The patch-clamp technique was used to study the electrophysiological properties of single smooth muscle cells obtained from the human cystic artery. These cells contracted on exposure to high $K^+$ and had a mean resting potential of $-36.2 \pm 7$ mV. Under current clamp, regenerative responses could not be elicited when depolarizing pulses were applied. Voltage-clamp measurements demonstrated that a large fraction of the outward current was inhibited by tetraethylammonium (5–10 mM) or $Ca^{2+}$ channel blockers and that it was enhanced by increasing $[Ca^{2+}]_o$, suggesting that it is a $Ca^{2+}$-activated $K^+$ current. In addition, spontaneous transient outward currents that were sensitive to extracellular $Ca^{2+}$ were observed in some cells. In cell-attached patch-clamp recordings, $Ca^{2+}$-activated $K^+$ channels that had a conductance of 117 pS were consistently identified. At negative potentials (approximately $-60$ mV), these single-channel events deactivated completely and very quickly, suggesting that they do not control the resting membrane potential in healthy cystic artery cells. $Ca^{2+}$ currents that were recorded using Ba$^{2+}$ (10 mM) as the charge carrier were enhanced by the dihydropyridine agonist, Bay K 8644, and blocked by nifedipine (0.1 $\mu$M). Only one type of $Ca^{2+}$ current, the L-type, could be identified in these cells. These results demonstrate that the major ionic currents in the human cystic artery are similar to other mammalian arteries and indicate that this tissue will be a useful model for studying the metabolic and pharmacological modulation of ionic currents in human vascular smooth muscle. (Circulation Research 1992;70:536–545)

**KEY WORDS** • voltage clamp • smooth muscle • humans • vascular smooth muscle • potassium current • calcium current

### Materials and Methods

**Cell Dispersion**

Cystic artery tissue was obtained from the gallbladders of patients admitted for elective cholecystectomy. Written consent was obtained from each patient before surgery. Tissues were obtained from 17 patients having a mean age of 51 (range, 23–87) years and an average blood pressure of 130/76 mm Hg. Thirteen of these patients were female.

Each excised artery was immediately placed in modified low $Ca^{2+}$ Krebs-Henseleit solution containing (mM) NaCl 115.3, KCl 4.6, CaCl$_2$ 0.08, MgSO$_4$ 1.1, NaHCO$_3$ 22.1, and glucose 7.8 and was equilibrated with 95% O$_2$–5% CO$_2$. It was then transported to the laboratory (within 15 minutes), where excess fat and connective tissue were trimmed away. Thereafter, the segment of artery was cut into small pieces (~0.5 x 0.5 cm), which were incubated at 35°C for 30 minutes in a low $Ca^{2+}$ solution containing collagenase (0.25 mg/ml), elastase (0.02 mg/ml), and trypsin inhibitor (0.5 mg/ml). The tissues were placed into a second enzyme-containing solution consisting of collagenase (0.25 mg/ml) and bovine serum albumin (1 mg/ml) and were triturated gently with a wide-bore pipette for ~25 minutes until single...
cells were released. The single cells were then transferred to a low Ca\textsuperscript{2+} Krebs' solution and were stored in this solution at room temperature. A small volume of this cell suspension was pipetted into the recording chamber, and the cells were allowed to settle to the bottom of the chamber before the experiment was started.

**Solutions**

All experiments were carried out at room temperature (21–23°C). A HEPES-buffered Tyrode's solution was used during electrophysiological recordings. It consisted of (mM) NaCl 135, KCl 5.4, NaH\textsubscript{2}PO\textsubscript{4} 0.33, HEPES 5, MgCl\textsubscript{2} 2, and glucose 5.5. This solution was equilibrated with 100% O\textsubscript{2}, and its pH was adjusted to 7.4 with NaOH (1N). The recording pipettes were filled with a solution containing (mM) potassium aspartate 100, KCl 30, HEPES 5, ATP (disodium salt) 2, MgCl\textsubscript{2} 1, and EGTA 0.1. The pH of this solution was adjusted to 7.2 with KOH. During experiments in which Ca\textsuperscript{2+} currents were studied, the K\textsuperscript{+} in the internal solution was replaced with Cs\textsuperscript{+}, and Ba\textsuperscript{2+} (10 mM) was substituted for Ca\textsuperscript{2+} (2 mM) in the external solution. Collagenase was obtained from Yakult Corp., Tokyo; all other reagents were from Sigma Chemical Co., St. Louis, Mo.

