Research article

Modulation of L-type Ca\(^{2+}\) current but not activation of Ca\(^{2+}\) release by the gamma\(_{1}\) subunit of the dihydropyridine receptor of skeletal muscle

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Abstract

Background: The multisubunit (\(\alpha_{1S}\),\(\alpha_{2-\delta}\), \(\beta_{1a}\) and \(\gamma_{1}\)) skeletal muscle dihydropyridine receptor (DHPR) transduces membrane depolarization into release of Ca\(^{2+}\) from the sarcoplasmic reticulum (SR) and also acts as an L-type Ca\(^{2+}\) channel. To more fully investigate the function of the \(\gamma_{1}\) subunit in these two processes, we produced mice lacking this subunit by gene targeting.

Results: Mice lacking the DHPR \(\gamma_{1}\) subunit (\(\gamma_{1}\) null) survive to adulthood, are fertile and have no obvious gross phenotypic abnormalities. The \(\gamma_{1}\) subunit is expressed at approximately half the normal level in heterozygous mice (\(\gamma_{1}\) het). The density of the L-type Ca\(^{2+}\) current in \(\gamma_{1}\) null and \(\gamma_{1}\) het myotubes was higher than in controls. Inactivation of the Ca\(^{2+}\) current produced by a long depolarization was slower and incomplete in \(\gamma_{1}\) null and \(\gamma_{1}\) het myotubes, and was shifted to a more positive potential than in controls. However, the half-activation potential of intramembrane charge movements was not shifted, and the maximum density of the total charge was unchanged. Also, no shift was observed in the voltage-dependence of Ca\(^{2+}\) transients. \(\gamma_{1}\) null and \(\gamma_{1}\) het myotubes had the same peak Ca\(^{2+}\) amplitude vs. voltage relationship as control myotubes.

Conclusions: The L-type Ca\(^{2+}\) channel function, but not the SR Ca\(^{2+}\) release triggering function of the skeletal muscle dihydropyridine receptor, is modulated by the \(\gamma_{1}\) subunit.

Background

In skeletal muscle, the dihydropyridine receptor (DHPR) consists of \(\alpha_{1S}\), \(\alpha_{2-\delta}\), \(\beta_{1a}\) and \(\gamma_{1}\) subunits [1]. This complex is responsible for the L-type Ca\(^{2+}\) current and serves as
the voltage sensor for excitation-contraction (EC) coupling. In the latter process, the movement of electrical charges in the \( \alpha_S \) subunit promotes a conformational change that opens the ryanodine receptor type-1 (RyR1) in the sarcoplasmic reticulum membrane (SR) leading to an increase in cytosolic Ca\(^{2+} \) [2,3]. The functional interactions between the DHPR subunits necessary for opening the Ca\(^{2+} \) channel are only partially known. Further, the interactions between DHPR subunits and RyR1 also are incompletely understood. The \( \alpha \) subunit is a large protein that contains the basic functional elements of the L-type Ca\(^{2+} \) channel, including the Ca\(^{2+} \) selectivity, voltage-dependent gating, and sensitivity to dihydropyridines. The cytoplasmic loop between repeats II and III of the \( \alpha_S \) subunit interacts closely with RyR1 and is an important determinant of skeletal type EC coupling [4,5]. A region in the cytoplasmic loop between repeats I and II of the \( \alpha \) subunit, referred to as the AID [6], binds tightly with a 30 amino acid region on the \( \beta \) subunit, referred to as the BID [7]. \( \beta \) subunits are ∼55 to 65 kDa proteins essential for channel assembly and/or membrane targeting, as well as for modulation of channel kinetics [8]. The \( \alpha_\gamma-\delta \) subunit is a highly glycosylated ∼175 kDa protein formed by two disulfide-linked peptides encoded by the same gene. Transmembrane topology and functional analyses suggest the \( \alpha_2-\delta \) subunit is composed of a single transmembrane domain and a short cytoplasmic tail of only five residues [9,10]. Given this topology, the \( \alpha_2-\delta \) subunit is most likely to interact with the \( \alpha \) and/or the \( \gamma_i \) subunits. The \( \gamma_i \) subunit is a ∼32 kDa skeletal muscle-specific protein with four presumptive transmembrane domains [11,12,13]. The transmembrane topology of the \( \gamma_i \) subunit and the critical binding domains are unknown at this time.

