter 0.61 mm). The catheter was filled with physiological saline containing heparin 50 IU/ml and attached at its proximal end (PE-50) to a pressure transducer (HP-1280C), which in turn was connected to a Hewlett packard multichannel recorder. The end to be inserted into the artery (PE-10) was marked approximately 3 cm from the tip to indicate when a sufficient length of the catheter had been inserted. A small incision was then made (under a microscope, magnification × 16) in the wall of the artery, and the catheter was eased down in the artery, as the recording of mean blood pressure was being monitored. At the point at which the catheter entered the left ventricle, the end diastolic pressure dropped to zero, whereas systolic pressure remained unchanged (figure). The regular pulse indicated that the catheter did not touch the internal wall of the ventricle. The catheter was then tied firmly in position by 2-3 ligatures placed around the catheter in the carotid artery to secure it. The exact position of the catheter in the left ventricle was verified at autopsy. The aortic catheter was inserted in the descending aorta about 5 mm above the bifurcation, through a median incision made on the ventral abdominal surface of the body. The other end of the aortic catheter (PE-50) was connected to a precalibrated withdrawal pump (B. Braun Melsungen AG), for collection of a timed reference blood sample.

Chromium-51 (Cr^{51}) labelled microspheres (15 µm in diameter) suspended in physiological saline (NEN Chemicals GmbH) were used. 0.1 ml of a suspension of microspheres (containing approximately 4×10^4 microspheres and a total radioactivity of $9.3 \times 10^4 \pm 0.5 \times 10^4$ cpm) was injected into the left ventricle of the heart. The total radioactivity injected into the system was determined by calculating the difference between the radioactivity taken in to the syringe at the start and that retained in the syringe, needle and catheter after the injection of the microspheres. Microspheres were injected over a 30-sec period, while the reference sample was being withdrawn from the aortic catheter beginning 10 sec before the start of the microsphere injection and continuing for more than 1 min. The blood was withdrawn into a heparinized syringe at the rate of 0.187 ml/min. At the end of the experiment, the animals were sacrificed using a large dose (500 mg/kg) of pentobar-

Percentage distribution of cardiac output and absolute blood flow in different organs in anaesthetized hamsters

Organ	Percentage of cardiac output per organ	Blood flow (ml/min/g)
Heart (ventricles)	17.62 ±2.75	10.40 ±1.31
Lungs	0.94 ±0.19	0.29 ±0.08
Liver (arterial)	0.46 ±0.15	0.035±0.003
Spleen	0.57 ±0.12	1.36 ±0.33
Right kidney	9.44 ±0.68	3.91 ±0.50
Left kidney	9.94 ±0.94	3.90 ±0.48
Front paws	0.53 ±0.11	0.079±0.012
Right cheek pouch	0.073±0.012	0.039±0.007
Left cheek pouch	0.091±0.017	0.047±0.013

Values are represented as mean ± SEM (n = 10).