**Electrical Recordings**

The gigaseal patch-clamp technique was used in the whole-cell configuration. The voltage amplifier was an Axopatch 1D (Axon Instruments, Burlingame, Calif.). Microelectrodes (1.5 mm o.d., World Precision Instruments, New Haven, Conn.) were prepared on a Flaming/Brown horizontal puller (Sutter Instrument Co., Novato, Calif.). The resistance of the filled patch pipettes was 3–5 M\textOmega. Current recordings were filtered at 1 kHz using a four-pole Bessel filter. Membrane potential and current were monitored on a storage oscilloscope (model V134, Hitachi, Tokyo) and were recorded at 2.5 kHz on a microcomputer (model 386SX, Zenith) using our data acquisition and display software. After the experiment, the stored data were sent to a DEC Vax 11-750 computer for analysis, graphing, and archiving. The capacitance of the pipette and cell was electronically compensated using the compensation circuit of the Axopatch amplifier. Preliminary experiments in which the capacitance transient was recorded at a fast time base and then integrated confirmed that the cell capacity readings obtained from the amplifier did not differ significantly from the integrated response of the capacitance transient. The series resistance was compensated (20–60%) using the series resistance compensation knobs of the Axopatch.

**Single-Channel Recordings**

Cell-attached patch recordings were obtained from some cells (n=6). The pipette was fire-polished and coated with Silgard. The solution in the pipette was the same as used for the whole-cell recordings (i.e., high K\textsuperscript{+}). The cells were bathed in a HEPES-buffered Tyrode's solution containing 2 mM Ca\textsuperscript{2+}. Single-channel events were stored on a Panasonic VHS recorder using a PC-1 device (Medical Systems Corp., Greenvale, N.Y.) and later were played back to the computer for single-channel analysis using the IPROC program (Axon Instruments). The recordings were filtered using the four-pole Bessel filter at 3 kHz. Single exponential functions were fitted to ensemble averages using a version of the DISCRETE program.

**Results**

**Cell Morphology and Passive Membrane Properties**

Our enzymatic dissociation procedure yielded 20–40% viable, relaxed cells. The remainder of the cell population was either partially contracted or completely rounded up. The average length of the relaxed cells that were used for recordings was 144±41 \textmu m, and their width was 8.5±1.7 \textmu m. These cells contracted on exposure to isotonic KCl and/or to noradrenaline (10 \textmu M). In general, a larger yield of relaxed cells was obtained from younger patients, although this trend was not evaluated systematically. A total of 74 cells were studied in these experiments.

The average resting potential of the relaxed cells from the human cysitic artery was −36±7 mV, and their average capacitance was 34.9±12.6 pF (mean±SD, n=42). Thus, assuming a specific capacitance of 1 \mu F/cm\textsuperscript{2}, the mean capacitative membrane area of the cell was 3,520±1,100 \mu m\textsuperscript{2}, whereas the geometric surface area, assuming a cylinder, was 1,922±478 \mu m\textsuperscript{2}. Part of this difference arises from extensive caveolae in smooth muscle cells.10,11 Multiplying the surface area by the input resistance gives a specific membrane resistance of 70.4 k\textOmega \cdot \text{cm}\textsuperscript{2}. When this parameter is used to calculate the DC space constant,12 assuming a specific resistivity of the myoplasm of 250 \Omega cm, a value of ∼2,440 \mu m is obtained. Since this is ∼15 times the length of a single cell from the human cysitic artery, the myocyte is virtually isopotential in both the resting state and when it is activated.