EC coupling is initiated by voltage-dependent charge movements in the S4 segments of the DHPR \( \alpha_\gamma \) subunit, whose expression is dependent on the presence of the \( \beta_\gamma \) subunit [14]. The C-terminus of the \( \beta_\gamma \) subunit has also been shown to be important in EC coupling, presumably by interaction with RyR1 [15]. The role of the \( \alpha_2-\delta \) subunit on skeletal EC coupling is unknown, but in heterologous expression systems it has been demonstrated to increase the amount of charge movement [16]. The role of the \( \gamma_i \) subunit in Ca\(^{2+} \) channel function is of particular interest given the discovery of a second \( \gamma \) subunit, \( \gamma_2 \) or stargazin, which is expressed in neurons and is responsible for the stargazer mutation in mice [17]. Subsequently, several other \( \gamma \) subunit genes have been identified and shown to be expressed in brain and peripheral tissues [18,19]. In the present study we examined the role of the \( \gamma_i \) subunit in L-type Ca\(^{2+} \) current and EC coupling in skeletal myotubes. Absence of the \( \gamma_i \) subunit slows inactivation and produces a depolarizing shift in the Ca\(^{2+} \) current inactivation vs. voltage curve, in agreement with results from an independently produced \( \gamma_i \) knockout mouse [20]. However, absence of \( \gamma_i \) does not affect the voltage dependence or the magnitude of charge movements and Ca\(^{2+} \) transients. Overall, the \( \gamma_i \) subunit appears to promote inhibition of the Ca\(^{2+} \) channel function of the skeletal DHPR. While this subunit is clearly non-essential for activation of the L-type Ca\(^{2+} \) channel and for triggering skeletal-type EC coupling, \( \gamma_i \) appears to specifically modulate the Ca\(^{2+} \) channel function of the skeletal DHPR.

Results and Discussion

Inactivation of the DHPR \( \gamma_i \) subunit by gene targeting

Gene targeting was used to replace the entire coding region of the \( \gamma_i \) gene with the neo gene, as described in Materials and Methods (Fig. 1). Targeted ES cell clones were injected into C57Bl/6J blastocysts and chimeras obtained. After additional breeding, a line containing the \( \gamma_i \) targeted allele was obtained. Fig. 1A shows Southern blots from DNA of control (+/+) and \( \gamma_i \) het (+/-) and \( \gamma_i \) null (-/-) mice, after digestion with EcoRI and hybridization to probe 1. The \( \gamma_i \) het and \( \gamma_i \) null mice show the predicted 12.5 kb restriction fragment indicative of the targeted allele (Fig. 1B). Similarly for the Southern blot of DNA digested with HindIII and hybridized to probe 2, the \( \gamma_i \) het and \( \gamma_i \) null mice show the predicted 3.5 kb restriction fragment indicative of the targeted allele (Fig. 1C). The \( \gamma_i \) het and \( \gamma_i \) null mice have no visible abnormalities and are fertile. Western blots showing the expression of the DHPR subunits are shown in Fig 1D. The amount of the \( \gamma_i \) subunit present in control, \( \gamma_i \) het and \( \gamma_i \) null samples as a function of total membrane protein was determined in two independent membrane preparations. These data indicate that the amount of \( \gamma_i \) expression in membranes from \( \gamma_i \) null muscle was not detectable above background (no primary antibody) as expected from the fact that the entire gene was deleted. In membranes isolated from the \( \gamma_i \) het mice, the level of expression of the \( \gamma_i \) subunit was 64% of control values for each of the two independent determinations, consistent with what might be expected from the loss of one allele. Whether there is compensation by increased expression of other \( \gamma \) subunits such as the \( \gamma_0 \) and \( \gamma_\beta \) subunits, which were shown recently to be present in skeletal muscle by RT-PCR [19], or alteration of the amount of the other subunits remains to be determined in the future.

Increase in Ca\(^{2+} \) current density and decrease in inactivation in \( \gamma_i \) null and \( \gamma_i \) het myotubes

We examined the impact of the \( \gamma_i \) null mutation on DHPR properties using electrophysiological techniques in primary cultures of skeletal myotubes derived from mouse embryos. Fig. 2A shows whole-cell Ca\(^{2+} \) currents in control, \( \gamma_i \) het and \( \gamma_i \) null myotubes in response to 500-ms depolarizing voltage steps from a holding poten-
Inactivation of the $\gamma_1$ subunit by gene targeting. A) i. The normal $\gamma_1$ gene, which contains 4 exons (represented by black boxes). ii. The targeting vector contains a fragment with 4.4 kb of homology to a region 5' of exon 1 and a 2.6 kb fragment with homology to a region 3' of exon 4 (thick horizontal lines). These fragments are separated by the neo gene, which is used to select the ES cells in culture. iii. The modified $\gamma_1$ locus. When recombination occurs in the regions indicated by the X's, all 4 exons of the $\gamma_1$ gene are replaced by the neo cassette. B, C) Southern blots of DNA, either digested with EcoRI and hybridized to probe 1 (B), or DNA digested with HindIII and hybridized to probe 2 (C), from control (+/+), $\gamma_1$ het (+/-) and $\gamma_1$ null (-/-) mice. These data show the predicted size bands for the wild type and targeted alleles. D) Western blots for each of the four skeletal DHPR subunits from control (+/+), $\gamma_1$ het (+/-) and $\gamma_1$ null (-/-) mice. The $\gamma_1$ subunit is absent from the $\gamma_1$ null mice and at approximately half the normal level in the $\gamma_1$ het mice.
tial of -40 mV. Current traces for only three voltage steps are shown for clarity. All three cell types displayed the slow, high-voltage activated L-type Ca\(^{2+}\) current typical of skeletal myotubes. When the holding potential was -80 mV, we also observed a T-type Ca\(^{2+}\) current, which was seen in approximately half of the cells tested (data not shown). Ca\(^{2+}\) conductance vs. voltage curves were computed in Fig. 2B for the three cell types. The sigmoidal lines correspond to a Boltzmann fit to the population mean while parameters fitted to each cell are shown in Table 1. These data show that the maximum conductance, \(G_{\text{max}}\), for both the \(\gamma_1\) het and \(\gamma_1\) null cells are significantly larger (p<0.05) than the values for control cells. However, the half-activation potential (\(V_{1/2}\)) and the steepness of the curve (factor k) were not significantly different (see Table 1). Thus, full or partial elimination of the \(\gamma_1\) subunit produced a significant increase in the Ca\(^{2+}\) current density without a change in the voltage-dependence of the Ca\(^{2+}\) conductance.

