Gating of Na Channels in the Rat Cortical Collecting Tubule: Effects of Voltage and Membrane Stretch

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ABSTRACT The gating kinetics of apical membrane Na channels in the rat cortical collecting tubule were assessed in cell-attached and inside-out excised patches from split-open tubules using the patch-clamp technique. In patches containing a single channel the open probability (Po) was variable, ranging from 0.05 to 0.9. The average Po was 0.5. However, the individual values were not distributed normally, but were mainly <0.25 or >0.75. Mean open times and mean closed times were correlated directly and inversely, respectively, with Po. In patches where a sufficient number of events could be recorded, two time constants were required to describe the open-time and closed-time distributions. In most patches in which basal Po was < 0.3 the channels could be activated by hyperpolarization of the apical membrane. In five such patches containing a single channel hyperpolarization by 40 mV increased Po by 10-fold, from 0.055 ± 0.023 to 0.58 ± 0.07. This change reflected an increase in the mean open time of the channels from 52 ± 17 to 494 ± 175 ms and a decrease in the mean closed time from 1,940 ± 350 to 336 ± 100 ms. These responses, however, could not be described by a simple voltage dependence of the opening and closing rates. In many cases significant delays in both the activation by hyperpolarization and deactivation by depolarization were observed. These delays ranged from several seconds to several tens of seconds. Similar effects of voltage were seen in cell-attached and excised patches, arguing against a voltage-dependent chemical modification of the channel, such as phosphorylation. Rather, the channels appeared to switch between gating modes. These switches could be spontaneous but were strongly influenced by changes in membrane voltage. Voltage dependence of channel gating was also observed under whole-cell clamp conditions. To see if mechanical perturbations could also influence channel kinetics or gating mode, negative pressures of 10-60 mm Hg were applied to the patch pipette. In most cases (15 out of 22), this maneuver had no significant effect on channel behavior. In 6 out of 22 patches, however, there was a rapid and reversible increase in Po when the pressure was applied. In one patch, there was a reversible decrease. While no consistent effects of pressure could be documented, membrane deformation could contribute to the variation in Po under some conditions.

INTRODUCTION

Epithelial Na channels in such tissues as the rat cortical collecting tubule (CCT), toad urinary bladder and in amphibian cells (A6) in culture are characterized by a high selectivity for Na over K, single-channel conductance of 4-5 pS at room temperature (8-9 pS at 37°C) and long mean open and closed times (Eaton and Hamilton, 1988; Garty and Benos, 1988; Smith and Benos, 1991; Palmer, 1992). The open probability of these channels is, however, quite variable. In the rat CCT under apparently identical physiological conditions Po can range from very low levels (<0.05) to very high values (>0.95) (Palmer and Frindt, 1988; Pacha et al., 1993). In some cases, two channels in the same patch can apparently have different values of Po (Palmer and Frindt, 1988; Marunaka et al., 1992). Variables that have been proposed to affect Po include intracellular pH (Palmer and Frindt, 1987), intracellular Ca (Silver et al., 1993), activation of PKC (Ling and Eaton, 1989), activation of G-proteins (Ohara et al., 1993), insulin, (Marunaka et al., 1992) and aldosterone (Kemendy et al., 1992).

Another factor that can influence Po is the membrane voltage. In contrast to the effects of voltage on Na channels from excitable cells, hyperpolarization of
the cell membrane activates epithelial Na channels while depolarization deactivates them (Palmer and Frindt, 1988; Frindt et al., 1993). The response to voltage was weak compared with classical voltage-gated channels, with \( P_o \) increasing by \( \sim 2\% \) per mV (Palmer and Frindt, 1988). The voltage dependence of the channels may be important in mediating feedback control of channel activity, especially during changes in the rate of Na entry into the cells (Frindt et al., 1993). These effects have not been studied in detail.

Recently epithelial Na channels have been cloned from the rat colon (Canessa et al., 1993; Lingueglia et al., 1993; Canessa et al., 1994). These clones called \( \alpha, \beta, \) and \( \gamma \) ENaC’s, show no significant homology to other known vertebrate ion channels. They do share, however, homology of both nucleotide and protein sequences as well as similarity of apparent membrane topology with several genes from the nematode Caenorhabditis elegans which are expressed in sensory neurons and are thought to be involved in the transduction of mechanical stimuli (Driscol and Chalfie, 1991; Chalfie et al., 1993; Huang and Chalfie, 1994). This raises the possibility that the epithelial Na channels are related to mechanosensory channels. The sensitivity of the Na channels to mechanical perturbations of the membrane, e.g., membrane stretch, has not yet been examined.