Regenerative responses were never observed when depolarizing pulses were applied under current clamp. The lack of an evoked action potential shows that the human cystic artery is a quiescent smooth muscle similar to the common carotid artery13 and the rabbit mesenteric artery.14 Small hyperpolarizing current pulses produced large electrotonic potentials, demonstrating a very high input resistance (2.0±0.8 [mean±SD] \textOmega F, n=20) in these cells, similar to other vascular smooth muscle cells, such as rabbit coronary artery cells (3.79 \textOmega F).15

**Outward Currents**

The inset in Figure 1 shows a family of outward currents. These were recorded, using the whole-cell voltage clamp, from a holding potential of −60 mV when the cell was depolarized to +40 mV for 200 msec in 10-mV increments. These currents reached a maximum value within the first 50–100 msec and remained at this level for the duration of the depolarizing pulse. Relatively large, noisy outward currents were consistently observed at potentials positive to −30 mV. The isochronal current–voltage curve measured at the end of the 200-msec voltage step exhibits outward rectification (mean±SEM, n=10). Hyperpolarizing voltage pulses elicited only very small inward currents. No net inward currents were observed during depolarization in any of the 50 cells that were studied.

To obtain information regarding the ionic selectivity of the channels carrying this outward current, reversal potential measurements were made using a standard
double-pulse protocol. Figure 2 shows tail currents recorded after activating the outward current with a 100-msec depolarization to +40 mV. The reversal potential was $-65 \pm 5$ mV in 5.4 mM $[K^+]_o$ (Figure 2). Assuming a $[K^+]_o$ of 130 mM, the equilibrium potential for $K^+$ can be calculated to be $-80$ mV at 23°C. The difference is likely due to a junction potential that develops between the pipette tip (containing $K^+$ asparagine) and the bathing solution (containing mainly NaCl). The measured junction potential was $-11$ mV with the filling solution used in these experiments. When $[K^+]_o$ was changed systematically, the resulting shift of the reversal potential gave a slope of 58 mV per 10-fold change in $[K^+]_o$ (i.e., the value predicted by the Nernst relation). These results show that the outward current(s) in human cystic artery is due to activation of $K^+$-selective channels.

Pharmacological experiments were conducted to characterize further the type(s) of $K^+$ channels that underlies this outward current. Tetraethylammonium (TEA) is known to block Ca$^{2+}$-activated $K^+$ currents in smooth muscle. Figure 3 shows that TEA (5 mM) reduced the outward $K^+$ current by $-70\%$ at +30 mV. However, increasing the TEA concentration to 10 mM did not result in further inhibition of the $K^+$ currents.
To study the Ca$^{2+}$ dependence of these K$^+$ currents, the [Ca$^{2+}]_o$ was increased from 2 to 5 mM. This resulted in a consistent increase in the outward current as shown in the current–voltage curve of Figure 4. When extracellular Ca$^{2+}$ was replaced with Co$^{2+}$ (2 mM), the outward current was strongly suppressed (55±7%, n=10) (Figure 5). Reduction in the outward current was also observed when either nifedipine (0.1 μM, 47±10%, n=2) or CdCl$_2$ (0.5 mM, 43±5%, n=3) was used to block the transmembrane Ca$^{2+}$ influx, as well as when the cells were dialyzed intracellularly with 10 mM EGTA. In combination, these results suggest that a significant component of the time- and voltage-dependent outward current is Ca$^{2+}$-activated K$^+$ current, which is dependent on the transmembrane influx of Ca$^{2+}$. 4-Aminopyridine (4 mM) had no effect on the outward currents, indicating that the residual current remaining in the absence of Ca$^{2+}$ was not the type of K$^+$ current that has been described in the portal vein.17

In some cells (n=20), immediately after rupturing the membrane, spontaneous transient outward currents (STOCs) were observed. The amplitude and frequency of these currents increased on depolarization, and they reversed between −60 and −70 mV (Figure 6). STOCs were sensitive to external Ca$^{2+}$, as shown in Figure 6C, where perfusion of the cell with nominal Ca$^{2+}$-free solution almost completely abolished STOC activity. When cells were dialyzed with 10 mM EGTA, no STOC activity could be detected. These results indicate that STOCs represent transient, perhaps random, activation of Ca$^{2+}$-activated K$^+$ channels. Both STOCs and the outward currents evoked on depolarization were blocked by TEA.

