Identification of Amino Acid Residues of the NR2A Subunit That Control Glutamate Potency in Recombinant NR1/NR2A NMDA Receptors

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The NMDA type of glutamate receptor forms a subfamily of ionotropic receptors with distinctive functional and biophysical properties. NMDA receptors play an important role in synaptic plasticity, memory and learning, brain development and excitotoxicity (for review, see Choi, 1988; McBain and Mayer, 1994; Collingridge and Bliss, 1995). Molecular cloning has identified a family of genes that code for subunits of ionotropic glutamate receptors (for review, see Nakanishi, 1992; Hollmann and Heinemann, 1994). One subfamily, GluR1–4, codes for the AMPA type of glutamate receptors, which can be either homo-oligomers or hetero-oligomers of GluR1–4 subunits. NMDA receptors are endowed with a set of properties that distinguish them from other ionotropic glutamate receptors. Magnesium ions block the NMDA receptor channel at the resting potential, and current flows only when the synaptic release of agonist coincides with depolarization, so the receptor may act as a coincidence detector. NMDA receptor channels show a high permeability for calcium ions, the influx of which can trigger important cellular processes.

In the context of this paper two features are of special interest. (1) Both glutamate and glycine have to be present for efficient gating of the NMDA receptor channel. This requirement for a coagonist is in contrast to all other known glutamate receptors. (2) NMDA receptors are hetero-oligomers that are composed of at least two types of subunit: the constant NR1 subunit, which comes in multiple splice variants, and one or more types of an NR2 subunit of which four (NR2A–D) variants are known. This is in contrast to the recombinant AMPA type of glutamate receptors, which can be either homo-oligomers or hetero-oligomers of GluR1–4 subunits.

After the initial cloning of the NR1 subunit (Moriyoshi et al., 1991), it was suggested that the binding sites for glutamate and glycine are located on the NR1 subunit. Indeed, site-directed mutagenesis of the NR1 subunit has identified amino acid residues that control glycine potency (Kuryatov et al., 1994; Wafford et al., 1995; Hirai et al., 1996; Williams et al., 1996). In these studies, single amino acid mutations reduced glycine potency by up to four orders of magnitude, with only small effects on glutamate potency. These studies suggest further that the glycine binding pocket has a bi-lobar structure, similar to that found by crystallography for a family of bacterial periplasmic amino acid binding proteins (Oh et al., 1993, 1994), in which the ligand is bound in a “Venus flytrap” mechanism.

Here we report the identification of amino acids, through site-directed mutagenesis, that control glutamate potency in recombinant NR1/NR2A receptors. These residues were found on the NR2A subunit, at positions homologous to those on the NR1 subunit that control glycine potency. Similar conclusions were published recently by Laube et al. (1997). Furthermore, we now provide evidence that at least some of these residues shape the glutamate binding site. Thus, the overall folding of the area of NR2 subunits that binds glutamate is likely to be structurally similar to the area of the NR1 subunit that binds glycine.
MATERIALS AND METHODS

Expression plasmid constructs. The wild-type expression plasmids for the rat NR1 (1a splice form) and NR2A NMDA receptor subunits were as in Kuner and Schoepfer (1996). Mutations were introduced into the NR2A sequence by a PCR-based strategy. To facilitate cloning and identification of mutants, some NR2A mutants carried additional silent nucleotide substitutions. PCR-generated DNA segments and cloning sites were confirmed by DNA sequencing.

Heterologous expression of NMDA receptors. cRNA was synthesized as runoff transcripts from linearized plasmid DNA using the Promega (Madison, WI) Ribomax RNA synthesis kit. Reactions were supplemented with 0.75 mM capping nucleotide m‘G(5)ppp(5)G (Pharmacia, Uppsala, Sweden) in the presence of 1.6 mM gTP. cRNA amounts and integrity were estimated by intensity of fluorescence in ethidium bromide-stained agarose gels, and concentration was determined also by OD260. NR1 and NR2A cRNAs were mixed at a nominal ratio of 1:1 and diluted with water to ~5 ng/μl of each, before injection. Xenopus laevis oocytes (stage V) were isolated manually after collagenase treatment (Sigma type V, 30 μg/ml, at room temperature for 1 hr). Oocytes were then defolliculated and injected (Drummond Injector) with 23 nl of cRNA mixture. For expression the oocytes were incubated at 19°C for 2–3 d in modified Barth’s solution (in mM): NaCl 88, KCl 1, NaHCO3 2.4, Ca(NO3)2 0.33, MgSO4 0.82, CaCl2 0.44, Tris–Cl 15, adjusted to pH 7.4 with NaOH, supplemented with 50 μM manganese (BRL, Bethesda, MD), 50 μg/ml streptomycin (BRL), and 30 μg/ml t(−2)-amino-5-phosphonopentanoic acid (APV) (Tocris), followed by storage at 4°C until they were used for electrophysiological measurements.