At intermediate potentials such as +30 mV (Fig. 2A), the Ca\(^{2+}\) current of \(\gamma_1\) het and \(\gamma_1\) null cells inactivated more slowly than that of control cells. To examine changes in inactivation, we used the paired-pulse protocol shown in Fig. 3A, which measured the Ca\(^{2+}\) current not inactivated by the pre-pulse. It has been shown that inactivation of
skeletal L-type Ca\(^{2+}\) channels approaches steady-state extremely slowly, on the time scale of tens of seconds [21]. However, for comparative purposes and to be consistent with previous measurements in Ca\(^{2+}\) channels lacking \(\gamma_1\) [20,22], we chose a much shorter protocol. Cells were held at -80 mV, then stepped to a series of depolarizing (pre-pulse) voltages for 2.5 s to promote inactivation, then stepped to -50 mV for 100 ms to close non-inactivated channels. The non-inactivated Ca\(^{2+}\) current was measured during a 200-ms test pulse to +20 mV. In control cells, there was almost complete inactivation of the Ca\(^{2+}\) current when the pre-pulse potential was in the positive range. This can be seen by comparing the amplitude of the Ca\(^{2+}\) current during the test pulse in traces 1, 2, and 3, corresponding to pre-pulse potentials to -70 mV, +30 mV, and +60 mV, respectively. In con-
trast, \( \gamma_1 \) het and \( \gamma_1 \) null cells showed less inactivation during the pre-pulse, and thus a significantly larger Ca\(^{2+} \) current during the test pulse. Furthermore, pre-pulse potentials larger than +30 mV did not promote additional inactivation, which can be seen by comparing traces 2 and 3 in the \( \gamma_1 \) het and \( \gamma_1 \) null cells. Fig. 3B shows the maximum Ca\(^{2+} \) current during the test pulse plotted as a function of the pre-pulse potential. In control cells, inactivation was complete when the pre-pulse potential was above +30 mV (-6.9 ± 0.8 pA/pF at -70 mV vs. -0.2 ± 0.2 pA/pF at +70 mV, n = 6 cells). In \( \gamma_1 \) null cells, inactivation reached a plateau when the pre-pulse potential was more positive than +20 mV (-8.5 ± 0.9 pA/pF at -70 mV vs. -3.2 ± 0.7 pA/pF at +70 mV, n = 8 cells). Incomplete inactivation was also seen in \( \gamma_1 \) het cells, although this was less pronounced than in \( \gamma_1 \) null cells. Since the pre-pulse was not sufficiently long for inactivation to reach steady-state, we are not certain whether the observed incomplete inactivation reflects a component of the \( \gamma_1 \) null Ca\(^{2+} \) current that inactivates extremely slowly or a "truly" non-inactivating component. To compare the voltage-dependence of the inactivation produced by the 2.5-second pre-pulse, the Ca\(^{2+} \) current during the test pulse was normalized and fit to the Boltzmann equation (Fig. 3C). The curves for \( \gamma_1 \) het and \( \gamma_1 \) null cells showed a significant (p<0.05) shift to more positive potentials compared to control cells. The same conclusion was reached by comparing the Boltzmann parameters of individual cells in Table 1. Since control cells had a more negative half inactivation potential, the higher Ca\(^{2+} \) conductance of \( \gamma_1 \) het and \( \gamma_1 \) null cells reported in Fig. 2 could be explained by a larger steady-state inactivation of the control Ca\(^{2+} \) current at the -40 mV holding potential. To determine if this was the case, we measured the Ca\(^{2+} \) conductance during the pre-pulse for the same cells analyzed in Fig 3B,C. The pre-pulse was delivered from a holding potential of -80 mV, and therefore the pre-pulse current should be less contaminated by steady-state inactivation. \( G_{max} \) at the peak of the Ca\(^{2+} \) current during the pre-pulse was 146 ± 12 pS/pF for controls, 172 ± 18 pS/pF for \( \gamma_1 \) het and 186 ± 13 pS/pF for \( \gamma_1 \) null cells with t-test statistical significance for controls vs. \( \gamma_1 \) null cells of p = 0.034 and for controls vs. \( \gamma_1 \) het cells of p = 0.064. Thus the higher Ca\(^{2+} \) conductance of \( \gamma_1 \) het and \( \gamma_1 \) null