In this paper we describe the kinetic properties of the epithelial Na channel from the rat renal cortical collecting tubule, and the effects of changes in membrane voltage and transmembrane pressures on channel gating.

**MATERIALS AND METHODS**

**Biological Preparations**

Sprague-Dawley rats of either sex (100-150 g) raised free of viral infections (Charles River Laboratories, Kingston, NY) were fed a low Na diet for at least one week to enhance Na channel activity. Animals were killed by cervical dislocation, the kidneys removed, and the CCT dissected free and opened manually to expose the luminal surface. The split tubules were attached to a small plastic rectangle coated with Cell-Tak (Collaborative Research, Bedford, MA) and placed in a perfusion chamber mounted on an inverted microscope. The chamber was continuously perfused with solution which was prewarmed to 37°C by a miniature water-jacketed glass coil (Radnoti Glass Technology, Monrovia, CA). Principal cells of the tubule were identified visually as described previously (Pacha et al., 1991). To apply pressure across the patch, suction was applied to the back of the pipette through the pipette holder using a micrometer syringe. The pressure was monitored using a pressure transducer (model DPM1-B; Bio-Tek).

**Solutions**

Tubules were superfused with a solution consisting of (millimolar): 140 NaCl, 5 KCl, 2 CaCl\(_2\), 1 MgCl\(_2\), 2 glucose, and 10 N-2-hydroxyethylpiperazine-N'-2-ethanesulfonic acid (Hepes) adjusted to pH 7.4 with NaOH. Modifications of the solution are described in the text. The patch-clamp pipettes were filled with solutions containing (millimolar): 140 LiCl, 5 MgCl\(_2\), and 10 Hepes, adjusted to pH 7.5 with LiOH. Li was used as the primary cation in the pipette because the single-channel conductance is higher to Li than to Na, giving a better signal-to-noise ratio (Silver et al., 1993).

For whole-cell recordings the tubules were bathed with a solution in which NaCl was replaced by Na methane sulfonate to reduce membrane Cl\(-\) conductance. The pipette solution contained (millimolar): 7 KCl, 123 aspartic acid, 20 CsOH, 20 TEA\(_2\), 5 EGTA, 10 Hepes, 3 MgATP, and 0.3 NaGTP with the pH adjusted to 7.4 with KOH.

**Electrical**

Basic patch-clamp methods were as described previously (Frindt et al., 1993; Silver et al., 1993). Recording of currents and analysis of data were carried out with an Atari 1040 ST computer equipped with interface and data acquisition software (Instrutech, Mineola, NY). Current records were stored on video tape using a pulse-code modulator (Instrutech). Computation of the mean number of open channels (\( N_{Po} \)), open probability (\( P_o \)) and mean open and closed times was carried out using the TAC program (Instrutech). In patches containing a single channel the open- and closed-time distributions were plotted and analyzed according to the method of Sigworth and Sine (Sigworth and Sine, 1987). Fitting of the distributions was carried out using the maximum likelihood method with the TAC program.

**RESULTS**

**Kinetics of Na Channels in Single-Channel Patches**

It is rare to find single Na channels in patches from the rat CCT. When the rat is on a low-Na diet, most patches contain many channels, with the average around five (Pacha et al., 1993). When the animal is on a normal diet, there is almost no Na channel activity at all. Intermediate conditions have been difficult to establish. Nevertheless over a period of about four years a reasonable number of patches which did contain just one active channel were observed. In such patches, only two current levels, corresponding to the open and closed state of the channels, were observed over a period of 3 min or more. These channels had conductances (12-13 pS) similar to those reported previously for the same conditions (Frindt et al., 1993). This value is higher than the 5 pS value associated with this channel (Palmer, 1992) because Li rather than Na was the major permeant ion and the measurements were made at 37°C rather than room temperature. The kinetics of these channels are described in Figs. 1-3. A striking finding was the variability in the mean open and closed times and the open probability within this group.