Whole-oocyte recording. Responses to glutamate were measured with a two-electrode voltage-clamp amplifier (TEC05, NPI electronics, Tamm, Germany) at −60 mV using 0.5–2.0 M electrodes filled with 3 M KCl. Oocytes were perfused with nominally Ca2+-free, low- Na+ NFR solution (in mM): NaCl 115, KCl 3, NaH2PO4 1.25, HEPES 20, CaCl2 0.85, pH 7.4, with NaOH supplemented with 20 μM glycine. “Low” glutamate concentrations were used. Because of the low glutamate potency on the T671A mutant, a “low concentration” means 30–200 μM, 100-fold larger than used for the wild type. Patch pipettes, made from thick-walled borosilicate glass (Clark Electromedical Instruments, Pangbourne, UK), contained (in mM): potassium gluconate 141, NaCl 2.5, HEPES 10, EGTA 11, pH 7.4, with KOH. After fire-polishing of their tips, pipettes had resistance of 10–20 MΩ, with a capacitance of ~20–30 pF. For subsequent analysis, data were filtered at 2 kHz (−3 dB) before digitization at 20 kHz. Channel durations and amplitudes were fitted with the SCAN program (Colequhou and Sigworth, 1995) and subjected to a 70 μsec imposed resolution. We define an open period as a period during which the channel appeared to be continuously open, regardless of transitions between different open levels.

RESULTS

NR2A mutations

Ten mutants containing single amino acid substitutions of the NR2A subunit were generated by site-directed mutagenesis, at positions that were chosen either because they control glycine potency on NR1 subunits, or because they control amino acid binding in bacterial binding proteins. Alignment of NR2 sequences with bacterial amino acid binding proteins (Fig. 1A) reveals limited linear sequence identity in segments N terminal of M1 and C terminal of M3, in extracellular domains referred to as S1 and S2, respectively (Stern-Bach et al., 1994). The corresponding segments on NR1 harbor residues that control glycine potency (Kuryatov et al., 1994). Mutants were generated according to the following rule: uncharged amino acids were mutated into alanine, positive charges were substituted by negative charges, and in addition aromatic residues were replaced by another aromatic residue.

Rearrangements coexpression of NR1 cRNA with any of the 10 mutant NR2A cRNAs resulted in robust inward currents after application of glutamate in the presence of glycine (Fig. 1B). Injection of similar amounts of cRNA resulted in maximal agonist-induced currents of comparable magnitude; in particular, I_{max} for the mutants was generally not lower than I_{max} for the wild type (the values are given in the legend of Fig. 2).
Reduced potency of glutamate

Dose–response curves were recorded (Fig. 1B) in conditions (180 μM Ba²⁺) designed to minimize the artifacts caused by agonist-induced influx of divalent cations (Leonard and Kelso, 1990; Williams, 1993). With the exception of NR2A(G664A), all of the mutations we tested shifted the dose–response curves for glutamate to the right (Fig. 2A). For all mutations except S670A (and G664A), the potency of glutamate was reduced by more than fivefold. Three mutations (H466A, G669A, and T671A) decreased the potency of glutamate by more than two orders of magnitude (Table 1). The fitted dose–response curves (Fig. 2A) show that the Hill coefficients for most of the 10 mutant receptors

Figure 1. Single amino acid substitutions in the NR2A subunit generated functional NR1/NR2A receptors. A, Amino acid sequence alignment of rat NR1 (Moriyoshi et al., 1991) and NR2 subunits (Monyer et al., 1992; Ishii et al., 1993) with the bacterial periplasmic binding proteins LAOBP (Oh et al., 1993) and HISJ (Oh et al., 1994). Residues that reduced glycine potency when mutated in NR1 are marked by an arrow above the sequence alignment. NR2A residues that were mutated in this study are marked below the sequence alignments by an arrow if the mutation reduced potency by more than a factor of 2; otherwise they are indicated by a minus sign. Numbers indicate the first residue of the alignment; numbering is for mature proteins without signal peptides. B, Glutamate-activated currents recorded in the two-electrode voltage-clamp configuration from an oocyte expressing wild-type receptors (left) and an oocyte expressing T671A mutant receptors (right). Increasing concentrations of glutamate were applied in the presence of 30 μM glycine.
are not much different from the value found for wild-type NR1/NR2A receptors. The slopes for H466F and V666A are probably lower, and for K465E may be higher, than for the wild type, but the three mutations with the largest increase in $EC_{50}$ show little or no change of the Hill slope.