| CELL TYPE        | G-V     | Q-V     |
|------------------|---------|---------|
|                  | \( G_{max} (\text{pS/pF}) \) | \( V_{1/2} (\text{mV}) \) | \( k (\text{mV}) \) | \( Q_{max} (\text{fC/pF}) \) | \( V_{1/2} (\text{mV}) \) | \( k (\text{mV}) \) |
| Control, +/-     | 131 ± 6.2 | 14.7 ± 2.1 | 4.2 ± 0.4 | 5.3 ± 0.4 | 11.4 ± 2.5 | 15.4 ± 1.1 |
| \( \gamma_1 \) het, +/- | 161 ± 11.5 | 14.3 ± 2.8 | 5.0 ± 0.8 | 5.3 ± 0.3 | 14.4 ± 0.6 | 14.7 ± 2.0 |
| \( \gamma_1 \) null, +/- | 170 ± 9.6 | 10.9 ± 0.9 | 4.5 ± 0.4 | 5.3 ± 0.9 | 17.8 ± 1.9 | 15.8 ± 2.2 |

Entries (mean ± SEM) correspond to Boltzmann parameters fitted to each cell (number of cells n, in parenthesis). The only statistically significant differences (p<0.05) were for either \( \gamma_1 \) het or \( \gamma_1 \) null cells compared to control cells as indicated. The t-test p values were 0.004, 0.009, 0.011, 0.009 for a-d, respectively.

Table 1: Boltzmann parameters of Ca\(^{2+} \) conductance, charge movements and Ca\(^{2+} \) transients of control, \( \gamma_1 \) het and \( \gamma_1 \) null myotubes.
cells persisted at the more negative holding potential. These Ca\(^{2+}\) conductances were approximately 10% higher than those at the -40 mV holding potential reported in Table 1. The additional current could have been contributed by T-type Ca\(^{2+}\) channels (not shown), which are available at the -80 mV holding potential but are inactivated at the more positive holding potential. Also, since the T-type Ca\(^{2+}\) current density was highly variable (see above), this could explain the higher p values compared to those reported in Table 1. In summary, decreasing the amount of the \(\gamma_1\) subunit protein or eliminating it completely resulted in myotubes with a higher Ca\(^{2+}\) current density, a similar voltage-dependence of activation, and a reduced rate of inactivation. These results, with the exception of the change in Ca\(^{2+}\) current density, are in remarkable agreement with those obtained when L-type Ca\(^{2+}\) channel subunits are coexpressed without the \(\gamma_1\) subunit in heterologous cells (see below). They also are similar to results obtained elsewhere in \(\gamma_1\) null myotubes using a 5-second pre-pulse [20]. Furthermore, the incomplete inactivation of the \(\gamma_1\) null Ca\(^{2+}\) current resembles that observed in ts A201 cells expressing DHPRs specifically lacking \(\gamma_1\) and using a 3-second inactivation protocol [22].

The effect of the \(\gamma_1\) subunit on Ca\(^{2+}\) current kinetics has been somewhat confusing until now, because it has not been possible to examine its role using homologous subunits and a homologous expression system. In amphibian oocytes, \(\gamma_1\) was assayed in a complex with \(\alpha_1C, \beta_{1a}\) and \(\alpha_2-\delta\) [23,24]. These investigators found \(\gamma_1\) produced a slight decrease in Ca\(^{2+}\) current density, no change in activation kinetics and a positive shift in the peak Ca\(^{2+}\) current vs. voltage curve. In HEK293 cells using the same subunit composition, the \(\gamma_1\) subunit did not alter the kinetics or density of Ca\(^{2+}\) currents [25]. However, in both oocytes and HEK293 cells, the \(\gamma_1\) subunit increased the rate of Ca\(^{2+}\) channel inactivation and produced a large hyperpolarizing shift in steady-state inactivation [23,25,26]. The results obtained here in skeletal muscle demonstrate that absence of the \(\gamma_1\) subunit increases Ca\(^{2+}\) current density, decreases the inactivation rate, and shifts the voltage-dependence of inactivation in the depolarizing direction. Freise et al. [20] showed that the increase in Ca\(^{2+}\) current was not a result of changes in single channel conductance and suggested the effects were due to changes in channel open probability. However, this remains to be tested directly. Expression of the \(\gamma_2\) subunit in BHK cells expressing \(\alpha_{1A}, \alpha_2-\delta\) and \(\beta_{1a}\) also shifts the voltage-dependence of inactivation ~7 mV in the hyperpolarizing direction [17]. In summary, the two \(\gamma\) subunits that have been characterized to date, namely \(\gamma_1\) in the homologous skeletal myotube system described here and \(\gamma_2\) described above in heterologous BHK cells [17], appear to be inhibitory. In each case, the presence of \(\gamma\) results in fewer Ca\(^{2+}\) channels available for activation.