**Single-Channel Recordings**

To obtain data describing the biophysical properties of these Ca$^{2+}$-activated outward currents, they were studied at the single-channel level. Cell-attached patch recordings were made when the cells were bathed in HEPES-buffered Tyrode’s solution and isotonic K$^+$ was present in the pipette. Only one type of K$^+$ channel activity was observed; it had a conductance of 117±6 pS (n=5) (Figure 7). This type of channel activity has been previously demonstrated to be due to Ca$^{2+}$-activated K$^+$ channels.18 To examine the voltage dependence of these single-channel events underlying the outward tails (Figure 2), a double-pulse protocol was used. The patch was held for 10 seconds at −10 mV (with respect to rest) and then stepped to test potentials ranging from −80 to −20 mV (with respect to rest) for 1 second. Figure 8 illustrates the single-channel current obtained on repolarization to these potentials; the single-channel events from four consecutive sweeps are shown at −80 mV (panel A), −60 mV (panel B), −40 mV (panel C), and −20 mV (panel D). At −80 and −60 mV, channel activity deactivated within 30 msec; at depolarized potentials, channel activity continued for the duration...
of the pulse. The ensemble averages from 20 sweeps of the single-channel activity at each potential are illustrated below the raw data. The time course of deactivation was fast at negative potentials, corresponding to the fast deactivation of the tail currents observed in the whole-cell measurements (see Figure 2). The time constants were obtained by single-exponential fits to the ensemble averages. At -80 mV, the time constant for deactivation was 22.3 msec; at -60 mV, it was 35.2 msec; at -40 mV, it was 36.6 msec; and at -20 mV, the deactivation time constant increased to 81.8 msec.

**Inward Currents**

To record the inward current(s) without interference from the outward current(s), the solution in the recording pipette was switched to one containing Cs⁺ instead of K⁺. Even under these conditions, only very small net inward currents (<10 pA) could be recorded in Tyrode’s bathing solution containing normal (2 mM) Ca²⁺. In these experiments, therefore, we recorded the inward currents in 10 mM Ba²⁺ solutions. Under these conditions, peak inward currents ranging from 30 to 100 pA were observed in these cells (Figure 9). The Ba²⁺ current activated at approximately -20 mV and peaked at +10 mV; its apparent reversal potential was near +60 mV.

Two types of Ca²⁺ currents have been recorded in some vascular smooth muscle cells. When these preparations are depolarized from relatively negative holding potentials (-80 mV), low-threshold, rapidly inactivating (T-type) Ca²⁺ currents are observed. In contrast, when cystic artery cells are held at -80 mV and depolarized to various potentials, the threshold for the activation of the Ba²⁺ current was similar to that from a holding potential of -40 mV (Figure 9), suggesting that, in these cells, no T-type inward current is present. To obtain additional information about the functional role of the transmembrane Ca²⁺ current in human cystic artery, the voltage dependence of its steady-state inactivation was measured using the double-pulse protocol. Conditioning pulses of varying amplitudes were applied for 10 seconds, preceding a fixed test pulse to +10 mV. A steady-state inactivation curve was obtained by dividing the peak current amplitude from various conditioning pulses by the maximal current. The data were fit by a Boltzmann distribution (Figure 9D). The half-inactivation potential estimated from this curve was -21.5 mV, and at this potential the slope factor was 9.7 mV.

This inward current was increased markedly (about twofold) by Bay K 8644 (1 μM) as shown in Figure 10. The current–voltage curve in the presence of Bay K 8644 was shifted to the left, as has been described in other tissues.¹⁰

**Figure 5.** Effect of Co²⁺ (2 mM) on K⁺ currents. Panel A: Current recordings in control conditions (left) and in the presence of 2 mM Co²⁺ (right). Holding potential was -70 mV. Current recordings are superimposed at 10-mV increments from -50 to +10 mV. Panel B: Graph showing current–voltage curve before (closed circles) and after (open triangles) Co²⁺.