Fig. 1 A shows an example of a channel with a low \( P_o \). The activity of the channel was recorded for 11.5 min. The \( P_o \) which averaged 0.095, varied somewhat over this time period but there was no systematic change...
(Fig. 1 B). It was necessary to establish that the \( P_o \) was constant to use the entire record for assessing the kinetic properties of the channel. The time interval was selected arbitrarily. The mean open time was 77 ms, and the mean closed time was 760 ms. Open and closed-time distributions are shown in Fig. 1, C and D. The open-time histogram could be fitted by two exponentials, as described in the figure legend. The closed-time histogram could be described fairly well with a single exponential, although a better fit was obtained with two exponentials. Five out of six of the channels within this group with \( P_o < 0.22 \) required two exponentials to fit the open-time distribution. In all of these cases the closed times could be reasonably well described with a single exponential.

Fig. 2 A shows a record from a patch with an intermediate \( P_o \). The total recording time was 7 min and \( P_o \) was stable with an average value of 0.55 (Fig. 2 B). The mean open time was 350 ms. As in the case above, the open-time distribution indicated two time constants (Fig. 2 C). The mean closed time was 286 ms and was fairly well described by a single exponential (Fig. 2 D). There were only three channels in the group with intermediate \( P_o \) values between 0.25 and 0.75. All indicated two open times. One of the three also required two time constants to fit the closed-time distributions. In this patch the pipette potential was zero, compared with +20 mV for Figs. 1 and 3. No comparable record at +20 mV was available for this group.

Fig. 3 A shows a record from a patch with a single channel with a high \( P_o \). This recording lasted for 10 min. The average \( P_o \) was 0.87 and again was relatively constant over the recording period (Fig. 3 B). The mean open time was 1.62 s. The open times were best described by two exponentials, although most (88%) of the events were within the distribution of longer openings (mean open time 1.81 s). The mean closed time was 237 ms. In this case the closed time histogram indicated the presence of two closed time constants of 340 and 41 ms.

The relationship between \( P_o \) and open and closed times is shown in Fig. 4. In this and subsequent figures

**Figure 1.** Kinetics of a channel with low \( P_o \) (0.093). (A) Portion of the original recording which was 695 s long. The pipette potential was +20 mV. (B) Plot of \( P_o \) as a function of time. (C) Open-time histogram. The kinetics are plotted according to the method of Sigworth and Sine (1987). The solid line represents a fit to two exponentials with time constants 123 ms (51% of total events) and 24 ms (49%). (D) Closed-time histogram. The solid line represents a fit to two exponentials with time constants 1,580 (27%) and 454 ms (73%).
Figure 2. Kinetics of a channel with intermediate $P_o$ (0.55). (A) Portion of the original recording that was 399 s long. The pipette potential was 0 mV. (B) Plot of $P_o$ as a function of time. (C) Open-time histogram. The kinetics are plotted according to the method of Sigworth and Sine (1987). The solid line represents a fit to two exponentials with time constants 761 ms (37% of total events) and 95 ms (63%). (D) Closed-time histogram. The solid line represents a fit to two exponentials with time constants 761 ms (37%) and 171 ms (63%).

In previous studies using mainly multichannel patches the mean values of $P_o$ were given as 0.4–0.5 (Palmer and Frindt, 1988; Pficha et al., 1993). Curiously, there were very few single channels which had a $P_o$ in this range. It is possible that the criterion of having a single channel selects for conditions with extreme values of $P_o$. Another possibility is that multichannel patches contain two populations of channels with high and low $P_o$ which tend to average. Indeed the average $P_o$ of the 19 single-channel patches was 0.50 ± 0.08. This suggests the presence of at least two gating “modes” for the channels, one with high $P_o$ and one with low $P_o$. Multichannel patches may then have a mix of these modes.