**Absence of changes in the EC coupling voltage sensor of \(\gamma_1\) null myotubes**

The impact of the \(\gamma_1\) null mutation on myotube EC coupling was inferred from intramembrane charge movements, the bulk of which have been shown to originate from the DHPR voltage sensor [27,28]. Additionally, we measured Ca\(^{2+}\) transients evoked by voltage (i.e., by the movement of the voltage sensor) and compared these two sets of data. To measure DHPR charge movements and to separate these charge movements from those produced by other voltage-gated channels, a) ionic currents were blocked; b) the linear component of the cell capacity was subtracted using a P/4 procedure and by analog compensation; and c) we used a pulse protocol that eliminated the immobilization-sensitive charge movements from voltage-gated Na\(^{+}\) channels and presumably also from T-type Ca\(^{2+}\) channels. Previous studies in dysgenic (\(\alpha_{S}\) null) myotubes transfected with \(\alpha_{S}\) showed that the pulse protocol and the internal and external solutions used here (see Materials and Methods) detected charge movements of a magnitude 5 to 8 fold larger than those present in non-transfected dysgenic myotubes [28]. Thus we are confident that the technique adequately measures intramembrane charge movements in DHPR voltage sensors present in the plasma membrane. Fig. 4A shows "gating-type" currents produced by charge movements in response to 3 of 14 step voltages delivered to each cell (-40, +10, and +70 mV). Charge movements did not occur at -40 mV in any of the cell types. Traces at +10 mV and +70 mV show a transient current at the onset of the voltage step, the ON charge, and an equal inverted current at the end of the voltage step, the OFF charge. The voltage-dependence of the OFF charge estimated by integration is shown in Fig. 4B. The OFF transient was usually less contaminated by ionic current than the ON transient, and for that reason it provides a better estimate of the total DHPR charge. The main contaminant of the ON transient was a non-linear outward current, presumably a K\(^+\) channel that was not always completely blocked by the pipette solution. The OFF charge increased in a sigmoidal fashion and saturated at potentials more positive than +60 mV in all cases. These data were adequately fit by a Boltzmann equation shown by the lines. We found that the fitted maximum charge (\(Q_{max}\)) expressed by the 3 cell types was ~ 5 fC/pF, in agreement with previous determinations in normal myotubes [29]. Furthermore, Boltzmann parameters determined for each cell and then averaged were not significantly different in any of two-by-two comparisons between control, \(\gamma_1\) het or \(\gamma_1\) null cells (Table 1). In summary, the magnitude of intramembrane charge movements and its voltage dependence were unaltered by...
partial or complete removal of the $\gamma_1$ subunit. Furthermore, in agreement with previous data [29], no additional charges moved when pulses were delivered from a holding potential of -120 mV (data not shown). Thus, failure to detect changes in the charge vs. voltage characteristics of $\gamma_1$ null myotubes compared to controls cannot be explained by charge movements in the $\gamma_1$ null myotubes occurring at potentials more negative than the chosen -80 mV holding potential.

Absence of alterations in the voltage dependence of charge movement in the $\gamma_1$ null cells should also be reflected in the voltage-dependence of Ca$^{2+}$ transients. Changes in intracellular Ca$^{2+}$ were measured using confocal line-scan imaging of fluo-4 fluorescence. Fig. 5A shows a line-scan image of a Ca$^{2+}$ transient stimulated by a 50 ms depolarization to +90 mV from a holding potential of -40 mV in each cell type. The scan direction was perpendicular to the long axis of the myotube in a region selected on the basis of a low resting fluorescence and a high stimulated fluorescence (red/yellow color). The two traces on top of each image correspond to the time course of the fluo-4 fluorescence averaged across the entire line-scan at -10 mV, which is close to the threshold, and at +90 mV, which produced the maximum Ca$^{2+}$ transient. Increase in cytosolic Ca$^{2+}$ started at the onset of the depolarization and peaked ~ 100 ms later in the three cell types. For all cells, the peak fluorescence change for the depolarization to +90 mV was >3 $\Delta F/F_0$ units, which is equivalent to >4 times the resting fluorescence. The relatively long recovery time following the depolarization is due to the low Ca$^{2+}$ buffering power of the internal recording solution and is in agreement with previous studies [27,30]. The voltage dependence of $\Delta F/F_0$
measured at the peak of the transient is shown in Fig. 5B. All curves had the same profile which consisted of a threshold for peak Ca\(^{2+}\) release at -10 mV, an increase in peak Ca\(^{2+}\) with pulse potential from -10 mV to +30 mV and a plateau at more positive potentials. Furthermore, two-by-two comparisons of Boltzmann parameters fitted to each cell did not reveal statistically significant differences (see Table 1). In summary, the voltage-dependence of Ca\(^{2+}\) transients and the maximum cytosolic Ca\(^{2+}\) concentration during the transient were not significantly affected by partial or total absence of the \(\gamma_1\) subunit. However, we cannot rule out changes in the rate of SR Ca\(^{2+}\) release since this measurement requires an entirely different internal solution [31]. A recent report indicates that the SR Ca\(^{2+}\) release rate is slightly higher in \(\gamma_1\) null myotubes [32].