**Figure 6.** Recordings of spontaneous transient outward currents (STOCs) in human cystic artery cells. Panel A: Voltage dependence of STOCs. Panel B: STOC activity recorded on a faster time base. Panel C: Effects of removal of extracellular Ca²⁺ on STOC activity. Upper bars show change of external Ca²⁺ from 5 mM to nominally Ca²⁺-free solution.
Nifedipine (0.1 μM) abolished these inward currents. These results indicate that the Ca\(^{2+}\) current in the human cystic artery is due to L-type Ca\(^{2+}\) channel activity.

**Discussion**

**Outward Currents**

Our results show that the predominant K\(^+\) current in the human cystic artery is a Ca\(^{2+}\)-activated K\(^+\) current, which is strongly dependent on Ca\(^{2+}\) influx. As expected for vascular smooth muscle, TEA strongly inhibits this outward current, and high intracellular EGTA, which decreases [Ca\(^{2+}\)], has a similar effect. Moreover, raising the [Ca\(^{2+}\)]\(_o\) enhances the Ca\(^{2+}\)-activated K\(^+\) current, and Ca\(^{2+}\) channel blockers significantly reduce it. A component of this outward current appeared to be insensitive to TEA, Ca\(^{2+}\) antagonists, Ca\(^{2+}\) removal, and 4-aminopyridine. These findings indicate that the extracellular Ca\(^{2+}\)-insensitive component of the outward current in the cystic artery differs from that described in the rabbit portal vein.\(^{17}\) In the absence of extracellular Ca\(^{2+}\), the K\(^+\) currents activated on depolarization may be activated by the Ca\(^{2+}\) released from the sarcoplasmic reticulum. Further experiments would be required to demonstrate the nature of the extracellular Ca\(^{2+}\)-insensitive outward current. STOCs were also observed in the cystic artery. STOCs have been described previously in a number of vascular and visceral smooth muscles.\(^{22,23}\) They are thought to be triggered by spontaneous Ca\(^{2+}\) release from the sarcoplasmic reticulum (i.e., a Ca\(^{2+}\)-induced Ca\(^{2+}\) release mechanism).

Single cells from the human cystic artery have somewhat depolarized resting membrane potentials. However, the resting potentials we measured are comparable to many other isolated vascular muscle cells (e.g., −33 mV in rabbit coronary artery,\(^{15}\) −30 to −60 mV in the guinea pig coronary artery,\(^{24}\) and −33 mV in single cells from the human saphenous vein\(^{5}\)). It is possible that the resting potential has not been recorded accurately (±10 mV) in our experiments. Human cystic artery cells have very high input resistances; therefore, an extremely high seal resistance (50–100 GΩ) must be formed before membrane potential can be measured accurately (to prevent significant shunting due to a voltage divider effect). In addition, since all of our recordings were made at 23°C, the activity of the Na\(^+\)-K\(^+\) pump current was decreased perhaps quite substantially. The contribution of the outward Na\(^+\)-K\(^+\) pump current to the membrane potential may be 5–10 mV,\(^{25}\) although direct measurements have not been made.

**Functional Role of the Ca\(^{2+}\)-Activated K\(^+\) Current**

It has been postulated that large conductance Ca\(^{2+}\)-activated K\(^+\) currents (100–250 pS) are responsible for the repolarization of smooth muscle cells.\(^{17,26}\) Trieschmann and Isenberg\(^{27}\) have also suggested that Ca\(^{2+}\)-activated K\(^+\) currents contribute to the resting potential in the pig coronary artery. In the human cystic artery,
FIGURE 8. Single-channel events obtained on repolarization to various test potentials. At the top of each panel (tracings at a), four consecutive 1-second sweeps are illustrated for each applied voltage, -80 mV (panel A), -60 mV (panel B), -40 mV (panel C), and -20 mV (panel D). Spikes at the beginning of each tracing represent capacitative artifact. Note that channel activity is recorded only at the beginning of the sweep at -80 mV but is present for longer times at -20 mV. At the bottom of each panel (tracings at b), ensemble averages of channel events appearing in tracings at a are shown. Twenty sweeps of single-channel activity were averaged for each applied potential. These ensemble averages were then fit by single exponentials using the DISCRETE program. Calibrations are indicated in panel A for tracings at a and b for all panels.
the observed voltage dependence of the Ca\(^{2+}\)-activated K\(^+\) channels show that the Ca\(^{2+}\)-activated K\(^+\) current deactivates quickly and completely at membrane potentials negative to approximately −60 mV (Figure 2); therefore, its contribution to the resting potential is likely to be small. Excised patch recordings\(^{28,29}\) have shown that Ca\(^{2+}\)-activated K\(^+\) channels are sensitive to [Ca\(^{2+}\)]\(_o\), and Trieschmann and Isenberg\(^{27}\) have reported that the open probability of these channels can be markedly altered by Mg\(^{2+}\). Although the protocol that we followed allows us to study the voltage dependence of the Ca\(^{2+}\)-activated K\(^+\) channels under conditions in which the metabolic state of the cells remains unaltered, an understanding of the physiological role of this current in the human cystic artery will require a more complete analysis of these modulating factors.