Effects of Voltage on Channel Gating

Further evidence for gating modes, and for interconversion between them, was obtained from observations of the effects of voltage on channel gating. We have previously reported that hyperpolarization can activate the epithelial Na channel (Palmer, 1988; Frindt, 1993). In the present study we examined this effect in more detail using patches in which the initial $P_o$ was low, such
as that shown in Fig. 1. Most (>80%) channels in this group could be activated by voltage, although the extent of the hyperpolarization required for activation was variable. The time course of the effect of voltage for five such patches is shown in Fig. 5. Three of these had channels with a low $P_o$ at the resting potential of the cell ($V_{pipette} = 0$) and activated after a hyperpolarization of 40 mV. The other two had channels which maintained a low $P_o$ for more than 1 min at $V_{pipette} = 40$ mV, but activated with a further 40 mV hyperpolarization. There were no consistent differences in the two data sets which are therefore combined. $NP_o$ increased within 30 s and continued to rise, on the average, for 3 min. We did not study more prolonged hyperpolarizations. Upon returning the patches to the initial level $NP_o$ decreased but remained above the original level for 1–2 min.

The kinetics of the channels before and after voltage activation are shown in Fig. 6. This summarizes experiments similar to those shown in Fig. 5, but where there was a single active channel in the patch. The initial voltage was 0 to 40 mV, and the patch was hyperpolarized by 40 to 80 mV. The increase in $P_o$ is shown in A, B and C show the increase in mean open times and the decrease in mean closed times, respectively. In these cases the recording time at any one voltage was not sufficient to resolve multiple open or closed states. Clearly, however, changes in both opening and closing rates contribute to the increased $P_o$.

As indicated in Fig. 5, the effects of voltage on channel behavior often involved substantial delays. An example of an individual record illustrating delayed activation is shown in Fig. 7. Here the pipette voltage was initially 0 mV and $NP_o$ was quite low. Hyperpolarization of the patch by 80 mV (changing $V_{pipette}$ to +80 mV) had little immediate effect on $P_o$ but after ~16 s the channel suddenly activated. The increased $NP_o$ was sustained for at least 20 s but was reversed by returning $V_{pipette}$ to 0. A time course of $NP_o$ for this patch is shown in Fig. 8. In this case the channel was activated three times in succession. In each case there was a significant delay between the hyperpolarization and the activation.
In other cases long delays in the deactivation of the channels by depolarization were observed. One striking example is shown in Fig. 9. Here again the channel activity was low but was increased by hyperpolarization. In this case the activation was sustained for at least two minutes despite the return of the voltage to its original value. $P_o$ eventually returned to low levels after a further depolarization of the membrane.

The delays in activation and deactivation are summarized in Fig. 10 for a series of patches in which the original $P_o$ was $<0.25$. This required a somewhat arbitrary definition of activation. We considered the channels to be activated if the relative value $NP_o$ increased by a factor of two and if the absolute value of $NP_o$ increased by at least 0.01. The second condition was necessary for channels with very low $P_o$ values where relatively small increases in activity could double the $NP_o$. The distributions show that $\sim 1/3$ of the patches activated quickly, i.e., within 5 s. A better resolution was not possible because measurement of $NP_o$ took a finite amount of time. In the other patches activation and deactivation involved delays of up to 30 s.

The possibility that the delayed activation of the channels by hyperpolarization might be due to a voltage-dependent biochemical reaction was tested using excised, inside-out patches. An example is shown in Fig. 11. The $P_o$ was initially stable at a low value. Hyperpolarization of the excised patch increased $P_o$ after a delay that was similar to that observed in cell-attached patches. Similar results were seen in two other inside-out patches. This does not rule out a voltage-dependent chemical reaction, but does imply that the factors involved in such an effect must be retained with the patch after excision.

The complicated, history-dependent effects of voltage on the kinetics of the channel make it difficult to define a precise relationship between open probability

![Figure 4](image-url)  
**Figure 4.** Plot of mean open and closed times for various values of $P_o$. Each pair of points represents a different patch. Three patches in which both the mean open and closed times were unusually short are circled.

![Figure 5](image-url)  
**Figure 5.** Activation of channels by hyperpolarization. Channel activity was measured for 1 min at $V_{pipette} = 0$ (three patches) or $+40$ mV (two patches), during which time $NP_o$ remained low. The voltage was then increased by 40 mV for 3 min and decreased to the initial level for an additional 3 min. $NP_o$ was monitored continuously. Values were averaged over 15 (initial period) or 30 s (final two periods) intervals, and are reported as means $\pm$ SEM for the five patches.