In contrast to the considerable effect of the mutations on glutamate potency, the potency of the coagonist glycine was barely affected; the $EC_{50}$ values for all 10 mutants lay within a factor of 1.7 of that of the wild type, as shown in Figure 2B.

Table 1. $EC_{50}$ and Hill values for glutamate and glycine

<table>
<thead>
<tr>
<th></th>
<th>Glutamate</th>
<th>Glycine</th>
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<tbody>
<tr>
<td></td>
<td>$EC_{50}$ ($\mu$M)</td>
<td>$n_H$</td>
</tr>
<tr>
<td>NR1/NR2A</td>
<td>2.89 ± 0.12</td>
<td>1.49 ± 0.09</td>
</tr>
<tr>
<td>NR1/NR2A(N463A)</td>
<td>17.9 ± 1.4</td>
<td>1.70 ± 0.18</td>
</tr>
<tr>
<td>NR1/NR2A(K465E)</td>
<td>29.3 ± 1.4</td>
<td>1.91 ± 0.18</td>
</tr>
<tr>
<td>NR1/NR2A(H466A)</td>
<td>624 ± 32</td>
<td>1.55 ± 0.09</td>
</tr>
<tr>
<td>NR1/NR2A(H466F)</td>
<td>38.4 ± 4.6</td>
<td>1.09 ± 0.10</td>
</tr>
<tr>
<td>NR1/NR2A(G664A)</td>
<td>2.08 ± 0.25</td>
<td>1.46 ± 0.21</td>
</tr>
<tr>
<td>NR1/NR2A(T665A)</td>
<td>19.0 ± 2.0</td>
<td>1.18 ± 0.11</td>
</tr>
<tr>
<td>NR1/NR2A(V666A)</td>
<td>33.4 ± 1.6</td>
<td>1.12 ± 0.05</td>
</tr>
<tr>
<td>NR1/NR2A(G669A)</td>
<td>927 ± 46</td>
<td>1.38 ± 0.07</td>
</tr>
<tr>
<td>NR1/NR2A(S670A)</td>
<td>4.97 ± 0.30</td>
<td>1.35 ± 0.09</td>
</tr>
<tr>
<td>NR1/NR2A(T671A)</td>
<td>2967 ± 279</td>
<td>1.30 ± 0.09</td>
</tr>
</tbody>
</table>
receptors in the absence (glutamate dose–response curves for one oocyte expressing T671A mutant being extrapolated for display purposes. Fitted lines have slopes constrained to be the same for all curves. Fitted lines have been extrapolated for display purposes. (A)

Figure 3. Schild analysis of the competitive antagonism of the glutamate binding site by APV. A. Partial, low-concentration, glutamate dose–response curves for one oocyte expressing wild-type receptors in the absence (open squares) and presence of 3 μM (filled diamonds), 10 μM (open pentagons), 30 μM (filled hexagons), and 100 μM (open circles) APV. Dashed lines represent free fits of the power function (see Materials and Methods) to the data, and solid lines show fits of the same function but with slopes constrained to be the same for all curves. Fitted lines have been extrapolated for display purposes. B. Partial, low-concentration, glutamate dose–response curves for one oocyte expressing T671A mutant receptors in the absence (filled pentagons) and presence of

Reduced affinity for APV
To examine the underlying mechanism that is responsible for the decreased potency of glutamate (see Discussion), we analyzed the NR1/NR2A(T671A) mutant in greater detail. Schild’s method (Arunlakshana and Schild, 1959) was used to determine the equilibrium constant, $K_B$, for the competitive antagonist APV (Fig. 3).

The slope of the Schild plot was $1.06 \pm 0.04$ for the wild type and $0.94 \pm 0.16$ for T671A. These values are close to (and do not differ significantly from) 1.0, so there is no evidence against the view that APV is a competitive antagonist on both receptors. Therefore the plots were refitted with the Schild equation (slope fixed at 1), as described in Materials and Methods, and these fits are shown in Figure 3C. For the wild type we found that $K_B = 1.26 \pm 0.07 \mu M$, which is similar to the value of $1.35 \mu M$ reported by Verdoorn et al. (1989) for rat brain mRNA expressed in oocytes. For the T671A mutant, however, we found a much reduced affinity for APV, with $K_B = 321 \pm 30 \mu M$. This 255-fold reduction in affinity is comparable with the 1000-fold reduction of potency for glutamate.