**Inward Currents**

Addition of Cs\(^+\) to the recording pipette to block K\(^+\) currents was an essential prerequisite for identification of the time- and voltage-dependent Ca\(^{2+}\) current(s). Even under these conditions, net inward currents could be detected consistently only when Ba\(^{2+}\) (10 mM) was present. Our results show that in the cystic artery there is only one type of calcium current and that it has many of the characteristics of the L-type channel.\(^{30}\) Thus, when Ba\(^{2+}\) inward currents were elicited from different holding potentials, a T-type current activity was never observed; that is, a fast transient component was not “unmasked” by hyperpolarizing holding potentials. Since T-type channels are equally permeable to Ca\(^{2+}\) and Ba\(^{2+}\), the substitution of Ba\(^{2+}\) for Ca\(^{2+}\) should not have blocked or significantly reduced T-channel activity. However, it can be argued that since T-type Ca\(^{2+}\) currents are relatively small, they may have been missed in our recordings. Perhaps the strongest evidence for the presence of predominantly L-type Ca\(^{2+}\) current in the human cystic artery is that the current is sensitive to the dihydropyridine Ca\(^{2+}\) agonists and antagonists; that is, the inward currents in the cystic artery were completely blocked by nifedipine (0.1 μM) and enhanced markedly by Bay K 8644 (1 μM). The voltage-dependent inactivation of the Ca\(^{2+}\) current is well described by a Boltzmann distribution, suggesting that the inactivation mechanism

**FIGURE 9.** Inward currents in the human cystic artery. Ba\(^{2+}\) (10 mM) was used as the charge carrier, and outward currents were blocked by Cs\(^+\) (130 mM) in the pipette. Inward current recordings obtained on depolarization from holding potential (V\(_h\)) of −40 mV (panel A) and −80 mV (panel B) are illustrated. The test voltages are given against each tracing. The current–voltage relation is plotted in panel C for peak Ba\(^{2+}\) currents from holding potentials of −40 mV (open triangles) and −80 mV (closed circles) (n=6, mean±SEM). The steady-state inactivation curve of the Ba\(^{2+}\) current is plotted in panel D and is fitted with the Boltzmann distribution (n=3, mean±SEM) as described in the text. I\(_p\), peak current amplitude; I\(_{max}\), maximal current.
is voltage dependent when Ba\(^{2+}\) is the charge carrier. However, an involvement of a Ca\(^{2+}\)-dependent inactivation mechanism cannot be ruled out on the basis of our results, since the currents were recorded in Ba\(^{2+}\) (10 mM) with intracellular EGTA (10 mM); therefore, Ca\(^{2+}\)-induced inactivation may have been obscured.

In summary, our data describe a number of important characteristics of the ionic currents in human vascular smooth muscle cells and suggest that this preparation may be a useful model for studying modulation of the ionic currents in single cells from a human artery.

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References


17. Beech DJ, Bolton TB: Two components of potassium current activated by depolarization of single smooth muscle cells from the rabbit portal vein. J Physiol (Lond) 1989;418:293–309


27. Trieschmann U, Isenberg G: Ca-activated K channels contribute to the resting potential of vascular myocytes: Ca sensitivity is increased by intracellular Mg. Pflugers Arch 1989;414:S183–S184


Ionic currents in single cells from human cystic artery.
H I Akbarali, D G Wyse and W R Giles

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