**Involvement of DHPR subunits in voltage sensing and EC coupling**

The \(\alpha_{1S}\) pore subunit of the skeletal DHPR is the main determinant of charge movements and EC coupling [4,27,28]. However, other subunits of the DHPR are critical modulators of both functions. In heterologous expression systems, the \(\alpha_\alpha-\delta\) subunit was shown to affect the rate of Ca\(^{2+}\) current inactivation and charge movement immobilization [33]. Studies investigating the role of the \(\beta\) subunit on charge movement vary with the expression system used. In amphibian oocytes, \(\beta\) subunits increase the coupling between charge movements and
channel opening [34,35]. We have shown in skeletal muscle cells lacking the β subunit that reintroduction of variants of β has little effect on either charge movement or the kinetics of activation of the expressed Ca\(^{2+}\) current [15]. However, the carboxyl terminus of β1α is an important determinant of the efficiency of the coupling between charge movement and Ca\(^{2+}\) release [15]. In contrast to these results, earlier studies in which similar β subunits were co-expressed with α1 subunits in oocytes indicated β subunits had major effects on Ca\(^{2+}\) current density and gating kinetics [36,37]. While the reason for these discrepancies is unknown, they highlight the fact that in skeletal muscle, the gating characteristics of the L-type Ca\(^{2+}\) channel might be controlled by interactions beyond those taking place among subunits of the DHPR. For example, a region of RyR1 has been shown to be essential for L-type Ca\(^{2+}\) current expression [38]. Such interaction is critical for skeletal DHPR function and is unlikely to occur in heterologous expression systems.

Conclusions

The ability of the γ1 subunit to selectively modulate the pore function of the DHPR without modulation of charge movements or the voltage dependence of Ca\(^{2+}\) transients is unique, especially since other DHPR subunits participate in both functions. In all likelihood, the charge movement protocol failed to detect gating currents responsible for opening the Ca\(^{2+}\) channel, which are quite small and are only resolved for depolarizations >200 ms [39] compared to the 25 ms used here (Fig. 4). A possible shift in the voltage-dependence of these charges recruited by long depolarizations would be consistent with the shift in Ca\(^{2+}\) current inactivation and remains to be resolved in γ1 null myotubes. However, the protocol accurately measures the immobilization-resistant charge movements that are known to be required for skeletal-type EC coupling [31,40]. Therefore, the γ1 subunit is unlikely to play a critical role in the activation of SR Ca\(^{2+}\) release, in agreement with a recent report [32]. However, shifts in voltage dependence below the limit of resolution (see Materials and Methods) and effects on charge movement and Ca\(^{2+}\) release inactivation cannot be completely ruled out. The γ1 knockout mice provide a unique resource to understand the function of this protein in myotubes in molecular detail.

Materials and Methods

Gene targeting

Bacteriophage clones containing portions of the γ1 gene were isolated from a mouse 129Sv genomic library (Stratagene, La Jolla, CA #940305). To inactivate the γ1 gene, a 4.4 kb BamHI/XhoI fragment upstream of exon 1 was subcloned into a multiple cloning site 5’ of the neo and Herpes virus TK gene that was in pBluescript (Stratagene). A second 2.6 kb EcoRI/BamHI fragment 3’ of exon 4 of the γ1 gene was cloned into a site between the neo and TK genes (Fig. 1Aii). Thus, the targeting vector contains 4.4 kb of identity with the native γ1 gene 5’, and 2.6 kb of identity 3’ of the neo cassette, respectively. The targeting vector also contains the HSV TK gene to allow for negative selection of non-recombinants. A unique BamHI restriction site was used to linearize the plasmid prior to its introduction into mouse ES cells by electroporation. The γ1 gene contains 4 exons, and correct targeting results in the deletion of all 4 exons, which will inactivate the gene (Fig. 1Aiii). Probe 2 was used for identification of targeted clones [41]. 5 μg of the targeting vector was electroporated into 5 × 10^6 Ab1 ES cells. G418 and FIAU (1-(2’-deoxy-2’-fluoro-D-arabinofuranosyl)-5-iodouracil) resistant clones were analyzed by Southern blotting after digestion with either EcoRI and hybridization to probe 1 or digestion with HindIII and hybridization to probe 2 (Fig. 1). Of three clones targeted on the 5’ end, two also were targeted on the 3’ end. These were expanded and used to produce germline chimeras. Mice homozygous for the targeted allele were viable, with no apparent abnormalities. Mice heterozygous and homozygous for the targeted mutation are referred to as γ1 het and γ1 null, respectively.