Single-channel conductance
The single-channel currents evoked by glutamate in NR1/NR2A(T671A) were remarkably similar to those evoked in the wild type (Fig. 4). The mean chord conductances of their main level and sublevel were estimated as $51.1 \pm 1.1$ pS and $38.6 \pm 0.9$ pS ($n = 5$), compared with values of $50.1 \pm 1.4$ pS and $38.3 \pm 1.3$ pS reported for wild-type NR1/NR2A by Stern et al. (1992). The relative areas for these two components, 85% and 15%, were also similar to those for wild type (80% and 20%). In addition, the frequencies of direct transitions from shut to main level, from shut to sublevel, and from main to sublevel of T671A channels (Table 2) were similar to those reported for the wild type by Stern et al. (1992). Like the wild-type NR1/NR2A, they showed no sign at all of the sort of temporal asymmetry shown by NR1/NR2D receptors (Wyllie et al., 1996).

Single-channel kinetics
In contrast to the close similarity of the conductances, there were kinetic differences between T671A and the wild type. For example, both mutant and wild-type open period distributions contained three exponential components (Fig. 5). The mean time constants for each component were $6.62 \pm 17.0$ μsec, $1.10 \pm 0.34$ μsec, and $6.91 \pm 2.45$ μsec for T671A channels ($n = 5$), and $47.6 \pm 9.3$ μsec, $0.823 \pm 0.375$ μsec, and $3.08 \pm 0.57$ μsec for wild-type channels ($n = 5$) (P. Béhé and D. Wyllie, personal communication). The overall mean apparent length of the open periods was $4.33 \pm 0.71$ msec for T671A compared with $1.69 \pm 0.16$ msec for the wild type (also see Fig. 4). These values are clearly different ($p = 0.0045$ by two-tailed randomization test).

For this comparison, both wild-type and mutant data were analyzed with the same resolution (70 μsec for both open and shut times). Such a resolution would mean that a substantial number of brief shuttings were missed; however, it is not possible to do a satisfactory correction for missed events without knowledge of a realistic kinetic mechanism (Colquhoun and Hawkes, 1995).
Kinetic differences between mutant and wild-type receptors were also observed in concentration jump experiments. Preliminary data, shown in Figure 6, indicate that deactivation of T671A receptors, after a 100 msec jump of 10 mM glutamate, occurs with a time constant of 18.2±3.0 msec (n=4). This is approximately one order of magnitude faster than that found for the deactivation of wild-type NR1/NR2A channels after “long” pulses of glutamate (Monyer et al., 1992; Medina et al., 1995) (D. Wyllie and P. Béhé, personal communication).

**DISCUSSION**

**Glutamate effects are mediated through NR2 subunits**

We have shown that mutagenesis of NR2A residues reduced the potency of glutamate by up to 1000-fold, with very little effect on glutamate responses of NR1/NR2B receptors in HEK293 cells (Anson et al., 1995). Moreover, NR1/NR2A channels are the major contributors to the NMDA response in all the cell types studied (Anson et al., 1995). Alternatively, it is possible that the NR2A subunits alter the pharmacology of glutamate receptors without changing their sensitivity to glutamate. Previous studies demonstrated that NR2A subunits modulate the affinity of CaM kinase II for the NR2A subunits (Miyazaki et al., 1994). These results suggest that the phosphorylation state and intracellular localization of CaM kinase II may be modulated by NR2A subunits, which in turn may affect the sensitivity of the NMDA receptor to glutamate.

**Table 2. Transitions between conductance levels for NR1/NR2A(T671A) channels**

<table>
<thead>
<tr>
<th>Transition type</th>
<th>n (N = 10897)</th>
<th>%</th>
</tr>
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<tbody>
<tr>
<td>Shut-sublevel</td>
<td>400</td>
<td>3.67</td>
</tr>
<tr>
<td>Sublevel-shut</td>
<td>366</td>
<td>3.36</td>
</tr>
<tr>
<td>Shut-main level</td>
<td>4210</td>
<td>38.63</td>
</tr>
<tr>
<td>Main level-shut</td>
<td>4241</td>
<td>38.92</td>
</tr>
<tr>
<td>Sublevel-main level</td>
<td>860</td>
<td>7.89</td>
</tr>
<tr>
<td>Main level-shut</td>
<td>820</td>
<td>7.53</td>
</tr>
</tbody>
</table>

Experiments were performed in the presence of 20 μM glycine and between 60 and 200 μM glutamate.