![Figure 6](image-url)  
**Figure 6.** Effect of voltage activation on $P_o$ (*left*), mean open time (*center*), and mean closed time (*right*). All the data were from cell-attached patches selected to have a single channel and an initial $P_o$ of $<0.3$. Straight lines connect measurements from the same patch before and after hyperpolarization. The initial $V_{pipette}$ ranged from 0 to $+100$ mV. The increase in $V_{pipette}$ ranged from 40 to 100 mV.
Figure 9. Time course of $P_o$ for a patch containing a single channel in which deactivation by depolarization was delayed. $V_{pipette}$ was originally +40 mV, and $P_o$ was low. Hyperpolarization to $V_{pipette} = +80$ mV activated the channel after a delay of $\sim 20$ s. $P_o$ remained high for $\sim 1$ min after returning the potential to its original level. Further depolarization to $V_{pipette} = 0$ mV resulted in the eventual deactivation of the channel.

and voltage at the single-channel level. Therefore, the effects of voltage on Na channels were further assessed using the whole-cell clamp configuration of the patch-clamp technique. Previous studies showed that principal cells in the rat CCT are not electrically coupled, so that individual cells within an intact epithelium can be clamped (Frindt et al., 1990). Cells were held at 0 mV and hyperpolarized to various voltages up to 100 mV for 10 s. The protocols were then repeated in the presence of 10 μM amiloride. A typical response is shown in Fig. 12 A. "Instantaneous" currents were measured after the decay of the capacitative transient, within a few milliseconds of the voltage change. The inward current then increased rapidly, over the course of several hundred milliseconds, and then more slowly during the next 5–10 s. Neither the rapid nor the slow rise were seen in the presence of amiloride. The average initial and steady state levels of amiloride-sensitive current are plotted vs voltage in Fig. 12 B. The difference in the two currents presumably represents the voltage-dependent activation of the channels. There was an 80% increase in current at $-100$ mV. Substantial, although somewhat smaller increases were seen in the physiological voltage range $\sim -60$ to $-80$ mV.

Effects of Stretch on Channel Gating

It has been suggested that the epithelial Na channel is related through evolution to mechanosensitive channels found in nematodes. We therefore examined whether mechanical perturbation could alter the gating kinetics or gating mode of the channels in the CCT. To test this possibility we used the protocol of applying suction to the patch-clamp pipette during recording from a cell-attached patch (Sachs, 1992). To optimize
the chances of seeing an effect of mechanical perturbation, patches with a low basal $P_o$ were selected. In most cases negative pressures of up to 80 mm Hg had no obvious effect on the channels (Fig. 13A). However, in six cases there was a substantial, reversible activation of the channels (Fig. 13B). One of these positive results is illustrated in Fig. 14. In this case the negative pressure was increased in three steps up to 24 mm Hg. The first step to 8 mm Hg had no significant effect on channel gating. The second step to 16 mm Hg elicited only a brief burst of channel openings. Increasing the pressure to 24 mm Hg brought about a prompt, sustained activation of the channels. The increase in $P_o$ was reversed after releasing the pressure. This deactivation was not immediate, being complete 20 s after the pressure changed. In the six responsive patches $P_o$ was increased by application of between 10 and 30 mm Hg. The mean value of $NP_o$ in these patches increased from 0.048 ± 0.04 to 0.22 ± 0.09. In one other patch (not shown), $P_o$ was reduced by negative pressure and increased again when the pressure was released.

**DISCUSSION**

**Gating Modes**

The main purpose of this study was to examine the variations in open probability of the epithelial Na channel

**Figure 10.** Histograms of the delay times for activation by hyperpolarization (A) and deactivation by depolarization (B).

**Figure 11.** Delayed activation in an excised, inside-out patch. The patch was originally held at $V_{pipet} = +100$ mV. $P_o$ increased upon hyperpolarization to $V_{pipet} = +140$ mV after a delay of ~30 s.