Western blots

Total microsomes were prepared from the forelimb and hind limb muscles of control, γ1 het, and γ1 null adult mice as previously described [42]. Total microsomes were washed twice with 0.6 M KCl buffer and 100 μg of total membrane protein was applied to a 5-15% SDS polyacrylamide gel. Proteins were transferred to PVDF membranes and analyzed with either anti-α1 subunit antibody [43], or anti-α1 affinity-purified guinea pig antibody #1 [13], or anti-β1a affinity-purified sheep #6 antibody [44], or with anti-γ1 affinity-purified guinea pig antibody #16 and the appropriate secondary antibodies [11]. The subunits were visualized using SuperSignal ECL reagent (Pierce, Rockford, IL). The images were captured on a Chemi-Imager (Alpha Innotech, San Leandro, CA) set to a level just below saturation.

Primary cultures of mouse myotubes

Primary cultures were prepared from hind limbs of day 18 embryos (E18) as described previously [45]. Dissected muscles were incubated for 9 min at 37°C in Ca\(^{2+}/\)Mg\(^{2+}\)-free Hanks balanced salt solution (in mM: 136.9 NaCl, 3 KCl, 0.44 KH\(_2\)PO\(_4\), 0.34 NaHPO\(_4\), 4.2 NaHCO\(_3\), 5.5 glucose, pH 7.2) containing 0.25% (w/v) trypsin and 0.05 % (w/v) pancreatin (Sigma, St. Louis, MO). Mononucleated cells were resuspended in plating medium containing 78% Dulbecco’s modified Eagle’s medium (DMEM) with low glucose (Life Technologies, Rockville, MD), 10 % horse serum (HS, Sigma, St. Louis, MO), 10 % fetal bovine serum (FBS, Sigma, St. Louis, MO), 2% chicken em-
bryo extract (CEE, Life Technologies, Rockville, MD) and plated on plastic culture dishes coated with gelatin at a density of -1 x 10⁴ cells per dish. Cultures were grown at 37°C in 8 % CO₂. After the fusion of myoblasts (~7 days), the medium was replaced with a FBS-free medium (88.75 % DMEM, 10 % HS, 1.25% CEE) and cells were incubated in 5% CO₂. All media contained 0.1 % v/v penicillin and streptomycin (Sigma, St. Louis, MO). All measurements were made when the cells had been in culture for 7-10 days. During this time the maximal current density did not change [45].

Electrophysiological measurements

Whole-cell recordings were performed as described previously [29] using an Axopatch 200B amplifier (Axon Instruments, Foster City, CA). Linear capacity and leak currents were compensated with the circuit provided by the manufacturer. Series resistance, Rs, was compensated up to the point of amplifier oscillation with the Axopatch circuit. Considering all cells in the present study, the linear capacitance (mean ± SE) of control, γnull, and γhet myotubes was 256 ± 25 pF (38 cells), 246 ± 16 pF (34 cells) and 223 ± 24 (23 cells), respectively. Rs of the same cells before analog compensation was 7 ± 0.5 MΩ, 7.2 ± 0.4 MΩ, and 6.3 ± 0.5 MΩ, respectively. Following compensation, the effective Rs was <2 MΩ in all cases measured, although this was not measured systematically. Considering a mean cell capacitance of 240 pF, a typical maximum current density of 5 to 10 pA/pF (see Fig. 3B), and Rs (effective)<2 MΩ, the voltage error was <2.4 to <4.8 mV. All experiments were performed at room temperature. Patch pipettes had an open tip resistance of 1-2 MΩ when filled with the pipette solution. Ca2+ currents were measured from a holding potential of -40 mV. Test pulses of 500 ms in 5 mV increments ranged from -40 mV to +60 mV. Ca2+ conductance vs. voltage curves were obtained by extrapolation of the Ca2+ current to the reversal potential of the cell. To measure inactivation, cells were held at -80 mV, then stepped to a series of depolarizing (pre-pulse) voltages, from -70 mV to +70 mV in 10 mV increments, for 2.5 seconds to promote inactivation, then stepped to -50 mV for 100 ms to close non-inactivated channels, then stepped for 200 ms to the test potential of +20 mV to measure Ca2+ currents, then stepped to the -80 mV holding potential for 5 seconds to permit recovery from inactivation. Charge movement was measured using a protocol that eliminated immobilization-sensitive components [15,28,29]. Voltage was stepped from a holding potential of -80 mV to -35 mV for 750 ms, then to -50 mV for 5 ms, then to test potential for 25 ms, then to -50 mV for 30 ms and finally to the -80 mV holding potential. Subtraction of the linear component was assisted by a P/4 procedure following the pulse paradigm listed above. P/4 pulses were separated by 500 ms and had a duration of 25 ms. Previous studies showed this choice of pulse protocol and internal and external solutions resulted in nifedipine-sensitive charge movements of a magnitude ~5 fold larger than those detected in dysgenic myotubes lacking α1S [29].