*N* refers to the total number of transitions, longer than 2.5 *t<sub>r</sub>*, analyzed from five recordings.
the potency of glycine. Our evidence strongly suggests (see below) that the binding site for glutamate on NMDA NR1/NR2A receptors is formed by NR2A subunits. The residues we found to be important in NR2A are fully conserved within the NR2 family (Fig. 1A), which suggests that in all types of NR1/NR2 NMDA receptors glutamate effects are mediated by the NR2 subunit. Comparison of our data with the results of Kuryatov et al. (1994) and Wafford et al. (1995) on glycine potency of NR1 mutations suggests that the glutamate effects mediated through the NR2 subunits, and glycine effects mediated through the NR1 subunit, are each based on the same structural design.

While the experiments described here were in progress, Laube et al. (1997) reported that mutation of NR2B residues reduced glutamate potency on NR1/NR2B receptors. Three of their mutations were equivalent to ours; for two of them we both found a comparable (approximately 10-fold) reduction in potency of glutamate (H466F, V666A), whereas for one (K465E, equivalent to NR2B K459E) we found a considerably smaller effect. For NR2A S670, we analyzed the effect of the mutation to alanine, S670A, and found only a minimal effect; the same serine to alanine mutation at the equivalent position on the NR1 subunit also has little effect on glycine potency (Kuryatov et al., 1994). In contrast, Laube et al. (1997) report a 180-fold reduction of glutamate potency for a mutation into glycine at the homologous position (S664G). The mutations that gave the three largest increases in glutamate EC50 in Laube et al. (1997) (236-, 180-, and 118-fold) were all at different positions from those that gave the largest shifts here (1027-, 321-, and 216-fold). Furthermore their mutant with the largest shift actually showed a decreased IC50 for APV, although that with the 180-fold shift showed a 40-fold increase in IC50. Our most-shifted mutant, T671A, showed a 255-fold increase in the equilibrium binding constant of APV. Laube et al. (1997) proposed an appealing three-dimensional arrangement for the structure of the glutamate binding site, and it will be interesting to see how this proposal can accommodate the rather different residues that seem, from the mutants tested here, to be important for glutamate binding.

Evidence that reduced potency is caused by impaired binding of glutamate

The basis for the inference that the mutated residues form part of the glutamate binding site requires clarification. A reduction of potency is not necessarily caused by a change in the binding site but can also be caused by a change of gating. In other words, the glutamate binding site might not be affected at all; furthermore, a binding assay for glutamate would not resolve this ambiguity (see Colquhoun and Farrant, 1993).

To infer that the mutated residues form part of the binding site it is necessary to show that the mutation has altered the microscopic equilibrium constant for the initial binding step: the binding of glutamate to the resting state of the receptor. If this were the only change we could say that it was very likely that these domains form part of the glutamate binding site (we do not distinguish in this paper between residues that interact directly with the ligand and residues that shape the binding site without direct interaction with the ligand).

One way to examine this problem is to use a competitive antagonist rather than an agonist. This removes most of the complications in interpretation of the effect of a mutation, because a competitive antagonist, unlike an agonist, is not expected to cause a change of the receptor conformation to higher-affinity active states (i.e., gating). Thus any change in their binding is likely to result directly from a change in the binding site. The equilibrium constant for antagonist binding, K1, cannot be determined from IC50 measurements, so Schild’s method was used. It is possible that binding of the agonist might be altered in a mutant receptor without alteration of binding of a competitive antagonist, because the binding sites for the two compounds, although presumably overlapping, are not identical. However, we found
that the T671A mutation increased the equilibrium binding constant 255-fold for APV compared with wild type. Therefore in this mutant, APV binding is impaired greatly, which implies that its binding site, and therefore probably that for glutamate, has been altered.

The equilibrium concentration–response curves also suggest a primary effect of the mutation on the binding site for glutamate. The main finding is that mutations in the NR2 subunit can produce an enormous reduction in the potency of glutamate ($EC_{50}$), with little change in the Hill slope, and very little change in the potency of glycine (Table 1). Of the three most effective mutations, one precedes M1 (S1 domain) and the others follow M3 (S2 domain), which suggests that both regions contribute to the binding site.