**Figure 12.** Effect of voltage under whole-cell clamp conditions. Cell voltages were initially held at 0 mV and then switched to negative values between −10 and −100 mV. (A) The response of an individual cell to a hyperpolarization to −100 mV. Traces from the same cell in the absence (top) and presence (bottom) of 10 µM amiloride are shown. Negative values represent inward current. (B) The amiloride-sensitive current as a function of voltage. Currents were measured just after the decay of the capacitative transient (initial values) and at the end of the voltage pulse (steady state values). All currents were normalized to the value at 0 mV, and are presented as means ± SEM of four cells.
and to identify some of the mechanisms underlying this variability. The extent of the variation is illustrated in Fig. 4. Basal values of $P_o$ in single-channel patches ranged from $<0.05$ to greater than 0.9. This variability confirms previous studies showing similar ranges of apparent $P_o$ in multichannel patches from the rat CCT (Palmer and Frindt, 1988; Pácha et al., 1993). A large variability of $P_o$ has also been reported for Na channels in A6 cells, a culture model derived from the amphibian distal tubule (Ling and Eaton, 1989; Ohara et al., 1993). What was somewhat surprising was the clustering of these $P_o$ values toward the low and high end of the spectrum.

The records with high and low $P_o$ appear to represent the same basic channel type. All of the channels reported in this study had similar single-channel conductances of 12–13 pS, and strongly positive reversal potentials. More importantly, at least some of the channels with low $P_o$ could be converted to a high $P_o$ by membrane hyperpolarization or stretch. This suggests that the kinetic variations represent different gating modes, rather than different channel molecules. However, the data do not rule out a contribution of structural heterogeneity of channels to the variation in kinetic properties.

A gating mode is defined here as a set of kinetic states (open and closed states) governing channel gating which can undergo a shift to a second set of kinetic states, or a second set of rate constants for transitions between states. This shift occurs on a time scale which is slow relative to the lifetimes of open and closed states within a mode. Other ion channels have been shown to undergo such changes in gating mode. Voltage-dependent Ca channels, for example, can undergo changes in gating patterns which are influenced by dihydropyridines (Hess et al., 1984). Voltage-dependent Na channels can exhibit spontaneous shifts in their $P_o$-$V$ relationship even after reconstitution in planar lipid bilayers (O’Connell, 1992).

Previous studies had indicated that the basic Na channel kinetics in the absence of amiloride could be described in terms of a single open state and a single closed state (Palmer and Frindt, 1986; Eaton and Marunaka, 1990). In the case of the CCT, this conclusion was inferred primarily from recordings from multichannel patches. The relatively long single-channel recordings shown here reveal a more complex kinetic pattern. The presence of two time constants in the open-time and closed-time histograms suggest the existence of at least two open and two closed states. It is not yet clear, however, if the channel has access to all of these states at the same time. It is possible, for instance, that the different open and closed time constants arise from relatively short, reversible shifts in gating mode. This would predict that the distribution of open and closed times of sufficiently short records should be describable with single exponentials. Unfortunately, recordings of $<5$ min do not contain enough events to rigorously test this prediction.

Voltage Activation

The gating of the channels is influenced by the membrane voltage. Hyperpolarization increased $P_o$ as described previously (Palmer and Frindt, 1988; Frindt et al., 1993). A more complete examination of the effect of voltage revealed that the effect on $P_o$ involved considerable delays. These delays are not accounted for by a simple two-state model of channel gating in which the rates of opening and closing involve movement of charge through the electric field of the membrane and are therefore directly influenced by voltage. In this case the effect of changing the transmembrane voltage on the rates of channel opening and closure would be instantaneous, and the rate of change in $P_o$ would be de-
determined by the sum of the new opening and closing rates. The observations are consistent with the idea that the channels have two or more gating modes, with hyperpolarization favoring the high $P_o$ mode and depolarization favoring the low $P_o$ mode. This mechanism can account for the long delays and the hysteresis effects seen when the voltage was changed.

The observation of shifts in channel kinetics in excised patches suggests that the channels themselves, or a closely associated membrane component, is responding to the transmembrane voltage. This does not rule out the possibility that other events, such as biochemical modification of the channel, might also influence the gating mode. In A6 cells, for example, aldosterone was shown to shift channels from a low $P_o$ to a high $P_o$ state (Kemendy et al., 1992). In contrast, the $P_o$ of channels in the rat CCT was not influenced by mineralocorticoid status (Pácha et al., 1993). Increases in cytoplasmic Ca$^{2+}$ (Silver et al., 1993) and activators of protein kinase C (Ling and Eaton, 1989) reduce channel activity, possibly by favoring the low $P_o$ state. Insulin, on the other hand, increased $P_o$ of Na channels (Marunaka et al., 1992). Activation of G-proteins can also have a strong effect on channel gating. Both $P_o$ and mean open time of Na channels in A6 cells were increased by GDP and decreased by GTP in excised patches (Ohara et al., 1993).