Solutions

The external solution in all cases was (in mM) 130 TEA methanesulfonate, 10 CaCl₂, 1 MgCl₂, 10 HEPES titrated with TEA(OH) to pH 7.4. For Ca2+ transients and Ca2+ currents, the pipette solution was (in mM) 140 Ca²⁺-Aspartate, 5 MgCl₂, 0.1 EGTA (Ca²⁺ transients) or 5 mM EGTA (Ca²⁺ currents), and 10 MOPS-CSOH pH 7.2. For charge movements, the pipette solution was (in mM) 120 NMG (N-methyl glucamine)-Glutamate, 10 HEPES-NMG, 10 EGTA-NMG pH 7.3. For charge movements, the external solution was supplemented with 0.5 mM CdCl₂ and 0.5 mM LaCl₃ to block the Ca²⁺ current and 0.05 mM TTX to block residual Na⁺ current.

Ca²⁺ transient measurements

Ca²⁺ transients were measured using a confocal microscope in line-scan mode as described previously [46,47,48]. Cells were loaded with 4 mM fluo-4 (fluorescein acetoxymethyl (AM) ester, Molecular Probes, Eugene, OR) for 20 to 40 min at room temperature. Stocks of fluo-4 (1 mg/ml) were made in DMSO and stored frozen. All experiments were performed at room temperature. Cells were viewed with an inverted Olympus microscope with a 20X objective (N.A. = 0.4) and a Fluoview confocal attachment (Olympus, Melville, NY). The 488 nm spectrum line necessary for fluo-4 excitation was provided by a 5 mW argon laser attenuated to 20% with neutral density filters. The fluorescence intensity, F, was calculated by densitometric scanning of line-scan images and was averaged over the entire width of the cell. The background fluorescence intensity (Fo) was averaged in the same manner from areas of the same image prior to the voltage pulse. The fluorescence unit ∆F/Fo corresponds to (F-Fo)/Fo. The limit of fluorescence detection, based on microscope settings and the average resting fluo-4 fluorescence, was ~0.1 ∆F/Fo units. That is, we could detect a ~10% change above the cell resting fluorescent. Using a pseudo-ratiometric equation for estimating the cytosolic free Ca²⁺ [48] and assuming a resting free Ca²⁺ of 100 nM [48], the nominal limit of resolution of free Ca²⁺ change was ~200 nM.

Data and error analysis

For each cell, or for the population average, the voltage dependence of charge movements (Q), Ca²⁺ conductance (G), and peak intracellular Ca²⁺ (AF/Fo) were fitted according to a Boltzmann equation A = Amax/(1+exp(-(V-V1/2)/k)) where Amax was either Qmax, Gmax or AF/Fo; max; V1/2 is the potential at which A = Amax/2; and k is the slope factor. For a fit of steady-state inactivation, the
term (V-V1/2) in the equation above was replaced by (V1/2-V). Ca2+ conductance for each cell was computed from the extrapolated reversal potential and the maximal Ca2+ current at each voltage. In all figures, the symbols and error bars correspond to the population mean ± 1 SEM. The curves correspond to a Boltzmann fit of the mean. In addition to Student t-tests reported in Table 1, one-way ANOVA tests were conducted among the 11 sets of Boltzmann parameters of control, γ1 het and γ1 null cells of Table 1. The null hypothesis (equality of means) was not rejected at a level of significance p<0.05 except in the two cases also identified by t-tests, namely Gmax and V1/2 inactivation. An F-test, which formally tests for the difference between the variances of independent populations, was used to determine which formally tests for the difference between the variances of independent populations (Amax, V1/2, k) had the largest variance, and thus the largest experimental error. The ranking order in variance error was V1/2>Amax>k. Furthermore, the error in V1/2 was the largest for the fluorescence measurement and the smallest for the conductance measurement. This result was expected since the size of the voltage step used in the V1/2 determination was 5 mV for conductance, 10 mV for charge movements, and 20 mV for fluorescence. The number of voltage steps in fluorescence measurements was kept to a minimum to avoid rundown of the Ca2+ transient due to photobleaching. The limit of resolution of V1/2, the parameter with the largest error, was estimated with a z-test which formally tests for the difference between the mean of a population and a hypothetical mean. We determined the smallest difference between an experimental and a hypothesized V1/2 Resolvable at a level of statistical confidence p<0.05. In the calculations, we used the data with the largest V1/2 variance for each measurement (conductance, charge movement and fluorescence). Also, we assumed a hypothetical S.D. equal to the experimental S.D. The estimated limits of statistical resolution were ~3.9 mV for conductance, ~6.5 mV for charge movements, and ~8.5 mV for fluorescence measurements. Thus, a difference in population mean V1/2 of less than the indicated limit would not be statistically significant, given the variance and the number of determinations of the present study. Statistical analyses were performed with Analyse-it software (Leeds, UK).
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