A reduction in the open-shut equilibrium constant could produce this result without any detectable change in maximum response, provided that its reduced value is still such that most channels are open at equilibrium. How can we exclude this possibility? The first reason is that for many cooperative mechanisms we expect the shift of the $EC_{50}$ to be proportional to the binding constants but roughly proportional (in the case of two bindings being required) to the square root of the gating constant (as long as this equilibrium is well over toward the open state). In this case the 1000-fold increase in $EC_{50}$ seen with T671A would require a roughly millionfold reduction in the gating constant. If this were to occur without a drastic reduction of the maximum response, the gating constant for the wild-type channel would have to be enormously high. This does not appear to be the case, because glutamate actually behaves like a partial agonist on NMDA receptors with a maximum $P_{\text{open}}$ of ~0.3 (Gibb and Colquhoun, 1992; Dzubay and Jahr, 1996). Therefore, such a reduction in the gating constant would cause an enormous reduction in the maximum response. In common with almost every mutation study, we have no good evidence as to whether the maximum response is changed, because of uncertainties about the relative efficiency of expression of wild-type and mutant RNAs; however, there was no systematic difference between the maxima observed (Fig. 2 legend), so it is clear that changes by a factor of one million have not occurred.

Furthermore, most kinetic mechanisms predict that the Hill slope would be reduced substantially if there were such a large effect on the gating constant as would be required to account for our observations; we observed no such reduced Hill slope.

Interpretation of kinetic data of the T671A mutant

In principle, effects on channel gating can be measured from kinetic analysis of single-channel recordings (Colquhoun and Sakmann, 1985). We show that transition frequencies between the conductance levels of T671A (as well as the levels themselves) are similar to those of the wild type. Therefore, at least this aspect of gating is not altered in the mutant with the largest reduction in potency.

The observation that the mean apparent open period is longer for the T671A mutant could mean that the channels stay open rather longer, which would imply some effect on the shutting rate of the channel, i.e., on the gating process. Such a change, however, is in the wrong direction to explain reduced glutamate potency. In any case it is quite conceivable that there is no real increase in the length of individual openings; the apparent increase could well result from more rapid dissociation of the agonist, which would be expected to reduce the duration and frequency of brief shuttings such that fewer of them are detected and openings appear, incorrectly, to be longer.

Figure 6. Fast concentration jumps on the T671A mutant receptor. A, Two individual responses obtained from an outside-out patch after a 100 msec pulse of 10 mM glutamate (in the presence of 20 µM glycine). Single-channel currents can be seen clearly in these example traces. The timing and duration of the glutamate application are indicated above each pulse. B, The average of 25 such jumps. The deactivation of this ensemble current is rapid and can be reasonably well described by a single exponential with a time constant of 16 msec, shown as a white line.
Figure 6 shows that the decay of the response, after rapid removal of glutamate, is roughly 10-fold faster for T671A than for wild-type receptors. The decay rate of the current is a reflection of the length of the underlying channel activation, or burst (Colquhoun et al., 1997). We have found that the individual openings, if anything, are longer in the mutant receptor, so the fast decay cannot be explained by shorter open times. The length of an activation will depend on the number of times that the channel opens while agonist is still bound. The more rapid decay is therefore likely to result from there being fewer openings during an activation for the mutant channel compared with the wild type. This could result from either (1) an increased rate of dissociation of glutamate or (2) a reduced opening rate constant for the mutant channel. If we are right in assuming that wild-type and mutant receptors express with comparable efficiency, then, as discussed above, it is unlikely that the opening rate constant is greatly reduced in the mutant, so this is also unlikely to be the main reason for the faster decay (although a contribution from this source cannot be ruled out). Thus the main reason is likely to be that glutamate dissociates more rapidly, thereby reducing the probability that the channel will reopen. An increase in dissociation rate is consistent with a reduced affinity for glutamate.

Implications

In the absence of any completely satisfactory kinetic mechanism for the NMDA receptor, it is not possible to infer from the results, in a rigorous manner, numerical values for the microscopic equilibrium constant for the initial binding step or for the gating constants. As discussed above, it is most likely that the primary effect, at least for the three most effective mutations, is on glutamate binding.

The model of a conserved structural design of agonist binding sites of the extended ionotropic glutamate receptor family is further supported by other studies. Paas et al. (1996) investigated ligand binding to chick cerebellar kainate binding protein, and Kuusinen et al. (1995) found ligand binding by a recombinant fusion protein consisting only of fused S1 and S2 domains of an AMPA receptor.

REFERENCES