The voltage dependence of the channels could also be observed under whole-cell conditions. This indicates that the sensitivity of the channels to voltage is not an artifact of the mechanical isolation of the channels within the patch. These data showed that switching the voltage from 0 to $-100$ mV could activate the channels by 80%. Assuming that this activation involved only an increase in open probability, without a concomitant insertion of new channels into the membrane, it can be concluded that the average $P_o$ with a holding potential of zero was at most 0.56. Since the voltage activation showed no signs of saturation as the voltage was increased to $-100$ mV, the actual initial $P_o$ was probably considerably lower.

**Mechanical Activation**

Recent cloning of the epithelial Na channel from the rat colon (Canessa et al., 1993; Lingueglia et al., 1993; Canessa et al., 1994) indicated homology between the subunits of this channel and certain genes identified in the nematode *C. elegans* involved in touch-avoidance responses (Driscoll and Chalfie, 1991; Chalfie et al., 1993; Huang and Chalfle, 1994). The homology, together with the similarity of the predicted membrane topology of the two families, suggests that the *C. elegans* genes may encode a mechanosensitive ion channel. This raises the question of whether the epithelial Na channels may also be influenced by mechanical perturbations of the membrane. We were unable to elicit a consistent response of the channels in the CCT to changes in transmembrane pressure, although in a minority of cases channel activation was observed. The variability in the response could reflect differences in the mechanical deformations of the apical membrane within the tip of the pipette. Alternatively, the Na channels may not respond to pressure in the same way as do the well

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**FIGURE 14.** Original tracings from a patch which responded to negative pressure. $V_{pipette}$ was held constant at $+20$ mV. Activity in the absence of suction is shown above on the left. Application of 8 mm Hg did not activate the channel (above right). Increasing the negative pressure to 16 mm Hg elicited a brief burst of channel openings (second line). Increasing the negative pressure to 24 mm Hg resulted in a sustained increase in $P_o$ (third line). After release of the suction $P_o$ remained high for $\sim 10$ s and then decreased (bottom line). Times refer to duration of the applied suction at the beginning of the individual trace.
characterized stretch-sensitive channels (Sachs, 1992; Sackin, 1994). It is possible that the protocol used to test for mechanosensitivity was not the most appropriate one for these channels. It has been shown, for example, that cell swelling can activate Na channels in A6 cells (Wills et al., 1991). Whether this reflects a direct effect on the channels is not yet clear. Substantial delays have been observed between the change in cell volume and the increase in channel activity. As shown here, however, this does not necessarily preclude a direct influence on the gating mode.

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REFERENCES

Canessa, C. M., J.-D. Horisberger, and B. C. Rossier. 1993. Epithelial sodium channel related to proteins involved in neurodegeneration. Nature (Lond.). 361:467–470.

Canessa, C. M., L. Schild, G. Buell, B. Thorens, Y. Gautschi, J.-D. Horisberger, and B. C. Rossier. 1994. The amiloride-sensitive epithelial sodium channel is made of three homologous subunits. Nature (Lond.). 367:463–467.

Chalfie, M., M. Driscoll, and M. Huang. 1993. Degenerin similarities. Nature. 361:504.

Driscoll, M., and M. Chalfie. 1991. The mec-4 gene is a member of a family of Caenorhabditis elegans genes that can mutate to induce neuronal degeneration. Nature (Lond.). 349:588–593.

Eaton, D. C., and K. L. Hamilton. 1988. The amiloride-blockable sodium channel of epithelial tissue. In Ion Channels. T. Narashashi, editor. Plenum Publishing Corp., New York. 151–182.

Eaton, D. C., and Y. Marunaka. 1990. Ion channel fluctuations: “Noise” and single-channel measurements. Curr. Top. Membr. Trans. 37:61–113.

Fridt, G., H. Sackin, and L. G. Palmer. 1990. Whole-cell currents in rat cortical collecting tubule: low-Na diet increases amiloride-sensitive conductance. Am. J. Physiol. 258:F565–F567.

Fridt, G., R. B. Silver, E. E. Windhager, and L. G. Palmer. 1993. Feedback inhibition of Na channels in rat CCT. II. Effects of inhibition of Na entry. Am. J. Physiol. 264:F565–F574.

Garty, H., and D. J. Benos. 1988. Characteristics and regulatory mechanisms of the amiloride-blockable Na+ channel. Physiol. Rev. 68:399–373.

Hess, P., J. B. Lansman, and R. W. Tsien. 1984. Different modes of Ca channel gating behavior favored by dihydropyridine Ca agonists and antagonists. Nature (Lond.). 311:538–544.

Huang, M., and M. Chalfie. 1994. Gene interactions affecting mechanosensory transduction in Caenorhabditis elegans. Nature (Lond.). 367:467–470.

Kemendy, A. E., T. R. Kleyman, and D. C. Eaton. 1992. Aldosterone alters the open probability of amiloride-blockable sodium channels in A6 epithelia. Am. J. Physiol. 263:C825–C837.

Ling, B. N., and D. C. Eaton. 1989. Effects of luminal Na+ on single Na+ channels in A6 cells, a regulatory role for protein kinase C. Am. J. Physiol. 256:F1094–F1103.

Lingueglia, E., N. Voilley, R. Waldmann, M. Lazdunski, and P. Barbury. 1993. Expression cloning of an epithelial amiloride-sensitive Na+ channel. FEBS (Fed. Eur. Biochem. Soc.) Lett. 318:95–99.

Marunaka, Y., N. Hagiwara, and H. Tohda. 1992. Insulin activates single amiloride-blockable Na channels in a distal nephron cell line (A6). Am. J. Physiol. 263:F392–F400.

O’Connell, A. M. 1992. Modul gating behavior of batrachotoxin-modified sodium channels. Ph.D thesis. Cornell University Medical College, New York.

Ohara, A., H. Matsunaga, and D. C. Eaton. 1993. G protein activation inhibits amiloride-blockable highly selective sodium channels in A6 cells. Am. J. Physiol. 264:C352–C360.

Pachá, J., G. Fridt, H. Sackin, and L. G. Palmer. 1991. Apical maxi-K channels in intercalated cells of CCT. Am. J. Physiol. 261:F696–F705.

Pachá, J., G. Fridt, L. Antonian, R. Silver, and L. G. Palmer. 1993. Regulation of Na channels of the rat cortical collecting tubule by aldosterone. J. Gen. Physiol. 102:29–42.

Palmer, L. G. 1992. Epithelial Na channels: function and diversity. Annu. Rev. Physiol. 54:51–66.

Palmer, L. G., and G. Fridt. 1986. Amiloride-sensitive Na channels from the apical membrane of the rat cortical collecting tubule. Proc. Natl. Acad. Sci. USA. 83:2767–2770.

Palmer, L. G., and G. Fridt. 1987. Effects of cell Ca and pH on Na channels from rat cortical collecting tubule. Am. J. Physiol. 253: F335–F339.

Palmer, L. G., and G. Fridt. 1988. Conductance and gating of epithelial Na channels from rat cortical collecting tubules. Effects of luminal Na and Li. J. Gen. Physiol. 92:121–138.

Sachs, F. 1992. Stretch-sensitive ion channels: an update. In Sensory Transduction. D. P. Corey and S. D. Roper, editor. Rockefeller University Press, NY. 241–260.

Sackin, H. 1994. Stretch-activated ion channels. In Cellular and Molecular Physiology of Cell Volume Regulation. K. L. Strange, editor. CRC Press, Boca Raton, FL. 215–240.

Sigworth, F. J., and S. M. Sine. 1987. Data transformations for improved display and fitting of single-channel dwell-time histograms. Biophys. J. 52:1047–1054.

Silver, R. B., G. Fridt, E. E. Windhager, and L. G. Palmer. 1993. Feedback regulation of Na channels in rat CCT. I. Effects of inhibition of the Na pump. Am. J. Physiol. 264:F557–F564.

Smith, P. R., and D. J. Benos. 1991. Epithelial Na+ channels. Annu. Rev. Physiol. 53:509–530.

Wills, N. K., L. P. Millinoff, and W. E. Crowe. 1991. Na+ channel activity in cultured renal (A6) epithelium: regulation by solution osmolality. J. Membr. Biol. 121:79–90.